Research and Extension Needs for Integrated Pest Management for Arthropods of Veterinary Importance.

Proceedings of a Workshop in Lincoln, Nebraska, April 12-14, 1994

Last Updated – October 2001

Christopher J. Geden and Jerome A. Hogsette, Editors

Center for Medical, Agricultural, and Veterinary Entomology
USDA-ARS, Gainesville, Florida

Contents:

Introduction

In April, 1994, a workshop was held in Lincoln, Nebraska, to update the IPM document that resulted from a similar workshop in Manhattan, Kansas, in 1979 (Anonymous 1979). The workshop was initiated by Dr. Ralph A. Bram, USDA National Program Leader, and organized by Drs. Gustav D. Thomas, USDA-ARS, Lincoln, and John B. Campbell, University of Nebraska, North Platte. Participants were charged with assessing the current status of IPM programs for pests of veterinary importance, identifying needs for program improvements, and recommending future research and extension priorities. Participants, invited from federal and state government research and extension organizations, and the private sector, from the U.S. and Canada, were selected because of their expertise in various sectors of the field of veterinary entomology.

IPM needs of eight animal commodity groups were addressed at the Lincoln workshop: 1) poultry; 2) dairy cattle; 3) range beef cattle; 4) confined beef cattle; 5) swine; 6) sheep and goats 7) horses; and 8) dogs and cats. A subcommittee representing each commodity group prepared the chapters contained in the report. Formats vary somewhat from one chapter to another according to the subcommittees' needs, however each chapter contains an overall summary at the beginning, followed by a discussion of the major pests, research and extension needs, and issues pertaining to the animal group. The texts for these chapters were originally prepared in 1995 and updated in 2000-2001.

Printing this document. To upload, edit or print this document or its individual sections the best method is to first save the entire document on your hard drive. It can then be opened with MS Word (be sure to select either "all files" or "web pages" under the "files of type" dialogue box at the bottom of the File – Open menu). You can then either save/edit the document as .htm (for uploading to a local server) or save it as a Word or text file for editing/printing.

References

Anonymous. 1979. Proceedings of a Workshop on Livestock Pest Management: To Assess National Research and Extension Needs for Integrated Pest Management of Insects, Ticks, and Mites Affecting Livestock and Poultry, March 5 7, 1979, Kansas State University, Manhattan, Kansas. USDA, 322pp.

POULTRY SUMMARY

Poultry is a rapidly growing component of the agricultural economy of the United States ($15 billion/year) and the world, with sales in some areas exceeding the combined value of all other agricultural commodities. Broiler production has led the industry, with a doubling of production in the last 20 years. A significant portion of this new production goes to export markets, and this trend is expected to accelerate well into the next century as NAFTA and GATT expand the market for US poultry products. Although the poultry industry continues to be plagued by numerous arthropod and rodent pests, the number of research and extension personnel needed to develop pest management programs has declined during the last 10 years. More than ever it is essential for industry, state, and federal entomologists to work together to solve arthropod pest problems affecting American agriculture. This committee identified the following critical areas of research and extension needs for poultry IPM:

1. Cultural control, especially manure management, must be emphasized as the first line of defense against pest species of flies. Research is needed to determine facility design features and manure handling methods that limit the availability and suitability of manure for flies.

2. In the past 15 years litter beetles, especially the lesser mealworm (*Alphitobius diaperinus*), have emerged from obscurity to become the most serious pest affecting several types of poultry production systems. There is an urgent need to develop effective monitoring and management programs for these pests.

3. Biological control remains an uncertain pest management tool. Practical research is needed to develop biocontrol into an essential component of IPM programs for flies and litter beetles.

4. A renewed effort is needed to study nutrition, ecology and behavior of house flies and *Fannia* spp. in the field, especially appetitive orientation and dispersal behaviors.

5. High levels of insecticide resistance in house fly populations have made it increasingly difficult to control outbreak populations of this pest. There continues to be a need for novel chemical and biological insecticides that are compatible with other fly management strategies.

6. Rodents are serious pests in most poultry production systems and need to be included in overall pest management programs for the industry.

7. Research is needed to determine the efficacy of currently available pesticides and application methods against external parasites of poultry and, if possible, to discover non-chemical control methods.

8. Extension resources for program delivery have declined to critically low levels, and extension specialists are required to cover many more areas of responsibility than in the past. University researchers, Cooperative Extension, ARS, and industry representatives should work together as a national consortium to share information. An electronic data base and web site on the Internet would greatly facilitate this process.

Arthropod pest management does not occur in a vacuum, and IPM recommendations must be compatible with other management considerations such as flock health, nutrition, housing design, and the economics of production. In addressing these critical needs, entomologists should work with poultry scientists, agribusiness representatives, veterinarians, economists, agricultural engineers, rodent control specialists and others to ensure that research remains grounded in the practical needs of agriculture in the 21st century.

POULTRY

Committee Members

Christopher J. Geden (Co-chair)
James J. Arends, (Co-chair)
Richard C. Axtell
Donald R. Barnard
Douglas M. Gaydon
Leslie A. Hickle
Jerome A. Hogsette
William F. Jones
Bradley A. Mullens
Maxcy P. Nolan, Jr.
Maxcy P. Nolan, III

James J. Petersen
D. Craig Sheppard

Economic Significance of Poultry

In 1992 the farm value of principal components of the poultry industry was about $15 billion. Production of broilers and turkeys has increased dramatically in the last 20 years, reflecting a change in the eating habits of many Americans and an expanding export market (Table 1). Egg production has remained stable, with a decrease in per capita consumption of shell eggs (305/ year in 1972 to 235/year in 1992) largely offset by increases in demand for exports and for further processing. Other poultry commodities such as ducks and rattites represent small but economically robust components of the industry.

Broiler and turkey production has rapidly moved to a vertically integrated industry structure where the farmer provides land, labor and housing for an integrator, who provides birds, feed, animal health care and marketing of the final product. Egg production has shifted away from small independent producers towards larger agribusinesses with corporate structures that vary from one part of the country to another. The broiler industry continues to be concentrated in the southeastern US; the layer industry in California and midwestern and southern states; and the turkey industry in the Southeast and Midwest (Table 2). The broiler industry is expected to continue its expansion in the coming decade, with a significant portion of that expansion driven by exports.

Table 1. Poultry and Egg Production in the United States, 1972-1992.

Product		Production (millions)			Percent change		
		1972	1982	1997	'72-'82	'82-'97	'72-'97
Broilers	No. produced	3,075	4,149	7,764	34.9	87.1	152.5
	Farm value	$1,623	$4,502	$14,149	177.5	214.3	771.8
.							
Turkeys	No. produced	129	166	301	28.6	81.3	133.3
	Farm value	$537	$1,225	$2,884	133.7	135.4	437.1
.							
Eggs	No.	69,219	69,718	77,532	0.07	11.2	12.0

	produced						
	Farm value	$1,781	$3,459	$4,540	94.2	31.2	154.9
.							
Total Farm value		$3,900	$9,200	$21,600	135.9	134.8	453.8

Table 2. Ranking of states in broiler, layer, and turkey production.

Rank	Broilers		Turkeys		Layers	
	1982	1997	1982	1997	1987	1997
1	AR	GA	NC	NC	CA	OH
2	GA	AR	MN	MN	GA	CA
3	AL	AL	CA	AR	AR	PA
4	NC	MS	AR	VA	PA	IN
5	MS	NC	MO	CA	IN	IA
6	MD	TX	VA	MO	TX	GA
7	CA	MD	IA	IN	AL	TX
8	TX	VA	IN	PA	NC	AR
9	DE	DE	PA	SC	NY	MN
10	VA	MO	-	-	OH	NC

Pests of Poultry Production

The committee considered the following poultry pests:

House flies (*Musca domestica* and *Fannia* spp.)
Litter beetles (*Alphitobius*, *Dermestes*, others)
Northern fowl mite (*Ornithonyssus sylviarum)*
Rodents
Bed bugs (*Cimex lectularius*)
Red Mite (*Dermanyssus gallinae)*
Black flies and biting midges
Lice (especially *Menacanthus*)
Mosquitoes
Turkey chigger (*Neoschoengastia americana*)

Sticktight fleas (*Echidnophaga gallinacea*)
Fowl ticks (*Argus persicus*)

The focus of the remainder of this report will be on the three pests that the committee identified as having the largest impact on the industry under the widest range of geographic and management conditions: house flies, litter beetles and the northern fowl mite. Although rodents are serious pests of the industry and need to be addressed in overall IPM programs, the committee felt that it lacked sufficient expertise in this area to make specific recommendations on rodent research.

House Flies

Description and Biology. Several species of muscoid flies are pests on or around poultry operations. The most important pests develop primarily in accumulated manure and include the house fly, *Musca domestica*, and the little house fly, *Fannia canicularis*. *Hydrotaea* (=*Ophyra*) *aenescens* and *H. ignava (=leucostoma)* larvae can be abundant in manure and may be mistaken for house fly larvae. *Hydrotaea* larvae feed on manure and larvae of other flies (facultative predators). Immatures of other *Fannia* spp., notably *F. femoralis* and *F. benjamani*, also can be common, and occasionally the biting stable fly, *Stomoxys calcitrans*, is found in poultry manure as well. In some areas, particularly the southeastern U. S., soldier flies (*Hermetia illucens*) are found in large numbers in manure accumulations. Flies in the family Calliphoridae flies (blow flies) primarily develop in carrion and can become problems where poultry carcasses or broken eggs accumulate. Small dung flies of the family Sphaeroceridae are common throughout the world, and some species (e.g., *Coproica hirtula*) occasionally are very abundant, especially in the weeks after cleanout.

Literature on IPM of filth flies in poultry systems is extensive and was reviewed recently by Axtell and Arends (1990) and Axtell (1999). Literature cited here is meant to supplement rather than repeat those reviews. House flies oviposit on moist manure, and the eggs generally hatch within 24 hours. Development is temperature dependent, but larvae can develop through three instars, pupate in somewhat drier areas of the manure, and emerge as adults within 7-10 days at warm temperature. Adult flies tend to remain near the development sites, but may disperse several kilometers to colonize new areas. Little is known about factors that promote fly dispersal. The life cycle of the *Fannia* spp. is similar, except that the duration, particularly the pupal period, is longer, requiring about 2-3 weeks from egg to adult. While house flies are warm weather pests, the *Fannia* spp., particularly *F. canicularis*, develop poorly at temperatures above 27-30oC. *Hydrotaea aenescens* and *H. ignava* also require two weeks or more for development to the adult stage, while the blow flies may develop in only a few days. The life cycle of *Hermetia illucens* requires 4-8 weeks.

Because the worst fly pests develop in accumulated poultry manure, filth fly problems are most severe in caged laying hen operations and in breeder houses where birds are housed on slats. In these systems, manure falls beneath the birds and lies relatively undisturbed for several weeks to a year or more. Over time, accumulated manure is colonized not only by flies but also by numerous competitors, predatory beetles and mites that feed on fly eggs and larvae, and parasitic wasps that attack fly pupae. Biological control and habitat management to optimize activity of these natural enemies are significant components of IPM. Other poultry operations, such as grow out houses for turkeys or chickens, have loose litter (e.g., wood shavings) through which the birds move and forage. Accumulations of manure or significant numbers of flies usually do not occur in these settings.

Economic Importance. Filth flies are not known to cause direct production losses in poultry, except for their role in transmission of avian pathogens. Because the same pathogens also can be transmitted by other mechanical means, the exact role of flies in the epizootiology of most diseases of poultry is uncertain. We estimate that the industry currently spends about $20,000,000 per year on pesticides for fly control, exclusive of labor costs and other fly management efforts This figure is based on an estimated annual cost of $0.07 per bird.

The primary economic impact of filth flies is through irritation of people on and near the poultry operations. Flies regurgitate and defecate on resting surfaces, causing unsightly and unsanitary spots on walls, eggs, etc. Flies directly annoy workers and disperse to neighboring houses or businesses, where the presence of flies may violate health ordinances and lead to legal challenges. Producers may be compelled to take costly corrective action or be forced to close the operations in severe cases. Adults of *F. canicularis* are particularly visible because the males assume aerial patrolling stations and hover at eye level under house eaves or in the centers of residential rooms. *Drosophila repleta* is a pest in caged layer and breeder houses where feed is allowed to become wet such as under slats and on dropping boards and other areas that are difficult to treat or clean (Harrington and Axtell 1994)

Recently there has been renewed interest in house flies as carriers of pathogens of animals and humans. House flies have been investigated as carriers of rotaviruses (Tan et al. 1997), *Shigella* (Levine and Levine 1991, Cohen et al. 1991), trachoma (Emerson et al. 1999), *Helicobacter pylori* (Gruebel et al. 1997), mycobacteria (Fischer et al. 2001), *Escherichia coli* (Iwasa et al. 1999, Moriya et al. 1999, Sasaki et al. 200), *Corynebacterium pseutotuberculosis* (Braverman et al. 1999, Zurek et al. 2001), *Giardia lamblia* (Doiz et al. 2000), *Vibrio cholerae* (Fotedar 2001), and *Cryptosporidium parvum* (Graczyk et al. 1999).

Adults of some flies whose larvae are common in manure are not as pestiferous as the house fly and little house fly. Adult *H. aenescens*, for example, leave fecal and regurgitation spots but tend to remain in the manure pits and on vegetation adjacent to the poultry houses. The larvae of *H. illucens* churn and liquify manure when humidity is high. Although this can complicate manure storage and handling, larval activity of *H. illucens* suppresses house fly oviposition (Bradley and Sheppard 1984), and the adults generally are not regarded as pests.

Current Control and Monitoring Methods

Cultural Control. Fly control should begin with manure management and other cultural control practices that limit fly access to suitable oviposition and developmental substrates. Moisture control is an important part of manure management, and poultry houses need to be designed to allow for proper drainage and air movement. Freshly-deposited hen manure is about 80% water (ideal for house flies), but it begins to lose moisture after defecation. Good ventilation and elimination of water leaks promote manure drying. Dry manure (<50% moisture) is unattractive for fly oviposition, unsuitable for fly larval development, and enhances activity of natural enemies which prey on or parasitize fly immatures. Very wet manure (>85% moisture) also is unsuitable for fly larval development.

Frequent manure removal can eliminate most fly breeding. If manure is removed frequently, the interval should be short enough to prevent successful development of a complete generation; this timing will vary with the fly species and temperature conditions. Generally, cleanouts at less than 1-week intervals will interrupt fly life cycles. Infrequent removal, with long manure accumulation times, tends to result in the establishment of a diverse fauna of competitors and natural enemies of pest fly species. Fly production from such complex communities is generally lower than from fresh droppings, where species richness is much lower. Where manure does accumulate, a pad of dry manure left after cleanout is helpful to elevate the manure above ground level and encourage manure drying (in open-sided houses especially), and possibly to serve as a refuge for natural enemies (Mullens et al. 1996a). Removal of only a portion of the manure mass at a time may also enhance colonization of the cleaned areas by natural enemies (Mullens et al. 1996b). Composting poultry manure greatly reduces its ability to support fly development (Moon et al. 2001).

Under the right circumstances, light traps with electrocuting grids can be useful in managing populations of house flies in poultry housing (Rutz and Scoles 1988, Pickens et al. 1994). Large sticky traps can also be effective, but their use is limited by the rapid accumulation of dust on the sticky material (Kaufman et al. 2000a).

Biological Control. Biological control is a significant part of IPM, and cultural techniques mentioned above may help to foster and preserve existing natural enemies. The histerid beetle *Carcinops pumilio* and the mite *Macrocheles muscaedomesticae* are the principal predators of house fly eggs and larvae in poultry manure (Geden 1990). Larvae of *Hydrotaea* spp. also prey on larvae of other flies and could be manipulated to exclude the more pestiferous *M. domestica* (Turner and Carter 1990, Hogsette and Jacobs 1999, Farkas et al. 1998). Predators in general have received less research emphasis than parasitoids. Part of the reason for this is the relative difficulty in developing mass-rearing methods for predators. About a dozen species of pteromalid parasitoids, many of them in the genera *Muscidifurax* and *Spalangia*, oviposit on fly pupae and have been the subject of considerable research as biocontrol agents. Parasitoids have been released on numerous occasions and in some cases have resulted in reductions of fly populations (Rutz and Patterson 1990, Geden et al. 1992, Petersen and Cawthra 1995, Petersen and Currey 1996). There also have been many disappointing results with parasitoids, however, and the reasons for success and failure are poorly understood. It is still uncertain which species are most effective in poultry houses, or whether species combinations should be used (Kaufman et al. 2001a,b). Recent research on parasitoid life history may lead to a better understanding of niche characteristics of these species (Lysyk, 1998, 2000, 2001a, 2001b, Geden 1996, 1997a, 1999). The competitor *Hermetia illucens* often is abundant in the Southeastern US, where it provides control of house fly and lesser house fly for some egg producers who allow populations of soldier flies to develop (Sheppard 1983). The fungal pathogen *Entomophthora muscae* is a common and significant mortality factor in adult house fly populations (Mullens et al. 1987, Geden et al. 1993, Six et al. 1996), but the fungus is limited by high temperatures (Watson et al. 1993, Madeira 1998, Kalsbeek et al. 2001), and manipulations in operating poultry systems have not been attempted. Other entomopathogenic fungi such as *Beauveria bassiana* and *Metarhizium anisopliae* occur in house fly populations and are virulent for flies, but little is known about their utility as management tools (Barson et al. 1994, Geden et al. 1995b, Carswell et al. 1998, Renn et al. 1999). *Bacillus thuringiensis* delta endotoxins and beta exotoxins have some promise, but are still in the early stages of development. Other pathogens (other bacteria, viruses, protozoans) or their associated toxins may in some cases have potential applications (Johnson et al. 1998, Zhong et al. 2000). Steinernematid and heterorhabditid nematodes do not tolerate poultry manure conditions well and are rapidly inactivated, but may can be effective in certain delivery configurations (Renn 1994, 1998). A tylenchid nematode has been found in Brazil that causes parasitic castration of the adult fly (Coler and Nguyen 1994, Geden 1997b).

Chemical Control. Chemical control has been a mainstay of fly management, with the focus on residual or space sprays of organophosphates, pyrethrins, and pyrethroids as adulticides, and organophosphates as larvicides and organophosphates and carbamates in adulticidal bait formulations. Insecticides that are applied directly to the manure as larvicides can have severe nontarget effects on beneficial arthropods in the manure (Wills et al. 1990). An exception is cyromazine, a larvicide that does not appear to have mortality effects on beneficials. Reliance on insecticides for fly control is waning somewhat because of increased environmental constraints, regulatory limitations on use patterns, insecticide resistance in the target pests (Sheppard et al. 1992), and because fewer new products are being developed. Recently there has been some interest in reexamining older fly control materials such as borates (Hogsette and Koehler 1992, Mullens and Rodriguez 1992).

Monitoring. Current adult fly monitoring methods, using spot cards or baited jug traps, are satisfactory for monitoring relative fly activity within a production facility. Sticky cards are another option that can provide rapid information of fly abundance and distribution patterns (Hogsette et al. 1993). Positioning of cards or traps and the color or toxicant used in jug traps greatly affect the counts obtained (Lysyk and Axtell 1986, Burg and Axtell 1984). House flies tend to avoid resting in areas with high wind speeds, and air movement through modern enclosed poultry houses creates airflow patterns that have a substantial effect on fly counts using spot cards (Geden et al. 1999). These variations make comparisons of fly populations between farms difficult, although carefully placed spot cards can be used to estimate actual fly abundance if temperature data are available. Economic injury or nuisance thresholds have not been established for adult flies. House fly larvae can be monitored by pupal traps or extraction from manure using Berlese funnels or flotation in 0.6M sucrose (Stafford and Bay 1994, Tobin and Pitts 1999).

Needs for IPM. Chemical usage is declining for the reasons noted previously, but the need persists for effective, target-specific, and environmentally acceptable chemicals (including "biological insecticides") for short-term fly suppression. Research is needed on adult fly behavior and chemical or physical cues involved in attraction to bait stations or trapping devices. Sublethal effects (e.g., egg sterility) or delayed mortality effects seldom have been considered, but should be. Some materials or baits incorporating bacterial toxins, microbial or nematode pathogens, or toxic elements such as boron take several hours or days to exert their effects. Such materials may still be useful in fly control, but they are difficult to evaluate.

We still lack definitive data on effects and benefits of cultural manipulations such as manure handling and ventillation patterns. Similarly, there remain surprising gaps in our understanding of the fate of fly immatures in manure that is applied to land during cleanouts. For example, how long after spreading does manure remain suitable for fly development? Knowledge of how these manipulations affect habitat quality, fly oviposition and survival of immatures, and natural enemies is required to better incorporate cultural manipulations into IPM programs. Other means of altering habitat quality, such as hen strain or diet effects on manure moisture and chemical composition (Barnard and Harms 1992), offer potentially fruitful areas for research. Coordinated studies between entomologists and agricultural engineers on modified manure removal schedules, equipment, and storage and processing should be fostered and improved to document benefits or drawbacks of modified manure handling procedure such as continuous belt removal.

Biological control is being recognized as an increasingly important part of filth fly IPM. Biological insecticides offer new potential for periodic suppression (or even sustained suppression, pending adequate delivery systems) of adult or immature filth flies. Identification and isolation of new microorganisms, including subunit toxins, should continue, with appropriate screening and experimental evaluation for field utility and nontarget effects. With the exception of pathogens, naturally-occurring fly enemies have been reasonably well surveyed in representative parts of the U.S. Improved techniques and quality control for production of predators and parasitoids are needed. This should include experimental efforts to monitor and hopefully preserve or enhance desirable characteristics such as freedom from disease, searching ability, tolerance of adverse environmental conditions, or capacity for increase (Geden et al. 1992a, 1995a, Zchori-Fein et al. 1992, Becnel and Geden 1994). Many attempts have been made to evaluate parasitoid releases, but efforts often have been hampered by the inability to differentiate impacts of released material from naturally-occurring activity and by difficulties inherent in measuring fly and parasitoid density. Molecular tools such as cuticular hydrocarbon analysis and RAPD-PCR present us with opportunities to use modern methods for tracking released natural enemies (Geden et al. 1998, Taylor et al. 1997) and cooperative efforts are needed between field ecologists and insect biochemists and molecular biologists to use these tools properly. Molecular methods will also allow identification of parasitoids in the immature stages within host puparia, and even identification of parasitoids after emergence by examination of remains such as meconia and pupal exuviae (Carlson et al. 1999).

We also are still in need of basic research on the natural enemies, including their ability to disperse (Petersen and Pawson 1991, Kaufman et al. 2000b), locate and orient to hosts (Mandeville and Mullens 1990), habitat preferences (Geden 1999), host factors governing reproduction and sex ratios (King 1990), etc. Although predators such as *Hydrotaea aenesecens*, *Macrocheles muscaedomesticae* and *Carcinops pumilio* are known to be important natural mortality factors for flies, very little is known about how to utilize these beneficials as practical management tools. Similarly, although *Hermetia illucen*s often provides natural fly suppression in open sided poultry housing in the Southeast, current rearing technology does not allow practical use of this fly in areas outside its normal range. Other competitors such as litter beetles appear to suppress fly populations at times, but we have no practical way of limiting their destructive activities.

Basic work still is required on the flies themselves. Fly behavior in different production systems needs further investigation. There have been surprisingly few definitive studies on adult fly dispersal, field longevity, and larval ecology. We have only recently quantified such things as adult and larval temperature responses and intraspecific competition for food (Fletcher et al. 1990, Lysyk 1991, Barnard and Geden 1993) to the point where the data are useful for predictive purposes. Even less is known about *Fannia* spp., and this severely limits our management abilities. There is a continuing need to evaluate attractants, repellents and visual targets to manage the flies. (Singh and Singh 1992, Cosse and Baker 1996, Maganga et al. 1996, Gruetzmacher and Nakano 1997, Howard and Wall 1998, Chapman et al. 1998a,b, 1999, Liao 1999).

Modelling efforts have yielded the computer simulation model "Fly Management Simulator" (FMS) (Geden et al. 1990, Wilhoit et al. 1991a,b), which is now in need of validation and refinement for use in different regions of the country. Modelling work should continue, with particular emphasis on adding other organisms (e.g., *Fannia*, *Hydrotaea*, *Alphitobius*, *Hermetia*) to FMS. Fly management ideally should incorporate economic cost and benefit concerns. Some economic aspects, such as costs of on-farm insecticide use, natural enemy purchases and releases, and cultural manipulations, can be quantified. The main difficulty is in determining appropriate tolerance thresholds, for example, among neighbors of a poultry facility. Such thresholds are likely quite variable, and determining them will be a challenge. Indeed, national standards for acceptable fly populations may be impractical or impossible to define. Such thresholds should be developed on a local basis to consider the legal, demographic, social, environmental, and economic parameters that, taken together, define a tolerable level of fly activity.

In summary, the following areas of research in support of fly IPM are recommended. These areas are divided into "basic" research, or research with longer term and possibly high-impact implications for fly management, and "control-component" research, or research that addresses shorter term operational problems and opportunities for fly IPM.

Summary of Basic Research Needs on Flies

1) Basic studies are needed on fly nutritional ecology (including larvae), dispersal behavior, and appetitive orientation behavior that may lead to improved trap designs and allow forecasts of fly emigrations.

2) Biological control:

(a) Devise standards for matching appropriate parasitoids with different target regions and production systems.

(b) Devise quality control standards that allow economical mass-production of parasitoids that are vigorous and free of disease and genetic defects.

(c) Develop rearing and release technology for development of acarine and beetle predators into practical management tools for flies.

(d) Determine mechanisms of host location by parasitoids and the role of imprinting on parasitoid behavior.

(e) Evaluate dispersal behavior of parasitoids in the field.

(f) Develop molecular methods for discrimination of parasitoid biotypes.

(g) International exploration for novel biological control agents.

(h) Study the biology of adult *H. illucens* to close the gaps that prevent insectary production of this competitor.

(i) Determine the role of lesser mealworm in the regulation of flies in caged layer houses.

3) Determine the effects of bird genetics and nutrition on the suitability of manure to support fly development.

4) Toxicological and pathological studies to identify pesticides and pathogens with sublethal effects (e.g., on reproductive physiology) on flies that are compatible with beneficials.

5) Collection of basic data on ecology and biology of *Fannia* spp. to allow incorporation into the Fly Management Simulator (FMS) model developed at N. C. State University.

Summary of Control-Component Research Needs on Flies

1) Field studies of fly orientation behavior, especially attraction to traps and baits.

2) Evaluation of the factors affecting the suitability of manure for fly breeding after it has been applied to land.

3) Biological control:

(a) Incorporation of pathogens and/or *B.t.* subunit toxins into adult fly baits.

(b) Devise standards for assessing the success of parasitoid releases.

(c) Develop dose-response relationships for parasitoid releases that will allow knowledge-based release rate recommendations.

(d) Improve delivery systems for currently available parasitoids, predators and pathogens.

(e) Design mass-rearing methods for production of *Hydrotea.*

(f) Determine the compatibility of *Hydrotea* releases with other biological control agents.

(g) Develop a system for managing *H. illucens* in environmentally controlled layer houses.

4) Evaluation of the influence of different manure removal protocols (e.g., alternate row, leaving a pad) on establishment of beneficial organisms in manure.

5) Economics research to document the scope and costs of the fly problem on poultry

farms.
6) Validation of FMS in the field.

Litter Beetles

Description and Biology. There are several species of "litter beetles" that inhabit poultry droppings and litter. The most important are the lesser mealworm, *Alphitobius diaperinus* (Tenebrionidae), and two species in the dermestid genus *Dermestes*; the hide beetle (*D. maculatus*) and the larder beetle (*D. lardarius*). Other species of beetles that occasionally cause damage to poultry housing are *Dermestes ater, Tenebrio mollitor, Alphitobius laevigatus*, and *Trox* spp.

A. diaperinus is a cosmopolitan species that originated in sub-Saharan Africa. The larvae are yellowish-brown and reach a maximum length of about 8 mm; adults are black (reddish brown right after emergence) and about 5 mm long. Larvae of hide beetles and larder beetles are larger and slower-moving than those of the lesser mealworm. They are densely covered with long thick setae that give the larvae a dusty look and can make them difficult to see in the field. Adult hide and larder beetles also are larger than adult lesser mealworm. Hide beetle adults are uniformly dark brown dorsally with white patterns ventrally. Larder beetle adults are similar in size and shape to hide beetles but have light-colored bands that cross the elytra just behind the pronotum.

Lesser mealworm adults lay their eggs in cracks and crevices in the poultry house, in manure or litter, and in grain hulls (Turner 1986). Larvae hatch and complete development to the adult stage in 40-100 days depending on temperature and food quality (Wilson and Minor 1969, Sarin and Saxena 1973, Rueda and Axtell 1996). The larvae consume spilled feed, manure and, to a lesser extent, dead birds and cracked eggs. Beetle populations in broiler and turkey houses often are concentrated around lines of feeders, which provide the beetles with shelter and an opportunity to feed on spilled bird feed. Mature larvae disperse when they are crowded to find isolated pupation sites, and this behavior is responsible for much of their destructive activity (Ichinose et al. 1980, Geden and Axtell 1987). Crowded larvae leave the litter and tunnel into thermal insulation materials where they construct pupal cells. Polystyrene, polyisocyanurate, and fiberglass insulation are all vulnerable to beetle attack. The tunnels are expanded further when adult beetles eclose and leave the tunnels to find food (Vaughan et al. 1984). The lesser mealworm is nocturnal, with greatest activity of both larvae and adults occurring shortly after dark (Geden and Axtell 1987, Hoffman and Grosse 1987). Populations of lesser mealworm often reach high densities, especially in deep-litter broiler and turkey houses and in high-rise caged layer operations. It is not unusual for the litter of a broiler house to quiver from beetle activity or for 70% of the surface of manure in a high-rise house to be covered with adult beetles.

The habits of hide beetles and larder beetles in poultry operations are similar to those of lesser mealworm with two notable exceptions. First, *Dermestes* spp. thrive on protein rich media such as the carcasses of dead birds and rodents (Hinton 1945, Cloud and Collison 1986). As a result, local populations of these beetles often increase rapidly when new rodent control efforts are initiated. (The use of composting systems for handling dead birds also can aggravate hide beetle problems if the composts are not maintained properly). Second, *Dermestes* spp. are able to tunnel into wooden structures as well as soft thermal insulation materials (Stafford et al. 1988, Axtell and Arends 1990). The resulting damage to building support posts, beams, and plywood paneling can be severe and costly.

Economic Importance. The pest status of lesser mealworm arises from three causes. First, the beetles are known to harbor many important avian pathogens and parasites including *Salmonella typhimurium*, *Escherichia coli*, tapeworms, *Eimeria*, coronaviruses,and avian leukosis virus (Avancini and Ueta 1990, Axtell and Arends 1990, Despins and Axtell 1994, Despins et al. 1994, Davis and Wray 1995, Davis et al. 1996, Goodwin and Waltman 1996, McAllister et al. 1994, 1995, 1996,, Watson et al. 2000). This is of particular concern in floor-litter systems such as broiler, breeder, and turkey housing where the birds can consume large quantities of beetles, especially in the first week or so after placement. Consumption of large numbers of beetles also can have direct adverse effects on the health and performance of these young birds (Despins and Axtell 1994, 1995). Second, adult beetles cause public nuisance problems by invading the homes and businesses of neighbors. These problems are particularly acute during house cleanouts, when large numbers of beetles migrate from the fields on which infested litter is spread (Turner 1986, Schmitz and Wohlgemuth 1988). Third, the destruction of thermal insulation materials by beetle larvae results in increased energy consumption and costly replacement of the insulation (LeTorc'h and Retenneur 1983, Ichinose et al. 1980, Vaughan et al. 1984).

Assigning real-dollar losses to this damage requires extrapolation from a limited number of sources, yet such estimates may be useful for making rough comparison of beetles with other pests. Energy costs in beetle-damaged broiler houses are reported to be 67% higher than in buildings without damage; this represents increased annual operating costs of $4,000 per infested house under Georgia conditions (Anonymous 1990). Replacement of beetle-damaged insulation in a 50' x 350' high-rise caged layer house was estimated at $20,000 in 1982. In 1980 it was estimated that annual beetle losses to the Virginia poultry industry totalled $1.1 million for turkeys, $3.3 million for broilers, and $11.5 million for the layer industry (Turner 1986). Annual losses to beetle damage have been estimated at $12 million for Georgia (Sheppard and Noblet 1999). Some of these estimates do not include control costs or production losses due to disease and poor temperature control.

Lesser mealworm populations in high-rise caged layer houses also perform beneficial functions. Intense beetle activity helps to aerate and dry the manure, and the beetles are facultative predators of fly immatures. Although they are not particularly voracious predators on an individual basis, (Despins et al. 1988) high populations of beetles appear to suppress house fly populations in some cases (Wallace et al. 1985).

Current Control and Monitoring Methods

Cultural Control. Cold weather is a simple and inexpensive cultural control practice for producers in northern locations. Most beetles can be eliminated from poultry housing by opening the buildings and exposing them to sub-freezing temperatures for a week or more. In broiler and turkey houses, complete removal of litter and replacement with fresh shavings greatly reduces beetle populations compared with the practice of top-dressing old litter with fresh shavings between flocks. Frequent removal of manure from caged-layer houses eliminates beetle problems. Some types of insulation have proven to be more resistant to beetle penetration than others, although none is 100% refractory. Mechanical barriers can be used to prevent the beetles from reaching susceptible building construction matierals (Geden and Carlson 2001).

Biological Control. No biological control options are currently available for beetles. Several natural enemies of lesser mealworm have been found, including protozoa (*Farinocystus tribolii*), fungi (*Beauveria bassiana*), and a parasitic mite (*Acarophenax mahunkai*) (Steinkraus and Cross 1993, Steinkraus et al. 1991, 1992). Of these, *B. bassiana* holds the greatest promise for further development as a biological control agent (Crawford et al. 1998, Geden et al. 1998). Steinernematid nematodes (*Steinernema feltiae*) do not appear to provide long-term beetle control, but additional testing may reveal more effective species or strains (Geden et al. 1985, 1987). Some strains of *Bacillus thuringiensis* produce endotoxins that may have high activity against beetles, but much work remains before any such microbial product is commercially available.

Chemical Control. Several formulations of carbaryl are registered for use against lesser mealworm, including dusts, wettable powders, sprayable liquids, and baits. Products containing tetrachlorvinphos are available as dusts, wettable powders, or emulsifiable concentrates; this toxicant also is formulated in a combination product with dichlorvos. Cyfluthrin, permethrin and some other pyrethroids also are labeled as a premise treatment to protect poultry building structures from litter beetles. Boric acid is available as a soil and premise treatments for litter beetles in some states.

None of the available insecticides provide satisfactory control of beetle populations when at outbreak levels. An aggressive management program involving litter and premise treatments at cleanout followed by regular monitoring and baiting can provide partial suppression of beetle populations in broiler and turkey houses. Population management in high-rise caged-layer houses is nearly impossible if conditions are favorable for the beetles. In such instances producers have little choice but to rely on premise treatments as building protectants. Although beetles are vulnerable to many residual insecticides (Geden et al. 1987b, Vaughan and Turner 1984, Cloud and Collison 1985) the effectiveness of any premise treatment in the field is limited by the rapid accumulation of dust on treated surfaces in poultry houses (Despins et al. 1991).

Monitoring. Active beetle populations in the manure or litter can be sampled by the use of tube traps or by collecting manure samples and extracting beetles through Berlese funnels (Safrit and Axtell 1984, Stafford et al. 1988). Tube traps are sections of PVC pipe with rolled corrugated cardboard inserts; beetle adults and larvae enter the cardboard to moult and oviposit and the number of beetles present in the traps is typically counted on a weekly basis. The traps are simple to use and provide valuable data on relative beetle population sizes over time. One disadvantage of tube traps is a tendency to underestimate young larvae, which is important for characterization of population age structure. A second disadvantage is that tube traps become "saturated" with beetles when population densities are only moderately high. Because of this saturation effect, traps lose accuracy and sensitivity at high beetle densities. Research on beetle attractants and pheromones could lead to more sensitive monitoring methods (Levinson et al. 1981,Rakowski 1988, Rakowski et al. 1982, 1986, 1989).

Building premises can be surveyed by counting larvae on walls and posts in the early evening (Geden and Axtell 1987). "Sentinel" insulation also can be placed on walls and posts and monitored for entry holes on a regular basis. For greater sensitivity, "flat traps" can be used by stapling sections of corrugated cardboard to walls and posts; the cardboard is replaced and the beetles counted on a weekly basis. There is an urgent need to develop an understanding of the relationship between beetle densities in the manure or litter and the relative degree of risk to flock health and buildings.

Summary of Basic Research Needs for Litter Beetles

1) Identification and testing of aggregation and sex pheromones of lesser mealworm that could lead to improved monitoring methods and the development of more effective baits.
2) Surveys of beetle populations should be conducted in the US and the original home range of lesser mealworm to discover novel biological control agents.
3) A simulation model for litter beetles should be developed. To accomplish this, data are needed on some critical aspects of beetle life history, including temperature-dependent survival and development, nutritional effects on beetle fecundity, effects of crowding on beetle reproduction and survival, and factors affecting longevity and dispersal behavior of adults.
4) More information is needed on field ecology of litter beetles. For example, lesser mealworm is generally credited with promoting manure drying in caged layer houses, but there are no data to support this. Data are needed to characterize the relationship between beetle abundance, manure moisture and house fly density.
5) Research is needed to establish whether ingestion of beetles by young broilers and turkeys has deleterious effects on bird growth and feed conversion efficiency.

Summary of Control-component Research Needs for Litter Beetles

1) Better economic injury information is needed, as is the development of thresholds based on standardized sampling methods for each type of beetle, production system and type of housing.
2) Research is needed to determine whether any of the known natural enemies of litter beetles can be developed into practical management tools for use in IPM programs.
3) Although beetle adults are known to fly toward lights at night, nothing is known about the relative attractiveness of different light sources. Light traps could be used to manage beetle dispersal among poultry houses and to attract beetles near fields where infested litter is spread.
4) Barriers should be evaluated to determine whether beetles can be prevented from climbing out of the manure or litter and into vulnerable building materials.

5) The fate of beetles in fields where infested litter is spread needs to be determined, including survival times and dispersal distances under different weather conditions.

Northern Fowl Mite

Description and Biology. The entire life cycle of the Northern Fowl Mite (NFM), *Ornithonyssus sylviarum*, occurs on the host (Sikes and Chamberlain 1954, Loomis 1978); however, oviposition may occur in the nest of the host (Cameron 1938). At times, mites leave the host in large numbers and aggregate on cage structures and eggs (DeVaney 1986a). Even though the NFM has long been considered a winter pest (Loomis 1978), mites have been found on chickens year round (Kirkwood 1963, 1968), and will come out to the tips of the feathers in hot weather (Cameron 1938). When separated from the host, *O. sylviarum* will live from 2 to 4 weeks (Cameron 1938, Baker et al. 1956, Kirkwood 1963, Loomis 1978), compared with 34 weeks for the chicken mite, *Dermanyssus gallinae* (Kirkwood 1963).

The area on the host most preferred by the NFM is the vent region but in severe infestations, mites can be found over the entire body (Cameron 1938, Anonymous 1959, Loomis et al. 1970, Lemke et al. 1988). Cameron (1938) seldom found mites on young birds. Kirkwood (1968) also found this to be true and suggested that it may be due to lack of contour feathers. He and others (Payne 1930, Cameron 1938, Hansens 1951, Abasa 1965) stated that roosters have more mites than hens, possibly due to differences in plumage in the vent area. Males have more contour feathers near the vent, whereas female plumage in this area is characterized by proportially greater amounts of down. Feathers are preferred over down by *O. sylviarum* (Kirkwood 1968), and population reduction has been demonstrated by clipping feathers in the vent area (DeVaney 1986b).

Mites transfer from bird to bird, and populations rapidly rise and decline, but some birds remain entirely free of mites (Cameron 1938). Hall and Gross (1975) found that roosters with high levels of plasma corticosterone that were maintained at high levels of social stress had lower mite populations than when the conditions were reversed. Inherited levels of corticosterone had more effect on mites than did stress alone. It also was found that hens subjected to higher social stress had significantly lower mite populations than unstressed hens (Hall et al. 1978b, Turner 1978, Arthur and Axtell 1983). Additional experiments indicated that although hens first coming into production are most susceptible to NFM infestation, estrogen alone probably is not responsible for the difference in mite susceptibility between hens and roosters (Hall et al. 1978a).

Foulk (1964) found four major routes of infestation in layer flocks: 1) infested hatcheries and contract started-pullet farms; 2) infested trucks and crates used to carry infested birds; 3) infested personnel, equipment, or egg crates; and, 4) infested wild birds that enter poultry houses. Because the NFM has been found on the Norway rat and the house mouse, both of which can be common inhabitants or poultry houses (Hall & Turner 1976, Miller & Price 1977), it has been assumed that these rodents may aid in mite dissemination. Under current poultry production conditions, DeVaney (1986a) considers the most frequent method of dissemination to be via personnel or equipment.

Economic Importance. The NFM is an obligate hematophagous parasite of domestic and feral birds, and maintains a high ranking world wide as one of the major pests of poultry. Millions of dollars each year are lost from decreases in egg production and acaricide treatment costs. Excessive NFM populations can cause anemia and death in chickens, and mites seen crawling on eggs make workers reluctant to enter poultry houses or handle the eggs. Although NFM's have not been shown to use humans as hosts, short-term exposure to live or dead mites can produce symptomology ranging from a mild pruritus to severe allergic reactions.

Several economic studies have been done, but the results are equivocal. Combs et al. (1976) demonstrated that chemical removal of mites improved egg production. Arends et al. (1984) reported decreased egg production and feed efficiency in broiler breeder flocks caused by NFM's. However, neither Loomis et al. (1970) nor Hogsette (1979) could find significant differences in egg production due to mite populations. Bramhall (1972) discounted NFM's as a reason for reduced egg production and suggests that producers control mites only to prevent discomfort to workers. Eleazer (1978) reported that uncontrolled NFM infestations did not cause reduced egg production, and DeVaney (1979) found that during two separate 1-year trials, a significant reduction in egg production was produced by mites for only 1 month in one trial, and 2 months in the other. Internal quality of eggs is not affected by the presence of NFM's on hens (DeVaney 1981b). Lyon (1975) estimated that the NFM could be costing the poultry industry $80 million annually. DeVaney (1978) estimated an annual loss of $66 million due to external parasites causing decreases in egg production; parasite prevention might cost as much as $1.1 million. In Florida, Butler (1985) attributed a $3.4 million loss in poultry profits to the NFM in 1984.

Current Control and Monitoring Methods

Cultural Control. Wild birds have long been considered a source of reinfestation of poultry flocks by NFM (Hartman 1954). Although they continue to pose a problem where open housing is in use, the conversion to closed housing for most laying flocks has eliminated much of the problem. DeVaney (1986a) considered the most frequent method of dissemination to be via personnel or equipment, but the industry has overcome many of these problems by changes in management. During the past 15 years, the poultry industry has increased the level of cultural control of NFM by sanitizing filler flats and egg cases and by eliminating mite populations in pullet flocks before they are moved to layer houses. Different individuals and perhaps strains of birds show varying levels of susceptibility to the mites (Hogsette 1979), but little has been done to isolate the resistance mechanisms involved or to determine the genetic compatability of these resistance traits with other traits that have higher priority in breeding programs for commercial lines.

Biological Control. No natural enemies of NFM are known at this time. *Bacillus thuringiensis* exotoxins are effective against NFM (Mullens et al. 1988), but concerns over avian toxicity will probably limit their use.

Chemical Control. Chemical control is the only method available for controlling mite infestations once they have become established. The registered pesticides most commonly used for NFM control are carbaryl, malathion, tetrachlorvinphos, dichlorvos, tetrachlorvinphos-dichlorvos mixtures, and the pyrethroids. Research is needed on field application of miticides on poultry, as application methods can have a pronounced effect on efficacy (Arthur and Axtell 1982, Levot 1992). Poultry producers report widespread mite resistance to labeled miticides, but many of the resistance problems are caused by inadequate application methods (Eleazer 1978, Hogsette 1979; Hall et al. 1980, Hall et al. 1984a,b). Whenever possible, researchers should use a standard method for assessing resistance so that valid comparisons can be made between studies (Arthur and Axtell 1983, Fletcher and Axtell 1991). Diflubenzuron, MK-933, Avermectin B1, bacitracin, zinc bacitracin, all registered anticoccidials, and compounds showing systemic activity in other animals have been tested for systemic activity against NFM, but no activity has yet been found (DeVaney 1981a, 1984, DeVaney and Kubena 1982, DeVaney and Ivie 1980, 1984a,b). Some light oils or soaps kill NFM by suffocation, but associated air sac problems may preclude use on birds (McKeen et al. 1983). Similar data on the effectiveness of chemical control also are needed for other external parasites of poultry such as bed bugs, lice, and chicken mites (Fletcher and Axtell 1991, 1993).

Monitoring. Economic injury levels must be evaluated in the field by examining chickens and estimating the NFM populations. In the past, the two most common ways that producers were alerted to the presence of NFM were the presence of mites on eggs and complaints by employees working in the houses. These situations have been virtually eliminated by the use of automated equipment and a reduction in employees to one person per house (~100,000 hens). This leaves detection by direct examination of hens, something that is not routinely done.

A number of evaluations systems have been devised, and all involve visual estimation of NFM populations (Hall et al. 1978b). In large poultry houses with >100,000 birds, at least 50 should be examined (Drummond et al. 1976). This is a very time-consuming process, although sequential sampling models can streamline the process (Harris et al. 2000). High-level infestations can be monitored by counting mites on hen eggs (Mullens et al. 2000). Practical and accurate detection and sampling methods need to be developed for NFM, perhaps using infrared or other remote sensing technology.

Summary of Basic Research Needs for Northern Fowl Mite

1) More accurate and practical monitoring methods should be developed, perhaps using remote sensing, imaging and telemetric technologies.
2) More studies are needed on basic biology, ecology and population dynamics of the mites. We need to know why mite populations seem to appear suddenly and disappear just as fast. Similarly, we need to know why some birds are adversely affected and some are not. Ideally, this information should be used to develop a model that could be incorporated into a larger model of poultry pest management.
3) Better data are needed about the economic effects of NFM on laying hens, especially in modern facilities with new genetic strains of birds.

4) Dispersal studies are needed to establish the role of rodents and wild birds in the movement of NFM.

Summary of Control-component Research Needs for Northern Fowl Mite
1) The status of mite resistance to registered pesticides needs to be updated using standardized methods.
2) Application equipment for modern laying house cage designs needs to be developed and tested for efficacy.

Other Pests

The emphasis of this report has been on the three pests that cause the most damage to poultry overall; flies, litter beetles, and northern fowl mites. Other arthropod pests can cause very serious problems from time to time, especially lice and the "poultry premise ectoparasites"; red mites and bed bugs. Mite and bed bug problems occur infrequently but can be severe, especially in breeder flocks, and they are notoriously difficult to control. Although research funding is in short supply, there remains a critical need to study these occasional pests when opportunities arise and to share information on their biology and management with others. The proposed Internet site would be a logical place for such discussions to occur.

REFERENCES CITED

Arends, J. J. & S. M. Stringham. 1992. Poultry pest management. N. C. Coop. Ext. Serv. Pub. AG-474. 30 pp.

Arends, J. J., S. H. Robertson & C. S. Payne. 1984. Impact of northern fowl mite on broiler breeder flocks in North Carolina. Poult. Sci. 63: 1457-1461.

Arthur, F. H. & R. C. Axtell. 1982. Comparisons of permethrin formulations and application methods for northern fowl mite control on caged laying hens. Poult. Sci. 61: 4879-884.

Arthur, F. H. & R. C. Axtell. 1983a. Northern fowl mite population development on laying hens caged at three colony sizes. Poult. Sci. 62: 424-427.

Arthur, F. H. & R. C. Axtell. 1983b. Susceptibility of northern fowl mites in North Carolina to five acaricides. Poultry Sci. 62: 428-432.

Avancini, R. M. P. & M. T. Ueta. 1990. Manure breeding insects (Diptera and Coleoptera) responsible for cestoidosis in caged layer hens. J. Appl. Entomol. 110 :301-332.

Axtell, R. C. 1999. Poultry integrated pest management: status and future. Integrated Pest Management Reviews 4: 53-73.

Axtell, R. C. & J. J. Arends. 1990. Ecology and management of arthropod pests of poultry. Ann. Rev. Entomol. 35:101-126.

Baker, E. W., T. M. Evans, D. J. Gould, W. B. Hull & H. L. Keegan. 1956. A manual of parasitic mites of medical or economic importance. Natl. Pest Control Assn., Inc., N. Y.

Barnard, D. R. & C. J. Geden. 1993. Influence of larval density and temperature in poultry manure on development of the house fly (Diptera: Muscidae). Environ. Entomol. 22: 971-977.

Barnard, D. R. & R. H. Harms. 1992. Growth and survival of house flies (Diptera: Muscidae) in response to selected physical and chemical properties of manure. J. Econ. Entomol. 85: 1213-1217.

Barson, G., N. Renn. and A.F. Baywater. 1994. Laboratory evaluation of six species of entomopathogenic fungi for the control of the house fly (*Musca domestica* L.), a pest of intensive animal units. J. Invertebr. Pathol. 64: 107-113.

Becnel, J. J. & C. J. Geden. 1994. Description of a new species of microsporidia from *Muscidifurax raptor* (Hymenoptera: Pteromalidae), a pupal parasitoid of muscoid flies. J. Eukaryotic Microbiol.41:236-243.

Bradley, S. W. & D. C. Sheppard. 1984. House fly oviposition inhibition by larvae of *Hermetia illucens,* the black soldier fly. J. Chem. Ecol. 10: 853-859.

Bramhall, E. L. 1972. When should you spray for fowl mites: Poult. Dig. Dec.: 596.

Braverman Y.,A. Chizov-Ginzburg, A. Saran, and M.Winkler.1999. The role of houseflies (*Musca domestica*) in harbouring *Corynebacterium pseudotuberculosis* in dairy herds in Israel. Rev Sci Tech. 18(3):681-90.

Burg, J. G. & R. C. Axtell. 1984. Monitoring house fly , *Musca domestica* (Diptera: Muscidae), populations in caged-layer poultry houses using a baited jug trap. Environ. Entomol. 13: 1083-1090.

Butler, J. F. 1985. Estimated losses in Florida in 1984 due to arthropod damage, pp. 1-7. In Vet. Entomol. Ann. Sum. Res. Report for 1984, State Project EY 1773, Univ. of Fla, Gainesville.

Cameron, D. 1938. The northern fowl mite (*Liponyssus sylviarum* C. & F., 1877). Can. J. Res., Sect. D. 16: 230-254.

Carlson, D. A. C. J. Geden, and U. R. Bernier. 1999. Identification of pupal exuviae of *Nasonia vitripennis* and *Muscidifurax raptorellus* parasitoids using cuticular hydrocarbons. Biol. Control. 15: 97-106.

Carswell, I., R. Spooner-Hart, amd R. J. Milner. 1998. Laboratory susceptibility of *Musca domestica* L. (Diptera: Muscidae) and *Bactrocera tryoni* (Diptera: Tephritidae) to an isolate of *Metarhizium anisopliae* (Metsch) Sorokin. Australian J. Entomol. 37: 281-284.

Chapman, J. W., P. E. Howse, J. J. Knapp, and D. Goulson. 1998. Evaluation of three (Z)-9-tricosene formulations for control of *Musca domestica* (Diptera: Muscidae) in caged-layer poultry units. J. Econ. Entomol. 91: 915-922.

Chapman, J. W., P. E. Howse, J. J. Knapp, and D. Goulson. 1998. An evaluation of (Z)-9-tricosene and food odors for attracting house flies, *Musca domestica*, to baited targets in deep-pit poultry units. Entomol. Exper. Appl. 89: 183-192.

Chapman, J. W, J. J. Knapp and D. Goulson. 1999. Visual responses of *Musca domestica* to pheromone impregnated targets in poultry units. Med. Vet. Entomol. 13: 132-138.

Cloud, J. A. & C. H. Collison. 1985. Laboratory evaluation of insecticides for control of adult and larval hide beetles, *Dermestes maculatus* Degeer, from poultry houses. J. Agr. Entomol. 2:297-308.

Cloud, J. A. & C. H. Collison. 1986. Comparison of various poultry house litter components for hide beetle (*Dermestes maculatus* DeGeer) larval development in the laboratory. Poultry Sci. 65: 1911-1914.

Cohen, D., M. Green, C. Block, R. Slepon, R. Ambar, S. Wasserman, and M. M. Levine. 1991. Reduction of transmission of shigellosis by control of houseflies (*Musca domestica*). Lancet 337: 993-997.

Coler, R. R. and K. B. Nguyen. 1994. *Paraiotonchium muscadomesticae* n. sp. (Tylenchida: Iotonchiidae) a parasite of the house fly (*Musca domestica*) in Brazil and a key to species of the genus *Paraiotonchium*. J. Nematol. 26: 392 401.

Combs, R. L., Jr., J. V. Robinson & K. Waits. 1976. Evaluation of chlordimeform 95%SP, Rabon 24% EC as agents for control of the northern fowl mite, *O. sylviarum* on caged layers. Miss. Agric. & Forestry Exp. Sta.

Cosse, A. A. and T. C. Baker. 1996. House flies and pig manure volatiles: Wind tunnel behavioral studies and electrophysiological evaluations. J. Agric. Entomol. 13: 301-317.

Crawford, P. J., W. M. Brooks, and J. J. Arends. 1998. Efficacy of field-Isolated strains of *Beauveria bassiana* (Moniliales: Moniliaceae) as microbial control agents of the lesser mealworm (Coleoptera: Tenebrionidae). J. Econ. Entomol. 91: 1295-1301.

Davies R. H. and C. Wray 1995. Contribution of the lesser mealworm beetle (*Alphitobius diaperinus*) to carriage of *Salmonella enteritidis* in poultry. Vet. Rec. 137(16):407-8.

Davis, J. F., A. E. Castro, J.C. de la Torre, H. J. Barnes, J. T. Doman, M. Metz, H. Lu, S. Yuen, P. A. Dunn, and M. N. Teng, MN. 1996. Experimental reproduction of severe hypoglycemia and spiking mortality syndrome using field-derived and embryo-passaged preparations. Avian Dis. 40: 158-172

Despins J. L. and Axtell R. C. 1994. Feeding behavior and growth of turkey poults fed larvae of the darkling beetle, *Alphitobius diaperinus*. Poult Sci 73:1526-33

Despins JL, and R. C. Axtell RC. 1995. Feeding behavior and growth of broiler chicks fed larvae of the darkling beetle, *Alphitobius diaperinus*. Poult Sci 74:331-6

Despins, J. L., E. C. Turner, Jr. & D. G. Pfeiffer. 1991. Evaluation of methods to protect poultry house insulation from infestations by lesser mealworm (Coleoptera: Tenebrionidae). J. Agr. Entomol. 8: 209-217.

Despins, J. L., R. C. Axtell,D. A. Rives, J. S. Guy & M. D. Ficken. 1994. Transmission of enteric pathogens of turkeys by darkling beetle larva (*Alphitobius diaperinus*) J. Appl. Poultry Res. 3: 1-5.

DeVaney, J. A. 1981a. Activity of selected anticoccidials as feed additives for control of the northern fowl mite, *Ornithonyssus sylviarum* (Canestrini and Fanzago). Poultry Sci. 60: 2033-2036.

DeVaney, J. A. 1981b. Effects of the northern fowl mite, *Ornithonyssus sylviarum* (Canestrini and Fanzago), on egg quality of White Leghorn hens. Poultry Sci. 60: 2200-2202.

DeVaney, J. A. 1984. Evaluation of bacitracin and zinc bacitracin as a feed additive for control of the northern fowl mite. Insectic. Acaric. Tests. 9: 434.

DeVaney, J. A. 1986a. Symposium: Arthropods of economic importance to the poultry industry. Ectoparasites. Poultry Sci. 65: 649-656.

DeVaney, J. A. 1986b. Effects of different feather lengths in the vent area of White Leghorn hens on northern fowl mite populations. Poultry Sci. 65: 452-456.

DeVaney, J. A. & G. W. Ivie. 1980. Systemic activity of coumaphos, famphur, crufomate, ronnel, and phosmet given orally to hens for control of the northern fowl mite, *Ornithonyssus sylviarum* (Canestrini & Fanzago). Poultry Sci. 59: 1208-1210.

DeVaney, J. A. & G. W. Ivie. 1984a Evaluation of MK-933 and avermectin B1 for control of northern fowl mites, 1979-1982. Insectic. Acaric. Tests. 9: 433.

DeVaney, J. A. & G. W. Ivie. 1984b. Evaluation of Closantel as a feed additive for control of northern fowl mites, 1983. Insectic. Acaric. Tests. 7 :435.

DeVaney, J. A. & L. F. Kubena. 1982. Evaluation of diflubenzuron as a feed additive for control of northern fowl mites. Insectic. Acaric. Tests. 7: 247.

Doiz O., A. Clavel, S.Morales, M. Varea, F. J. Castillo, C. Rubio C; R. Gomez-Lus. 2000. House fly (*Musca domestica*) as a transport vector of *Giardia lamblia*. Folia Parasitol (Praha) 47(4):330-1.

Drummond, R. O., J. L. Lancaster, Jr. & P. D. Ludwig. 1976. Appendix for guidelines for registering pesticides for the control of arthropod parasites of livestock, poultry, fur and wool bearing mammals. EPA.

Eleazer, T. H. 1978. Fowl mites found not resistant to insecticides. Poultry Dig. 37: 538.

Emerson, P. M., S. W. Linday, G. E. L. Walraven, H. Faal., C. Bough, K. Lowe, and R. L. Bailey. 1999. Effect of fly control on trachoma and diarrhoea. Lancet 353: 1401-1403.

Farkas, R. J. A. Hogsette and L. Borzonyi. 1998. Development of *Hydrotaea aemescens* (Weideman) and *Musca domestica* L. (Diptera: Muscidae) in poultry and pig manure of different moisture content. Environ. Entomol. 27: 695-699.

Fotedar R. 2001. Vector potential of houseflies (*Musca domestica*) in the transmission of *Vibrio cholerae* in India. Acta Trop. 78(1):31-4.

Fischer, O., L Matlova, L. Dvorska, P. Svastova, J. Bartl, I. Melicharek, R. T. Weston and I. Pavlic. 2001. Diptera as vectors of mycobacterial infections in cattle and pigs. Med. Vet. Entomol. 15:208-211.

Fletcher, M. G. & R. C. Axtell. 1991. Susceptibility of northern fowl mite, *Ornithonyssus sylviarum* (Acarina: Macronussidae), and chicken mite, *Dermanyssus gallinae* (Acarina: Dermanyssidae), to selected acaricides. Exp. Appl. Acarol. 13: 137 142.

Fletcher, M. G. & R. C. Axtell. 1993. Susceptibility of the bedbug, *Cimex lectularius*, to selected insecticides and various treated surfaces. Med. Vet. Entomol. 7: 79 92.

Fletcher, M. G., R. C. Axtell & R. E. Stinner. 1990. Longevity and fecundity of *Musca domestica* (Diptera: Muscidae) as a function of temperature. J. Med. Entomol. 27: 922-926.

Foulk, J. D. 1964. Dissemination of the northern fowl mite, *Ornithonyssus sylviarum* (Acarina: Mesostigmata) and experiments on its control. Ph.D. dissertation, Cornell University, Ithaca.

Geden, C. J. 1990. The role of coleopteran and acarine predators in house fly population regulation in poultry production facilities. Pp. 177-200 in: Biocontrol of Arthropods Affecting Livestock and Poultry, D. A. Rutz and R. A. Patterson (eds). Westview Press, Boulder.

Geden, C. J. 1996. Modeling host attacks and progeny production of *Spalangia gemina, S. cameroni*, and *Muscidifurax raptor* under contstant and variable temperatures. Biol. Control 7: 172-178.

Geden, C. J. 1997a. Development models of the fly parasitoids *Spalangia gemina, S. cameroni*, and *Muscidifurax raptor* under contstant and variable temperatures. Biol. Control 9: 185-192.

Geden, C. J. 1997b. Evaluation of *Paraiotonchium muscaedomesticae* (Nematoda: Tylenchida: Iotonchiidae), a potential biological control agent of the house fly (Diptera: Muscidae). Biol. Control 10: 42-47.

Geden, C. J. 1999. Host location by house fly (Diptera: Muscidae) parasitoids in poultry manure at differerent moisture levels and host densities. Environ. Entomol. 28: 755-760.

Geden, C. J. & R. C. Axtell. 1987. Factors affecting climbing and tunneling behavior of the lesser mealworm, *Alphitobius diaperinus* (Coleoptera: Tenebrionidae). J. Econ. Entomol. 80: 1197-1204.

Geden, C. J. and D. A. Carlson. 2001. Mechanical barrier for preventing climbing by lesser mealworm (Coleoptera: Tenebrionidae) and hide beetle (Coleoptera: Dermestidae) larvae in poultry houses. J. Econ. Entomol. 94: 1610-1616.

Geden, C. J., R. C. Axtell & W. M. Brooks. 1985. Susceptibility of the lesser mealworm, *Alphitobius diaperinus* (Coleoptera: Tenebrionidae) to the entomogenous nematodes *Steinernema feltiae, S. glaseri* (Steinernematidae) and *Heterorhabditis heliothidis* (Heterorhabditidae). J. Entomol. Sci. 20: 331 339.

Geden, C. J., J. J. Arends & R. C. Axtell. 1987a. Field trials of *Steinernema feltiae* (Nematoda: Steinernematidae) for control of *Alphitobius diaperinus* (Ceoleoptera: Tenebrionidae) in commercial broiler and turkey houses. J. Econ. Entomol. 80: 136-141.

Geden, C. J., T. D. Edwards, J. J. Arends & R. C. Axtell. 1987b. Efficacies of mixtures of insecticides and disinfectants. Poultry Sci. 66: 659-665.

Geden, C. J., R. E. Stinner, D. A. Kramer & R. C. Axtell. 1990. MACMOD: a simulation model for *Macrocheles muscaedomesticae* (Acari: Macrochelidae) population dynamocs and rates of predation on immature house flies (Diptera: Muscidae). Environ. Entomol. 19: 578-586.

Geden, C. J., L. Smith & D. A. Rutz. 1992a. Rapid deterioration of searching behavior, host destruction, and fecundity of the parasitoid *Muscidifurax raptor* (Hymenoptera: Pteromalidae) in culture. Annals, Entomol. Soc. Amer. 85: 179 187.

Geden, C. J., D. C. Steinkraus, R. W. Miller & D. A. Rutz. 1992b. Suppression of house flies on New York and Maryland dairies using *Muscidifurax raptor* in an integrated management program. Environ. Entomol. 21: 1419-1426.

Geden, C. J., D. C. Steinkraus & D. A. Rutz. 1993. Evaluation of two methods for release of *Entomophthora muscae* (Entomophthorales: Entomophthoraceae) to infect house flies (Diptera: Muscidae) on dairy farms. Environ. Entomol. 20: 1201-1208.

Geden, C. J., S. J. Long, D. A. Rutz & J. J. Becnel. 1995. *Nosema* disease of the parasitoid *Muscidifurax raptor* (Hymenoptera: Pteromalidae): prevalence, patterns of transmission, management, and impact. Biol. Control. 5: 607-614.

Geden, C. J., D. C. Steinkraus & D. A. Rutz. 1995. Virulence of different isolates and f*ormulations of Beauveria bassiana for house flies and the parasitoid Muscidifurax raptor*. Biol. Control 5:615-621.

Geden, C. J., J. J. Arends, D. A. Rutz, and D. C. Steinkaus. 1998. Laboratory evaluation of *Beauveria bassiana* (Monilales: Moniliaceae) against the lesser mealworm, *Alphitobius diaperinus* (Coleoptera: Tenebrionidae), in poultry litter, soil and a pupal trap. Biol. Control 13: 71-77.

Geden, C. J., J. A. Hogsette, and R. D. Jacobs. 1999. Effect of airflow on house fly (Diptera: Muscidae) distribution in poultry houses. J. Econ. Entomol. 92:416 420.

Geden, C. J., U. R. Bernier, D. A. Carlson, and B. D. Sutton. 1998. Identification of *Muscidifurax* spp., parasitoids of muscoid flies, by composition patterns of cuticular hydrocarbons. Biol. Contro. 12: 200-207.

Goodwin, M. A. and W. D. Waltman. 1996. Transmission of *Eimeria*, viruses, and bacteria to chicks: darkling beetles (*Alphitobius diaperinus*) as vectors of pathogens. J Appl. Poultry Res. 5: 51-55.

Graczyk, T. K., M. R. Cranfield, R. Fayer and H. Bixler.1999. House flies (*Musca domestica*) as transport hosts of *Cryptosporidium parvum*. Am. J. Trop. Med. Hyg. 613: 500-504.

Gruebel, P, J. S. Hoffma, F. K. Chong, N. A. Burstein, C. Mepani, and D. R. Cave. 1997 Vector potential of houseflies (*Musca domestica*) for *Helicobacter pylori*. J Clin Microbiol 35:1300-1303.

Gruetzmacher, D. D. and O. Nakano. 1997. Behavior of house fly, *Musca domestica* L. in relation to transparent plastic bags with water. An. Soc. Entomol. Brasil 26: 455-461.

Hall, R. D. & W. B. Gross. 1975. Effect of social stress and inherited plasma corticosterone levels in chickens on populations of the northern fowl mite, *Ornithonyssus sylviarum*. J. Parasitol. 61: 1096-1100.

Hall, R. D. & E. C. Turner, Jr. 1976. The northern fowl mite collected from rats in a chicken house. J. Med. Entomol. 13: 22-223.

Hall, R. D., W. B. Gross & E. C. Turner, Jr. 1978a. Preliminary observations on northern fowl mite infestations on estrogenized roosters and in relation to initial egg production in hens. Poult. Sci. 57: 1088-1090.

Hall, R. D., E. C. Turner, Jr. & W. B. Gross. 1978b. Effect of cage densities on northern fowl mite populations in commercial caged layer operations. Poultry Sci. 57: 564-566.

Hall, R. D., L. M. English, J. M. Vandepopuliere, K. E. Doisy, J. J. Lyons, M. C. Foehse & D. R. Berkebile. 1980. Chickens, evaluation of standard acaricides for control of northern fowl mites. Insectic. Acaric. Tests. 5: 226-227.

Hall, R. D., J. M. Vandepopuliere, F. J. Fischer, J. J. Lyons & J. D. Van Horn. 1984a. A new in-cage treatment system for control of northern fowl mites on laying hens. Poultry Sci. 63: 628-632.

Hall, R. D., J. M. Vandepopuliere, W. Jaynes & F. J. Fischer. 1984b. Prophylactic efficiency and longevity of polyvinyl chloride strips containing permethrin for control of northern fowl mites (Acari: Macronyssidae) on caged chickens. J. Econ. Entomol. 77: 1224-1228.

Hansens, E. J. 1951. Recent developments in the control of insect and parasitic pests. Amer. Vet. Med. Assoc. J. 118: 39.

Harrington, L. C. & R. C. Axtell. 1994. Comparisons of sampling methods and seasonal abundance of *Drosophila repleta* in caged-layer poultry houses. Med. Vet. Entomol. 8: (in press).

Harris, M. A., M. J. Brewer, J. A. Meyer. 2000. Presence-absence sequential sampling plan for northern fowl mite, *Ornithonyssus sylviarum* (Acari: Macronyssidae), on caged-layer hens. J. of Econ. Entomol. 93: 544-549.

Hartman, R. C. 1954. Keeping chickens in cages, 5th ed. Author Commercial Press, Redlands, Calif.

Hinton, H. E. 1945. A monograph of the beetles associated with stored products. Vol. 1. Br. Mus. Nat. Hist.

Hoffman, K. & W. R. Grosse. 1987. Studies on the circadian locomotor activity of *Alphitobius diaperinu*s (Panzer, 1797). Wiss Z Martin Luther Univ. Halle Wittenberg Math-Naturwiss Reihe 36(3): 84-91 (in German).

Hogsette, J. A. 1979. The evaluation of poultry pest management techniques in Florida poultry houses. Ph.D. dissertation, University of Florida, Gainesville.

Hogsette, J. A. & P. G. Koehler. 1992. Comparative toxicity of aqueous solutions of boric acid and polybor 3 to house flies (Diptera: Muscidae). J. Econ. Entomol. 85: 1209-1212.

Hogsette, J. A. and R. D. Jacobs. 1999. Failure of *Hydrotaea aenescens*, a larval predator of the house fly, *Musca domestica*, to establish in wet poultry manure on a commercial farm in Florida, U.S.A.,. Med. Vet. Entomol. 13: 349-354.

Hogsette, J. A., R. D. Jacobs & R. W. Miller. 1993. The sticky card: device for studying the distribution of adult house fly (Diptera: Muscidae) populations in closed poultry houses. J. Econ. Entomol. 86: 450-454.

Howard, J. J. and R. Wall. 1998. Effects of contrast on attraction of the housefly, *Musca domestica*, to visual targets. Med. Vet. Entomol. 12:322-324.

Ichinose, T., S. Shibazaki & M. Ohta. 1980. Studies on the biology and mode infestation of the tenebrionid beetle *Alphitobius diaperinus* (Panzer) harmful to broiler-chicken houses. Japan. J. Applied Zool. 34: 417-421 (in Japanese).

Iwasa, M, S-I Makino, H. Asakura, H. Kobori, Y. Morimoto. 1999. Detection of *Escherichia coli* O157:H7 from Musca domestica (Diptera: Muscidae) at a cattle farm in Japan. J. Med. Entomol. 36: 108-112.

Johmson, C. A. H. Bishop, and C. L. Turner. 1998. Isolation and activity of strains of *Bacillus thuringiensis* toxic to larvae of the housefly (Diptera: Muscidae) and tropical blowflies. J. Invertebr. Pathol. 71: 138-144.

Kalsbeek, V. J. K. Pell and T. Steenberg. 2001. Sporulation by *Entomophthora schizophorae* (Zygomycetes:Entomophthorales) from housefly cadavers and the persistence of primary conidia at constant temperatures and relative humidities. J. Invertebr. Pathol. 77, no. 3, pp. 149-157.

Kaufman, P. E., D. A. Rutz and S. Frisch. 2000a. Sticky traps for large scale house fly (Diptera: Muscidae) trapping in New York poultry facilities. J. Agric. Urban Entomol. 18: 43-49.

Kaufman, P. E., S. J. Long, D. A. Rutz, and C. S. Glenister. 2000b. Prey and density mediated dispersal in *Carcinops pumilio* (Coleoptera: Histeridae), a predator of house fly (Diptera: Muscidae) eggs and larvae. J. Med. Entomol. 37: 929-932.

Kaufman, P. E., S. J. Long, D. A. Rutz, and J. K. Waldron. 2001a. Parasitism rates of *Muscidifurax raptorellus* and *Nasonia vitripennis* (Hymenoptera: Pteromalidae) after individual and paired releases in New York poultry facilities. J. Econ. Entomol. 94: 593-598.

Kaufman, P.E., S. J. Long, and D. A. Rutz. 2001b. Impact of exposure length and pupal source on *Muscidifurax raptorellus* and *Nasonia vitripennis* (Hymenoptera: Pteromalidae) parasitism in a New York poultry facility. J. Econ. Entomol. 94: 998-1003.

King, B. H. 1990. Interspecific differences in host (Diptera: Muscidae) size and species usage among parasitoid wasps (Hymenoptera: Pteromalidae) in a poultry house. Environ. Entomol. 19: 1519-1522.

Kirkwood, A. C. 1963. Longevity of the mites *Dermanyssus gallinae* and *Liponyssus sylviarum*. Exp. Parasitol. 14: 358-366.

Kirkwood, A. C. 1968. Some observations on the feeding habits of the poultry mites, *Dermanyssus gallinae* and *Liponyssus sylviarum*. Entomol. Exp. Appl. 11: 315 320.

Lancaster, J. L. & J. S. Simco. 1967. Biology of the lesser mealworm, a suspected reservoir of avian leucosis. Univ. Arkansas Agric. Expt. Stn. Rep. No. 159.

Lemke, L. A. C. H. Collison, K. C. Kim. 1988. Host digestion to determine populations of the northern fowl mite, *Ornithonyssus sylviarum* (Acari: Macronyssidae), on mature chickens. J. of Med. Entomol. 25: 183-185.

Levine, O. S. and M. M. Levine. Houseflies (*Musca domestica*) as mechanical vectors of shigellosis. Revi. Inf. Dis. 13:688-696.

Levinson, A. R., Levinson, H. Z., Francke, W. 1981. Intraspecific attractants of the hide beetle *Dermestes maculatus* (De Geer). Mitt. Stsch. Ges. Allg. Angew. Entomol. 2: 235-237.

Levot, G. W. 1992. Chemical control of *Ornithonyssus sylviarum* on caged layer hens. Med. Vet. Entomol. 6:131-134.

Liao, S.-C. 1999. Mortality and repellency effects of essential oils from citrus against the housefly and German cokgoach. Chinese J. Entomol. 19: 153-160.

Loomis, E. C. 1978. External parasites, pp. 667-704. In M. S. Hofstad [ed,], Diseases of poultry, 7th ed. Iowa St. Univ. Press, Ames.

Loomis, E. E., E. L. Bramhall, J. A. Allen, R. A. Ernst & L. L. Dunning. 1970. Effects of the northern fowl mite on white Leghorn chickens. J. Econ. Entomol. 63: 1885 1889.

Lyon, W. F. 1975. How external parasites cut profits. Poult. Dig. Jan.: 33. Metcalf, C. L., W. P. Flint & R. L. Metcalf. 1962. Destructive and useful insects, 4th ed. McGraw-Hill, New York.

Lysyk, T. J. 1991. Effects of temperature, food, and sucrose feeding on longevity of the house fly (Diptera: Muscidae). Environ. Entomol. 20: 1176-1180.

Lysyk, T. J. 1998. Relationships between temperature and life history parameters of *Trichomalopsis sarcophagae* (Hymenoptera: Pteromalidae). Environ. Entomol. 27: 488-498.

Lysyk, T. J. 2000. Relationships between temperature and life history parameters of *Muscidifurax raptor* (Hymenoptera: Pteromalidae). Environ. Entomol. 29: 596 505.

Lysyk, T. J. 2001a. Relationships between temperature and life history parameters of *Muscidifurax zaraptor* (Hymenoptera: Pteromalidae). Environ. Entomol. 30: 147 156.

Lysyk, T. J. 2001b. Relationships between temperature and life history parameters of *Muscidifurax raptorellus* (Hymenoptera: Pteromalidae). Environ. Entomol. 30:982-992.

Lysyk, T. J. & R. C. Axtell. 1986. Comparison of baited jug-trap and spot cards for sampling house fly, *Musca domestica* (Diptera: Muscidae), populations in poultry houses. Environ. Entomol. 14: 815-819.

Madiera, N. G. 1998. Persistence of conidia of *Entomophtora muscae* in relation to age, temperature, and humidty. BioControl 43:87-95.

Maganga, M. E. G. Gries, and R. Gries. 1996. Repellency of various oils and pine oil constituents to house flies (Diptera: Muscidae). Environ. Entomol. 25: 5, 1182-1187.

Mandeville, D. A. & B. A. Mullens. 1990. Host preference and learning in *Muscidifurax zaraptor* (Hymenoptera: Pteromalidae). Annals, Entomol. Soc. Amer. 83: 1203-1209.

McAllister, J. C., C. D. Steelman, and J. K. Skeeles. 1994. Reservoir competence of the lesser mealworm (Coleoptera: Tenebrionidae) for *Salmonella typhimurium* (Eubacteriales: Enterobacteriaceae). J. of Med. Entomol. 31: 369-372.

McAllister, J. C, C. D. Steelman, L. A. Newberry, and J. K.Skeeles. 1995. Isolation of infectious bursal disease virus from the lesser mealworm, *Alphitobius diaperinus* (Panzer). Poult. Sci. 74:45-49

McAllister J. C., C. D. Steelman J. K. Skeeles, L. A. Newberry and E. E. Gbur. 1996. Reservoir competence of *Alphitobius diaperinus* (Coleoptera:Tenebrionidae) for *Escherichia coli* (Eubacteriales:Enterobacteriaceae). J. Med. Entomol. 33: 983-987.

Miller, W. V. & F. C. Price. 1977. The avian mite, *Ornithonyssus sylviarum*, on mammalian hosts with references to transmission to poultry. J. Parasitol. 63: 417.

Moon, R. D., J. L. Hinton, S. D. O'Rourke, and D. R. Schmidt. 2001. Nutritional value of fresh and composted poultry manure for house fly (Diptera: Muscidae) larvae. J. of Econ. Entomol. 94: 1308-1317.

Moriya, K., T. Fujibayashi, T. Yoshihara, A. Matsuda, N. Sumi, N. Umezaki, H. Kurahashi, N. Agui, A. Wada and H. Watanabe. 1999. Verotoxin-producing *Escherichia coli* O157:H7 carried by the housefly in Japan. Med. Vet. Entomol. 13: 214-216.

Mullens, B. A. & J. R. Rodriguez. 1992. Effects of disodium octaborate tetrahydrate on survival, behavior, and egg viability of adult muscoid flies (Diptera: Muscidae). J. Econ. Entomol. 85: 137-143.

Mullens, B. A., J. L. Rodriguez & J. A. Meyer. 1987. An epizootiological study of *Entomophthora muscae* in muscoid fly populations on southern California poultry facilities, with emphasis on *Musca domestica*. Hilgardia 55: 1-41.

Mullens, B. A., W. D. McKeen, J. L. Rodriguez & J. D. Mandeville. 1988. Evaluation of ABG-6208 (Thuringiensin) for control of northern fowl mite, 1987. Insecticide & Acaricide Tests 13: 405.

Mullens, B. A., N. C. Hinkle, and C. E. Szijj. 1996a. Role of the poultry manure pad in manure drying and its potential relationship to filth fly control. J. Agric. Entomol. 13: 331-337.

Mullens, B. A., N. C. Hinkle, and C. E. Szijj. 1996b. Impact of alternating manure removal schedules on pest flies (Diptera: Muscidae) and associated predators (Coleoptera: Husteridae, Staphylinidae; Acarina: Macrochelidae) in caged-layer poultry manure in southern California. J. Econ. Entomol. 89:1406-1417.

Mullens, B. A. N. C. Hinkle, and C. E. Szijj. 2000. Monitoring northern fowl mites (Acari: Macronyssidae) in caged laying hens: Feasibility of an egg-based sampling system. J. of Econ. Entomol. 93: 1045-1054.

Petersen, J. J. & B. M. Pawson. 1991. Early season introduction, population increase, and movement of the filth fly parasite *Muscidifurax zaraptor* (Hymenoptera: Pteromalidae). Environ. Entomol. 20: 1155-1159.

Petersen, J. J. and J. K. Cawthra. 1995. Release of a gregarious *Muscidifurax species* (Hymenoptera: Pteromalidae) for the control of filth flies associated with confined beef cattle. Biol. Control 5: 279-284.

Petersen, J. J. and D. M. Currey. 1996. Timing of releases of gregarious *Muscidifurax raptorellus* (Hymenoptera: Pteromalidae) to control flies associated with confined beef cattle. J. Agric. Entomol. 13: 55-63.

Pickens, L. G. G. D. Mills, R. W. Miller. 1994. Inexpensive trap for captoring house flies (Diptera: Muscidae) in manure pits of caged-layer houses. J. Econ. Entomol. 87:116-119.

Rakowski, G. 1988. Effect of illumination intensity on the response of the hide beetle, *Dermestes maculatus* , to aggregation pheromone. J. Insect Physiol. 34: 1101 1104.

Rakowski, G., Cymborowski, B. 1982. Aggregation pheromone in *Dermestes maculatus*: effects on larval growth and developmental rhythms fecundity. Int. J. Invertebr. Reprod. Dev. 4: 249-254.

Rakowski, G., Cymborowski, B. 1986. Some environmental and physiological factors influencing the response of the hide beetle, *Dermestes maculatus* to aggregation pheromone. Int. J. Invertebr. Reprod. Dev. 9: 35-41.

Rakowski, G., Sorensen, K. A., Bell, W. J. 1989. Responses of dermestid beetles, *Dermestes maculatus* , to puffs of aggregation pheromone extract (Coleoptera: Dermestidae). Entomol. Gen. 14: 211-215.

Raspi, A. and R. Antonelli. 1995. Influence of constant temperature on the development of *Dermestes maculatus* Deg. (Coleoptera Dermestidae). Frustula Entomol. 31: 169-176.

Renn, N. 1994. A comparison of baits containing different isolates of entomopathogenic nematodes for the control of *Musca domestica*, using a large-scale laboratory facility. In: Proc. Brighton Crop Protection Conf. 1994. Vol 3, The British Crop Protection Council, Farham, U.K. 1061-1066.

Renn, N. 1998. The efficacy of entomopathogenic nematodes for controlling houselfy infestations of intensive pig units. Med. Vet. Entomol. 12: 46-51.

Renn, N. A. F. Bywater and G. Barson. 1999. A bait formulated with *Metarhizium anisopliae* for the control of *Musca domestica* L. (Dipt. Muscidae) assessed in large-scale laboratory enclosures. J. Appl. Entomol. 123: 309-314.

Rueda L. M. and R. C. Axtell. 1996. Temperature-dependent development and survival of the lesser mealworm, *Alphitobius diaperinus*. Med. Vet. Entomol 10: 80-86.

Rutz, D. A. & G. A. Scoles. 1988. Evaluation of fly-electrocuting black light devices in caged-layer poultry facilities. Poultry Sci. 67: 871-877.

Rutz, D. A. & R. S. Patterson (eds). 1990. Biocontrol of Arthropods Affecting Livestock and Poultry. Westview Press, Boulder.

Safrit, R. D. & R. C. Axtell. 1984. Evaluations of sampling methods for darkling beetles (*Alphitobius diaperinus*) in litter of turkey and broiler houses. Poultry Sci. 63: 2368-2375.

Sarin, K. & S. C. Saxena. 1973. Effect of temperature and relative humidity on the development and mortality of *Alphitobius diaperinus* Panzer, a stored product pest. Folia Biol. 21: 223-228.

Sasaki, T., M. Kobayashi, and N. Agui. 2000. Epidemiological potential of excretion and regurgitation by *Musca domestica* (Diptera: Muscidae) in the dissemination of *Escherichia coli* O157:H7 to food. J. Med. Entomol. 37: 945-949.

Schmitz, M. & R. Wohlgemuth. 1988. Studies on mass development and behavior of the lesser mealworm *Alphitobius diaperinu*s Panz, in poultry houses and its control. Anz. Schadlinskdunde Pflanzenschutz Umweltschutz 61: 191-194 (in German).

Sheppard. D. C. 1983. House fly and lesser house fly control utilizing the black soldier fly in manure management systems for caged laying hens. Environ. Entomol. 12: 1239-1242.

Sheppard, D. C. and R. Noblet. 1999. Livestock and poultry insetcs. In : Summary of losses from insect damage and cost of control in Georgia, R, M. McPherson, G. K. Douce, and D. G. Riley, eds. Georgia Agric. Exper. Station, Tifton, GA, 17 pp.

Sheppard, D. C., D. M. Gaydon & R. W. Miller. 1992. J. Agric. Entomol. 9:257-260.

Sikes, R. K. & R. W. Chamberlain. 1954. Laboratory observations on three species of bird mites. J. Parasitol. 40: 691-697.

Singh, D and A. K. Singh. 1992. Repellent and insecticidal properties of essential oils against housefly, *Musca domestica* L. Insect Science and Its Application 12: 487-491

Six, D. L. and B. A. Mullins. 1996. Distance of conidial discharge of *Entomophthora muscae* and *Entomophtora schizophorae* (Zygomycotina: Entomophthorales). J. Invertebr. Pathology. 67: 253-258.

Stafford, K. C., III, C. H. Collison, J. G. Burg & J. A. Cloud. 1988. Distribution and monitoring lesser mealworms, hide beetles, and other fauna in high-rise, caged-layer poultry houses. J. Agric. Entomol. 5: 89-101.

Stafford, K. C., III and D. E. Bay. 1994. Dispersion statistics and sample size estimates for house fly (Diptera: Muscidae) larvae and *Macrocheles muscaedomesticae* (Acari: Macrochelidae) in poultry manure. J. Med. Entomol. 31:732-737.

Steinkraus, D. C. & E. A. Cross. 1993. Description and life history of *Acarophenax mahunkai* n. sp. (Acari, Tarsonemina: Acarophenacidae), an egg parasite of the lesser mealworm (Coleoptera: Tenebrionidae). Ann. Entomol. Soc. Amer. 86: 239-249.

Steinkraus, D. C., C. J. Geden & D. A. Rutz. 1991. Susceptibility of lesser mealworm (Coleoptera: Tenebrionidae) to *Beauveria bassiana*: effects of host stage, formulation, substrate and host passage. J. Med. Entomol. 28: 314-321.

Steinkraus, D. C., W. A. Brooks, C. J. Geden & D. A. Rutz. 1992. Discovery of the neogregarine *Farinocystis tribolii* & eugregarine in the lesser mealworm, *Alphitobius diaperinus*. J. Invert. Pathol. 59: 203-205.

Swatonek, V. F. 1970. Zur Biologie des Glanzendschwarzen Getreideschimmelkafers (*Alphitobius diaperinus* Panz. = *A. piceus* Oliv). Anz. Schadlinskunde Pflanzenschutz 43(7): 101-104 (in German).

Le Torc'h, J. M. & R. Letenneur. 1983. Laboratory tests of resistance of different thermic insulators to the boring of the tenebrionid *Alphitobius diaperinus* (Col. Tenbrionidae). Comptes Rendus Hebdomadairs des Sceances, Acad. Agric., Paris 806: 188-200 (in French).

Tan, S. W., K. L. Yap, and H. L. Lee. 1997. Mechanical transport of rotavirus by the legs and wings of *Musca domestica* (Diptera: Muscidae). J. Med. Entomol. 34: 527-531.

Tobin, P. C., and C. W. Pitts. 1999. Flotation method for sctracting insects from poultry manure samples. J. Med. Entomol. 36: 1210123.

Turner, E. C., Jr. 1978. Biology & control of northern fowl mites on caged layers. J. N. Y. Entomol. Soc. 86: 325.

Turner, E. C., Jr. 1986. Structural & litter pests. Poultry Sci. 65: 644-648.

Turner, E. C. & L. Carter. Mass rearing & introduction of *Ophyra aenescens* (Wiedemann) (Diptera: Muscidae) in high-rise caged layer houses to reduce house fly populations. J. Agric. Entomol. 7: 247-257.

Vaughan, J. A. & E. C. Turner, Jr. 1984. Residual & topical toxicity of various insecticides to the lesser mealworm (Coleoptera: Tenebrionidae). J. Econ. Entomol. 77: 216-220.

Vaughan, J. A., E. C. Turner, Jr. & P. L. Ruszler. 1984. Infestation & damage of poultry house insulation by the lesser mealworm, *Alphitobius diaperinus* (Panzer). Poultry Sci. 63: 1094-1100.

Wallace, M. M. H., R. G. Winks & J. Voestermans. 1985. The use of a beetle, *Alphitobius diaperinus* (Panzer) (Coleoptera: Tenebionidae) for the biological control of poultry dung in high-rise layer houses. J. Australian Inst. Agric. Sci. 51: 214-219.

Watson, D. W., B. A. Mullens & J. J. Petersen. 1993. Behavioral fever responses of *Musca domestica* (Diptera: Muscidae) to infection by *Entomophthora muscae* (Zygomyces: Entomophthoraceae). J. Invert. Pathol. 61: 10-16.

Watson, D. W. , J. S. Guy, and S. M. Stringham. 2000. Limited transmission of turkey coronavirus in young turkeys by adult *Alphitobius diaperinus* (Coleoptera: Tenebrionidae). J. Med. Entomol. 37: 480-483.

Wilhoit, L. R., R. E. Stinner & R. C. Axtell. 1991a. CARMOD: a simulation model for *Carcinops pumilio* (Coleoptera: Histeridae) population dynamics & predation on immature stages of house flies (Diptera: Muscidae). Environ. Entomol. 20: 1079-1088.

Wilhoit, L. R., R. E. Stinner & R. C. Axtell. 1991b. Computer simulation model house fly management in confined-animal production systems. North Carolina State University Technical Bulletin No. 296, 81pp.

Wills, L. E., B. A. Mullens & D. A. Mandeville. 1990. Effects of pesticides on filth fly predators (Coleoptera: Misteridae, Staphylinidae; Acarina: Macrochelidae, Uropodidae) in caged layer poultry manure. J. Econ. Entomol. 83: 451-457.

Wilson, T. H. & F. D. Minor. 1969. Influence of temperature on development of the lesser mealworm, *Alphitobius diaperinus* (Coleoptera: Tenebrionidae). J. Kansas Entomol. Soc. 42: 294-303.

Zchori-Fein, E., C. J. Geden & D. A. Rutz. 1992. Microsporidioses of pteromalid parasitoids of muscoid flies. J. Invert. Pathol. 60: 292-298.

Zhong, C. D. J. Ellar, A. Bishop, C. Johnson, S. Lin, and E. R. Hart. 2000. Characterization of a *Bacillus thuringiensis* delta endotoxin which is toxic to insects in three orders. J. Invertebr. Pathol. 76: 131-139.

Zurek, L., C. Schal and D. W. Watson. 2000. Diversity and contribution of the intestinal bacterial community to the development of *Musca domestica* (Diptera: Muscidae) larvae. J. Med. Entomol. 37: 924-928.

Zurek, L; Denning, SS; Schal, C; Watson, DW. 2001. Vector competence of *Musca domestica* (Diptera: Muscidae) for *Yersinia pseudotuberculosis.* J. Med. Entomol. 38: 333-335.

****

End of Poultry section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****

DAIRY CATTLE SUMMARY

1. IPM is recognized as a preferred best management practice to minimize pest problems in dairy cattle systems and help protect farm workers, consumers and the environment.

2. Much dairy cattle pest management information is currently available to extend, however, there are very limited resources available for extension outreach and continued research.

3. There has been an alarming trend towards a decreasing number of veterinary entomologists nationwide. With animal agriculture contributing 47% towards the total U.S farm income, the critical question, then, is who and how will the research, teaching, and extension needs of this significant segment of U.S. agriculture be met. Cornell University now has the only active research program for dairy cattle arthropod pests throughout the United States. Likewise, active extension programs for dairy cattle IPM appear limited to University of Minnesota, University of California, Riverside, and Cornell University.

4. The need for IPM research has become critical over the last number of years due to FIFRA 88. In 1988 the Federal Insecticide, Fungicide and Rodenticide Act (FIFRA) was amended. A requirement of FIFRA 88 was that all pesticides registered by the EPA prior to 1984 had to now go through the current, more rigorous reregistration process. Unfortunately, livestock and poultry are considered minor crops by the agrichemical companies. Therefore, many of these companies have made the economic decision not to pursue the reregistration of these pesticides due to the high costs of the additional required testing. This problem has been further exascerbated by the passage of the FQPA. Furthermore, we have had essentially no new chemistry for livestock and poultry arthropod pest management for almost 17 years.

This has generated tremendous problems for dairy cattle pest management by requiring the use of only a few pesticide active ingredients, which has resulted in exceptionally high levels of pesticide resistance in our pest populations. In the absence of effective pesticides, the dairy industry is now desperate for new, cost effective pest management options and is more willing to try pesticide alternatives such as biological control.

5. The use and application of the economic thresholds to dairy cattle systems should be reevaluated. Economic thresholds for animal arthropod pests are difficult to determine. In addition, it is difficult to apply this concept to pest problems which affect public health and/or cause a public nuisance. It is suggested that the concept of tolerance thresholds or aesthetic thresholds be evaluated for their potential application in dairy cattle IPM.

6. Researchers and extension personnel should continue to work towards development of user-friendly recommendations and systems which enhance adoption of dairy cattle IPM practices. Integration of appropriate dairy cattle IPM information into animal health, management, and production programs, such as integrated herd health and integrated reproduction management will help enhance adoption of IPM practices by clientele.

Workshop Recommendations

Continued development and transfer of dairy cattle IPM information is critical to address these pest management issues. To enhance these efforts, the following recommendations are offered:

- Regional research and extension centers are needed to help develop effective IPM strategies and help transfer dairy cattle IPM information to producers and the general public. Funding for these centers could be provided by regional, Smith-Lever, Hatch and industry money.
- Veterinary entomologists should seek ways to enhance networking, sharing of dairy cattle IPM educational materials, and research and extension efforts.
- The dairy industry has an important role in both the direct and indirect development of needed research on environmentally sound management alternatives, and as amplifiers for extending proven IPM technology.
- Departments of Public Health can help promote the use of IPM and potentially provide funding for addressing and resolving local public nuisance and public health concerns related to dairy cattle pest management.

DAIRY CATTLE

Committee Members

Dr. Don Rutz, Chair
Dr. Truman Fincher
Dr. John George
Dr. Reid Gerhardt
Dr. Carl Jones
Dr. Tim Lysyk
Dr. Dick Miller
Dr. Charlie Pitts
Dr. Ed Schmidtmann
Dr. Wes Watson

Economic Significance of Dairy Cattle

From modest beginnings in upstate New York in the 1840's, dairy production in the United States has developed into a major agricultural industry. Approximately 9 million cattle, milked on 117,000 farms, now produce 157 billion pounds of milk annually (Anonymous 1999). Revenue generated by the sale of dairy products in 1997 was $21 billion at the farm level (Anonymous 1999).

The high nutritional level of fluid milk, butter, cheese, and low-fat products is virtually uncontested. In addition to high protein levels, milk contains butterfat for energy metabolism, calcium and phosphorus for bone growth, and a well balanced supply of vitamins A, D, E, K, and riboflavin. Dairying, therefore, not only contributes strongly to the domestic economy, especially in rural areas, but also provides a high volume of nutritious and appetizing food. A thriving trade also exists in the sale of dairy beef, veal, and the export of registered dairy stock to foreign countries.

Animal agriculture, according to the latest available national agricultural statistics, generates $96.5 billion annually, which is 47% of the total U.S. agricultural cash receipts (Table 1). Specifically, the dairy industry contributes approximately 10% of the total U.S. farm income.

Regional shifts in milk production began about three decades ago and have accelerated in the last 20 years. Milk production appears to have grown disproportionately in the Pacific region and to some extent in the mountain states and Southern Plains (Table 2).

References Cited

Anonymous. 1999. Statistical Highlights of U.S. Agriculture. USDA-NASS. http://www.usda.gov/nass/pubs/stathigh/1999

Perez, A. M. 1993. Changing Structure of U.S. Dairy Farms. USDA/ERS Agricultural Report.

Table 1. 1997 U.S. Cash receipts by commodity.		
Commodity	Billions of Dollars	% of Total
Livestock Products:		
Meat Animals*	49.9	24
Dairy Products	21.0	10
Poultry and Eggs	22.2	11
Misc. Livestock	3.5	2
Total	96.6	47
.		
Crops:		
Food Grains	10.6	5
Feed Crops	27.6	13
Cotton	6.5	1
Tobacco	2.9	3
Oil Crops	19.9	10
Vegetables	15.1	7
Fruits and Nuts	12.8	6
All Other Crops	16.7	8
Total	112.1	52
.		
TOTAL, ALL COMMODITIES	208.7	100
.		
* Beef, hogs, sheep, and lambs		

Table 2. Percent change in the number of milk cows during 1977 to 1992, by region. (Source: Perez 1993).		
Region	States in Region	% Change

Northeast	CT, DE, ME, MD, MA, NY, NH, NJ, PA, RI, VT	-16.3
Lake States	MI, MN, WI	-14.2
Corn Belt	IL, IN, IA, MO, OH	-22.6
Appalachia	KY, NC, TN, VA, WV	-24.5
Southeast	FL. GA, SC	-17.9
Delta	AL, AR, LA, MS	-40.3
Northern Plains	KS, NE, ND, SD	-27.1
Southern Plains	OK, TX	+9.2
Mountain	AZ, CO, ID, MT, NV, NM, UT, WY	+36.3
Pacific	AK, CA, HI, OR, WA	+35.8

Arthropod Pests of Dairy Cattle

Associated with dairy cattle in barns, dry lots and pastures across the country are a wide variety of arthropod pests dependent upon bovine tissues or feces for their growth and reproduction. Lowered levels of milk production, reduced feed conversion efficiency, exposure to debilitating disease-causing agents, hide damage and public health and nuisance concerns result from arthropod pest activity. The time and manpower "costs" involved in manure disposal, as well as the purc Associated with dairy cattle in barns, dry lots and pastures across the country are a wide variety of arthropod pests dependent upon bovine tissues or feces for their growth and reproduction. Lowered levels of milk production, reduced feed conversion efficiency, exposure to debilitating disease-causing agents, hide damage and public health and nuisance concerns result from arthropod pest activity. The time and manpower "costs" involved in manure disposal, as well as the purchase and application of pesticides, also contribute to pest-induced economic loss.

The arthropod fauna associated with dairy cattle can be grouped into several ecologically-based pest complexes. They are: 1) Muscoid flies inhabiting barns and dry lots (house fly and stable fly); 2) muscoid flies found on pastured cattle (face fly and horn fly); 3) indigenous lower Diptera that blood feed from pastured cattle (mosquitoes, horse flies, black flies and biting midges); and 4) winter-active ectoparasites such as lice and mange mites. The population density and hence economic impact of these groups vary greatly with the season of the year, geographical location and type of dairy cattle housing system. Specific information concerning the bionomics, economic damage and estimated research and extension needs for developing integrated pest management (IPM) programs for each complex are presented.

House Fly

Description and Biology. Despite its long association with man, the house fly remains one of the most difficult livestock pests to control. In dairy barns and immediate surroundings, adult house flies frequent posts, walls, ceiling, stanchions and are also found feeding from spilled and fermenting feed and decomposing manure. Concentrations of vomit or fecal spots indicate sites of house fly aggregation. Inside of barns, house flies are also found on cattle, especially new born calves, where they frequent the eyes and nostrils. Outside the barn, house flies spend little time on cattle. In areas where the face fly is absent, however, house flies may frequent the eyes and nostrils of cattle, feeding from lacrymal secretion and nasal discharges. At summer temperatures, the complete life cycle takes about 8-16 days. This short generation time underlies the rapid increase in house fly populations observed during the summer months. House fly eggs are deposited in decaying organic matter. Manure mixed with straw is a favored medium for house fly larval development. Warm temperatures in dairy barns, may permit breeding throughout the year (Hanec 1956).

Economic Importance. Freeborn et al. (1925) demonstrated a 3.3% loss in milk production attributable to house fly annoyance; however, a follow-up study three years later showed no effect (Freeborn et al. 1928). Estimated annual losses from 1973-1975 due to house fly on dairy cows has been reported to be $30 million (USDA 1976). Another report noted that it cost $493,500 for the year 1976 to control house fly in Arkansas dairy barns (USDA 1978). In a summary report of national research efforts, it was estimated that losses attributed to house fly on a national level exceeded $100 million/year (Anonymous 1976).

The major economic effect of house flies on dairy cattle management stems from the public health and public nuisance importance of adult populations. To maintain a high quality, milk must be produced, handled, and stored in a clean and sanitary environment. Public health authorities consider numerous flies in and around the dairy as evidence that sanitary standards are not being met. Further, milk produced under unsanitary conditions will tend to have higher bacterial counts. With more cows being milked in parlors where milk goes directly into a bulk tank this may be less of a problem than in the past. As more and more people move from the suburbs into previously rural areas, the public nuisance caused by flies dispersing from dairies becomes an ever-increasing problem (Miller 1993).

Other problems in this regard include odors, lack of land for manure disposal, and possible contamination of groundwater with run off from the farms. Areas of land used for manure disposal, which are often some distance from the main farm, may at times, create the same problems with the neighbors as the farm itself.

Methods of Control.

(1) Conventional Control Methods. With the development of residually active chlorinated hydrocarbons in the 1940s, dairy farmers developed a dependency upon the use of residual sprays applied to the ceilings and walls of dairy barns for control of house flies. As resistance developed to organochlorine products, newly developed organophosphates were tested and used. Today, residual sprays are less widely used owing to widespread resistance and the unavailability of new products. Other chemical methods used are space sprays of pyrethrin or dichlorvos applied as needed, attractant bait granules and an oral larvacide. The construction of open enclosure free stall barns and sheds has brought about less need for residual sprays and greater use of space sprays.

At our 1979 workshop resistance of the house fly to organochlorine and organophosphorus insecticides was cited as a problem. About the same time synthetic pyrethroid insecticides became available for control of flies on dairy farms. Soon after the start of their use, however, there were reports of house fly resistance. Harris et al. (1982) reported that on Canadian farms which relied heavily on insecticides for fly control, house flies were resistant to all organochlorine, organophosphorus, carbamate, and pyrethroid insecticides. MacDonald et al. (1983) reported that the buildup of resistance to permethrin could be slowed down by alternating the use of permethrin and dichlorvos. Meyer et al. (1987) documented the resistance of house flies to permethrin on southern California dairy farms. The more frequently permethrin was used, the higher the level of resistance. Scott et al. (1989) reported high frequencies and high levels of resistance to crotoxyphos, dimethoate, and tetrachlorvinphos on New York dairies. Resistance to permethrin was high on one-half of the dairies sampled.

Today, because of resistance problems and unavailability of new products, residual sprays for house flies are used less than in the past. According to a survey conducted by Partridge et al. (1991) and Harrington et al. (1997) the use of baits and fogging/misting in barns were the most frequently used methods for house fly control by dairymen in New York state. Since new insecticides will probably not be registered for control of the house fly in the near future, fly control methods must be developed, which rely less on traditional insecticides.

(2) Insect Growth Regulators. In 1979, two growth regulators were mentioned as being tested as feed additives for fly control, but because of the high dosages needed for house fly control, their practical use was questioned (Miller & Uebel 1974). It was also pointed out that there were reports of resistance to juvenile hormone analogues (Plapp 1976). Since that time, however, there have been reports of two insect growth regulators that may play a role in the control of house flies on dairy farms. Miller and Schmidtmann (1985) reported on the feeding of cyromazine to dairy calves for the control of house flies. Cyromazine incorporated into the milk of young dairy calves is excreted in the urine and prevents house fly development when mixed with the calf bedding. This study was followed up by a two-year IR-4 study. IR-4 headquarters was to have processed these data and were to have submitted a package to EPA in 1994.

The other insect growth regulator evaluated for house fly control with dairy cattle is the diflubenzuron (Vigilante) bolus. Miller et al. (1991) reported that this product had no adverse effect on the milk yield or percentage of fat or protein when compared with untreated control cows. There was also no indication that administration of diflubenzuron boluses reduced the overall house fly populations on commercial dairy farms. In later work Miller (1994) reported that the bedding from the heifers administered diflubenzuron boluses spread on the field produced less house flies than bedding from non-treated cows. Pyriproxyfen boluses were also tested against the house fly in *in vivo* tests, but were not as active as diflubenzuron (Miller & Miller 1994).

Two compounds which would not technically be classified as insect growth regulators have been tested for activity against house flies. Hogsette and Koehler (1992) tested boric acid and disodium octaborate tetrahydrate and Mullens and Rodriguez (1992) tested the latter compound for activity against adult house flies. Although rather high concentrations of the compounds were needed to kill flies, with increasing unavailability of new insecticides, boric acid could play a role in house fly control on dairy farms.

An extensive literature exists on the effect of chemosterilants for house fly control (Fye et al. 1973). The use of chemosterilants for house fly control must be carefully re-evaluated in terms of practicability and whether it will ever reach the implementation phase.

(3) Cultural, Mechanical and Sanitation. The importance of limiting house fly breeding by employing proper sanitation on dairy farms is paramount. Pickens et al. (1967) emphasized that without sanitation measures, fly dispersal from adjacent areas will offset any control measures. Further, good sanitation will result in a reduction in house fly populations to two-thirds that found at farms with poor sanitation (Pickens et al. 1967). Since 1979, there has been research conducted to determine and characterize both house fly and stable fly breeding sites in Nebraska (Meyer & Peterson 1983), California (Meyer & Shultz 1990), and Canada (Lysyk 1993). Stored manure, manure mixed with hay or straw, silage, and fermenting grain were major sources of house fly breeding. Schmidtmann (1988) reported that bedding used in outdoor calf hutches on dairy farms was an important source of both house fly and stable fly development.

Since common breeding sites for house flies on dairy farms are well identified, it seems reasonable to expect that sanitation and mechanical methods could be used to reduce fly populations with a limited use of insecticides. Schmidtmann et al. (1989) and Schmidtmann (1991) demonstrated that when calf hutches were bedded with materials such as woodchips, ground corncobs, or sawdust, house fly and stable fly development was suppressed. Thus a major source of house flies on dairy farms could be reduced by this relatively simple change in management practices.

As part of an integrated program for house fly control on dairy farms Geden et al. (1992) had farmers clean out their calf pens on a weekly basis. It was felt that this component played a major role in the house fly reduction found on the treated farms. Lazarus et al. (1989) reported that it was economically feasible to reduce insecticide application on dairy farms for house fly control if manure was removed from the barns every seven days.

Lately there has been a renewed interest in using traps for control of house flies on dairy farms. Pickens and Miller (1987) reported that traps with an improved artificial bait and sticky pyramidal traps captured large numbers of house flies on dairy farms. Miller et al. (1993b) reported that in field trials these traps reduced house fly populations on commercial dairy farms. In a latter study a combination bait-visual trap failed to reduce house fly populations on dairy farms if a sanitation component was not included in the treatment (Miller et al. 1993a).

Various traps with UV-emitting bulbs behind an electrocuting grid are on the market and being sold for dairy fly control. Pickens and Thimijan (1986) discussed the design parameters that affect performance of these traps. They also discuss the literature, which is somewhat controversial, as to the value of these traps in controlling house flies on dairy farms.

Pickens and Mills (1993) reported that a solar-powered electrocuting trap attracted and killed large numbers of both house flies and stable flies. Pickens et al. (1994) reviewed the use of all types of traps for house fly and stable fly control around livestock operations. They concluded that a combination of cultural, mechanical, and sanitation can lower house fly populations on dairy farms.

Morgan et al. (1972) significantly reduced fly populations in dairy barns by simply hanging doorway curtains in the entrance way. As cows enter, house flies are brushed from the backs and flanks of the animal. This technique had little effect on the entry of stable flies.

(4) Biological. Attempts have been made to catalog the literature on the biological control agents of insects of veterinary importance (Jenkins 1964; Legner et al. 1974; Bay et al. 1976; Roberts & Strand 1977). A publication, edited by Clausen (1978) provides a world review of all the introduced parasites and predators of arthropod pests. In addition, several books and journal review articles have been restricted to the biological control agents of this synanthropic fly (Legner & Olton 1970; West & Peters 1973).

The effects of a predatory mite (Axtell 1963) and parasitoid wasps (Morgan et al. 1976) were evaluated as natural control agents of house fly in dairy barns. In these reports it was stated that further research was needed on the use of parasitoids and predators in dairy barns with respect to (1) manure handling systems, (2) host-parasite population dynamics, and (3) pesticide compatibility with natural enemies. Research in these areas have been conducted since 1979.

Miller and Rutz (1990) and Smith and Rutz (1991a) surveyed dairy farms in Maryland and New York to determine seasonal and relative abundance of hymenopterous parasitoids attacking house fly pupae. The most common parasitoid found in both states was *Muscidifurax raptor*. The next most common species found in New York were *Urolepis rufipes*, *Phygadenon fumator*, and *Spalangia cameroni*. The first two species were not found in Maryland and the second most abundant species found in Maryland was *S. cameroni*. Total parasitism peaked one to two months after the house fly population peaked.

Greene et al. (1989) found that in the winter on dairy farms in Florida, the most common parasitoid of the house fly was *S. cameroni,* with *Muscidifurax* sp. the second most common species collected. *S. nigroaenea* and *S. endius* were also recovered. In general these same species were found associated with open silage in eastern Nebraska (Petersen & Meyer 1983).

The relationship of microhabitat to incidence of house fly immatures and parasitoids found on dairy farms in New York has been studied extensively to better understand which parasitoids are best to be used under various conditions (Smith & Rutz 1991bc). Smith et al. (1989) studied the influence of habitat and temperature on dispersal of *M. raptor* and *U. rufipes* during an inundative release at a dairy farm. Pawson and Petersen (1988) studied the dispersal of *M. zaraptor* on dairies in eastern Nebraska. Long et al. (1998) working on a New York dairy farm, reported that *M. raptorellus* produced multiple progeny only when temperatures exceeded 18šC and that parasitism rates did not differ from those observed with *M. raptor* when compared within 3 temperature ranges. These studies provide the background data as to how parasitoid releases might play a role in the control of house flies on dairy farms.

Since hymenopterous pupal parasitoids are found naturally on dairy farms, there have been attempts to release certain parasitoids to reduce house fly populations. Morgan et al. (1976) and Morgan and Patterson (1977) reported when they made inundative releases of *S. endius* on dairy farms, a high percentage of house fly pupae were parasitized. It was suggested that if the farms on which the parasitoids were released had been isolated adult house fly populations would have been reduced.

Meyer et al. (1990) released commercially reared *S. endius, M. raptorellus,* and *M. zaraptor* on dairy farms in southern California. They found that the parasitoid treatments had no apparent impact on adult populations of either the house fly or stable fly or on overall parasitism rate of field-collected house fly or stable fly pupae. Petersen et al. (1983) also released *S. endius* on eastern Nebraska feedlots and found that they were ineffective in controlling house fly or stable fly populations. These last two studies suggest that parasitoid releases, when used as the only control method, on large dairies or feedlots will probably not reduce adult populations of house flies.

Miller et al. (1993b) reported that when they released *M. raptor* on dairy farms in Maryland, house fly populations tended to be reduced, however, these differences were not statistically significant. Geden et al. (1992), however, found that when releases of *M. raptor* were part of an overall management plan which included a sanitation component, and limited use of insecticides, house fly populations were reduced on dairy farms in New York and Maryland.

In a recent study, Cornell workers (VanKirk 1994) implemented an IPM study to manage house flies and stable flies on dairy farms in New York state. The study was conducted as a demonstration extension project during 1992 and 1993. Sanitation, the release of *M. raptor* and the judicious use of insecticides were the main components of the IPM program. Based on spot card counts, IPM dairies in 1992 had fewer flies than those farms that were conventionally managed. Insecticide purchases and frequency of application were less for the IPM farms than the conventionally managed farms. The results were not as good in 1993 and the investigators are trying to determine the reason.

The fungus *Entomophtora muscae* has been isolated from house flies on dairy farms in California (Mullens 1985), Nebraska (Watson & Petersen 1993) and New York (Steinkraus et al. 1993). The effect of temperature and humidity on this fungus has been studied (Watson & Petersen 1993); epizootics appear to occur primarily in late summer or early fall (Watson & Petersen 1993, Steinkraus et al. 1993). Steinkraus used laboratory infected *E. muscae* flies to induce epizootics on dairy farms in New York state. Six and Mullens (1993) released a new form of *E. muscae*, *E. schizophorae* on California dairies in 1991. Although they had found that this species outcompetes *E. muscae* within the fly in the laboratory, *E. schizophorae* did not establish or spread on the dairy. It was concluded that *E. schizophorae* was not promising for biological control.

Steinkraus et al. (1990) found another fungal pathogen, *Beauveria bassiana*, in wild house flies on New York dairy farms. Most infected flies were collected in September and early October. They reported that dry conidia stored at room temperature for two years remained viable and infective to house fly adults. Watson et el. (1995) reported that dust formulations of *B. bassiana* were more effective for controlling adult stable flies and house flies than aqueous formulations. They further reported both a dose dependent response and following treatment, a loss in pathogenicity over time. Larval house flies were also found to be susceptible to *B. bassiana*. Watson et al. (1996) introduced *B. bassiana* into dairy calf hutches bedded with straw or sawdust. The number of fly (house and stable) larvae recovered from sawdust bedding was lower than the number from straw bedding. The prevalence of *B. bassiana* in the adult fly population was higher in hutches sprayed with conidia than in untreated control hutches.

Miller (1970) reported that a commercial *Bacillus thuringiensis* formulation had little effect on house fly larvae in feces from cattle fed the compound. More recently Schmidtmann et al. (1993) reported on the presence of *Bacillus spp.*, including *B. thuringiensis*, in calf pen bedding. Since some of these species have activity against house fly larvae, the authors suggest they may have potential in suppressing muscoid fly populations.

(5) Genetic Control. Some research has been developed in the area of genetic and sterile male release for house fly control (Bushland 1971; Labrecque & Weidhaas 1970; and Pal & LaChance 1974), but these techniques appear to have limited potential for controlling this particular pest. More recently, Iowa State workers (Black & Krafsur 1986, and Krafsur et al. 1992) have conducted some basic genetic studies on the house fly, but, again this work has little practical application for control of the house fly.

(6) Other. Even though a sex attractant has been identified, isolated, and synthesized for house fly (Carlson et al. 1971; Uebel et al. 1976), its use in any management program appears to be limited because it is not a long distance attractant. Vanillin has also been reported to be a fly attractant (Steinkraus et al. 1993). Hogsette and Jacobs (1993) reported on a house fly bait station that killed large numbers of house flies. Although these test were conducted in a poultry house, these stations might be able to be used on dairy farms.

Success has been achieved using the response of the adult to baits. The addition of insecticides to dry sugar baits for control of house flies in dairy barns in Florida was evaluated by Bailey et al. (1970). Mulla et al. (1977) reported on a slow-release solid synthetic fly attractant formulations that when combined with insecticidal bait stations offered promise for house fly control. Baits are widely used by dairymen to control house flies around barn areas (Partridge et al. 1991).

Current Support. Less than 1 SY, combined between ARS and CSRS, is currently being devoted to research on house fly in dairies.

Summary of Research and Extension Needs

(1) Economic Injury Level. Information correlating house fly populations with public health and public nuisance injury levels has not been determined. Miller (1993), however, suggests that as suburbs encroach upon previously rural areas this is an item that needs to be quantified if possible.

(2) Sampling. The attraction of adults to visual traps has been shown to be an effective monitoring tool (Morgan et al. 1970; Thimijan et al. 1970; Thimijan et al. 1972). Pickens et al. (1972) reported that light traps would provide the investigator with a method to sample fly populations. This technique, and/or one that may be more amenable to use by farmers and milk inspectors, should be used to monitor public health-nuisance levels. Keiding (1976) listed the advantages and disadvantages of using the various sampling methods currently used. Miller et al. (1993b.) used light traps similar to those suggested by Pickens et al. (1972) to monitor house fly populations on dairy farms. In a later study Miller et al. (1993a.) used cylindrical steel traps baited with Beltsville bait (Pickens & Miller 1987) to survey for house flies on dairy farms. Geden et al. (1992) used three methods to survey for house flies on dairy farms, i.e. (1) fly fecal and vomit spots were counted on 10 white index cards (7.6 X 12.7 cm) that were attached to walls, rafters, and support posts throughout the barns (Lysyk & Axtell 1985), (2) 10 visual counts made of live flies resting per 0.1 m2 in areas adjacent to the spot cards, and (3) an index of the number of flies in the milk room. The spots on the index cards is probably the most suitable method to be used when large numbers of dairy farms are to be surveyed.

(3) Available Components. Current chemical control in dairies is compromised by limited availability of new synthetic pyrethroids. Another problem is that of resistance as discussed earlier in this report. More effort must be made, similar to previous studies (Batth & Stalker 1970, Bailey et al., 1971; Georghiou & Bowen 1966; Hansens &

Anderson 1970; Mathis et al. 1972), to identify resistant fly populations on a regional basis; and with the assistance of extension, control programs taking this information into consideration must be implemented. The dollars wasted by farmers who are trying to control resistant fly populations have not been evaluated.

(4) Biological-Ecological. In a bibliography on the house fly, West and Peters (1973) included 5,720 references on every imaginable aspect of the fly: its genetics, physiology, ecology, etc. What is needed is an evaluation of the already available information concerning the bionomics of the house fly as it relates to the establishment of an IPM program for the dairy industry.

(5) Pest-Host Models. Systems models research in the house fly has thus far been neglected. The information needed to construct computer models is already available but it has yet to be synthesized and integrated into workable model at least for house flies around dairies. More research effort should be put into the evaluation of various methods to control house flies and support the development of computer models to assist in evaluating trapping potential and effectiveness (Hienton 1974; Weidhaas & Haile 1978).

(6) Area/lndividual Producer Control. The major responsibility for house fly control, because of the ubiquity and dispersal of adults, should be at the individual producer level.

(7) Legal Barriers. FIFRA 1988 has had a significant negative impact, reducing the number of pesticides currently available to help manage animal arthropod pests. Little new chemistry is currently in development for arthropod pests affecting animals. This situation emphasizes the need to utilize an IPM approach to protect animals from pest problems, to optimize management of registered pesticides to extend their useful life span, and minimize the development of pesticide resistance. Work is needed to maintain registration of existing pesticides. The IR-4 program should be utilized to address present and future animal pesticide registration and re-registration needs, since livestock, poultry and companion animals are considered "minor crops."

References Cited

Anonymous. 1976. *In* ARS National Research Prog., NRP 20480. Control of insects affecting livestock. USDA. 99 pp.

Axtell, R. C. 1963. Effects of *Macrochelidae* (Acarina: Mesostigmata) on house fly production from dairy cattle manure. J. Econ. Entomol. 56: 317-321.

Bailey, D. L., G. C. Labrecque & T. L. Whitfield. 1970. Insecticides in dry sugar baits for control of house flies in Florida dairy barns. J. Econ. Entomol. 63: 2000-2001.

Bailey, D. L., G. C. Labrecque & T. L. Whitfield. 1971. Resistance of house flies (Diptera: Muscidae) in Florida to trichlorfon and dichlorvos formulated in sugar baits. Can. Ent. 103: 853-856.

Batth, S. S. & J. M. Stalker. 1970. A survey of Canadian populations of the house fly for resistance to insecticides. J. Econ. Entomol. 63: 1947-1950.

Bay, E. C., C. O. Berg, H. C. Chapman & E. F. Legner. 1976. Biological control of medical & veterinary pests, pp. 457-479. *In* C. B. Huffaker & P. S. Messenger (eds.), Theory and Practice of Biol. Control. Academic Press, NY.

Black, W. C. & E. S. Krafsur. 1986. Temporal and spatial trends in allozyme frequencies in house fly populations, *Musca domestica* L. Theor. and Appl. Genetics. 71: 673-681.

Bushland, R. C. 1971. Sterility principle for insect control, pp. 8-11. *In* Sterility Principle for Insect Control or Eradication. Internat. Atom. Energy Agency, Vienna. 542 pp.

Carlson, D. A., M .S. Mayer, D. L. Shihacek, J. D. James, M. Beroza & B. A. Bierl. 1971. Sex attractant pheromone of the house fly: isolation, identification and synthesis. Science. 174: 76-78.

Clausen, C. P. (ed.). 1978. Introduced parasites and predators of arthropod pests and weeds. USDA Agr. Handbook No. 480. 545 pp.

Freeborn, S. B., W. M. Regan & A. H. Folger. 1925. The relation of flies and fly sprays to milk production. J. Econ. Entomol. 18: 779-790.

Freeborn, S. B., W. M. Regan & A. H. Folger. 1928. The relation of house flies and fly sprays to milk production. J. Econ. Entomol. 21: 494-501.

Fye, R. L., C. W. Woods, A. B. Borkovec, & P. H Terry. 1973. Chemisterilants for the house fly: further screening tests. J. Econ. Entomol. 66: 38-44.

Geden, C. J., D. A. Rutz, R. W. Miller & D. C. Steinkraus. 1992. Suppression of house flies (Diptera: Muscidae) on New York and Maryland dairies using releases of *Muscidifurax raptor* (Hymenoptera: Pteromalidae) in an integrated management program. Environ. Entomol. 21: 1419-1426

Georghiou, G. P. & W. R. Bowen. 1966. An analysis of house fly resistance to insecticides in California. J. Econ. Entomol. 59: 204-214.

Greene, G. L., J. A. Hogsette & R. S. Patterson. 1989. Parasites that attack stable fly and house fly (Diptera: Muscidae) puparia during the winter on dairies in northwestern Florida. J. Econ. Entomol. 82: 412-415.

Hanec, W. A. 1956. A study of the environmental factors affecting the dispersion of house flies (*Musca domestica* L.) in a dairy community near Fort Whyte, Manitoba. Can. Entomol. 88: 516-519.

Hansens, E. J. & W. F. Anderson. 1970. House fly control and insecticidal resistance in New Jersey. J. Econ. Entomol. 63: 1924-1926.

Harrington, E. P., D. B. Weingart, P. E. Kaufman, J. K. Waldron, W. G. Smith & D. A. Rutz. 1997. Pest and Pesticide use assessment and personal protective equipment use for dairy cattle production systems in New York state for 1997. NAPIAP Report. Cornell University. 61 pp.

Harris, C. R., S. A. Turnbull, J. W. Whistlecraft & G. A. Surgeoner. 1982. Multiple resistance shown by field strains of house fly, *Musca domestica* (Diptera: Muscidae), to organochlorine, organophosphorus, carbamate, and pyrethroid insecticides. Can. Entomol. 114: 447-454.

Hienton, T. E. 1974. Summary of investigations of electric insect traps. USDA Tech. Bull. No. 1498. Washington, DC. 136 pp.

Hogsette, J. A. & P. G. Koehler. 1992. Comparative toxicity of aqueous solutions of boric acid and Polybor 3 to house flies (Diptera: Muscidae). J. Econ. Entomol. 85: 1209-1212.

Hogsette, J. A. & R. D. Jacobs. 1993. Evaluation of AO-159 house fly bait stations in large caged-layer houses in north central Florida. Livestock Insect Worker's Conf. Report.

Jenkins, D. W. 1964. Pathogens, parasites and predators. Bull. WHO Suppl. 30. 150 pp.

Keiding, J. 1976. The house fly - biology and control. WHO, VBC, 76.650. pp. 1-82.

Krafsur, E. S., J. M. Helm & W. C. Black. 1992. Genetic diversity at electrophoretic loci in the house fly *Musca domestica* L. Biochemical Genetics. 30: 317-328.

Labrecque, G. C. & D. E. Weidhaas. 1970. Advantages of integrating sterile-male releases with other methods of control against house flies. J. Econ. Entomol. 63: 379-382.

Lazarus, W. F., D. A. Rutz, R. W. Miller & D. A. Brown. 1989. Costs of existing and recommended manure management practices for house fly and stable fly (Diptera: Muscidae) control on dairy farms. J. Econ. Entomol. 82: 1145-1151.

Legner, E. F., R. D. Sjogren & M. Hall. 1974. The biological control of medically important arthropods. CRC Critical Reviews in Environmental Control. 4: 85-113.

Legner, E. F. & G. S. Olton. 1970. Worldwide survey and comparison of adult predator and scavenger insect populations associated with domestic animal manure where livestock is artificially congregated. Hilgardia. 40:225-265.

Legner, E. F. & G. S. Olton. 1971. Distribution and relative abundance of dipterous pupae and their parasitoids in accumulations of domestic animal manure in the southwestern United States. Hilgardia. 40: 505-535.

Long, S. J., P. E. Kaufman, J. K. Waldron & D. A. Rutz. 1998. Evaluation of temperature on searching ability and parasitism rates of *Muscidifurax raptorellus* and *Muscidifurax raptor* in dairy barns and calf hutches. 1997 New York State Livestock and Field Crops Project Reports Relating to IPM. NYS IPM Publication 317. pp. 51-55.

Lysyk, T. J. 1993. Adult resting and larval development sites of stable flies and house flies (Diptera: Muscidae) on dairies in Alberta. J. Econ. Entomol. 86: 1746-1753.

Lysyk, T. J. & R. C. Axtell. 1985. Comparison of baited jug trap and spot cards for sampling house fly, *Musca domestica* (Diptera: Muscidae) populations in poultry houses. Environ. Entomol. 14: 815-819.

MacDonald, R. S., G. A. Surgeoner, K. R. Solomon & C. R. Harris. 1983. Development of resistance to permethrin and dichlorvos by the house fly (Diptera: Muscidae) following continuous and alternating insecticide use on four farms. Can. Entomol. 115: 1555-1561.

Mathis, W., A. D. Flynn & H. F. Schoof. 1972. House fly resistance in the Savannah, Georgia, area and its influence of chemical-control measures. J. Econ. Entomol. 65: 748-751.

Meyer, J. A., B. A. Mullens, T. L. Cyr & C. Stokes. 1990. Commercial and naturally occurring fly parasitoids (Hymenoptera: Pteromalidae) as biological control agents of stable flies and house flies (Diptera: Muscidae) on California dairies. J. Econ. Entomol. 83: 799-806.

Meyer, J. A., G. P. Georghiou & M. K. Hawley. 1987. House fly (Diptera: Muscidae) resistance to permethrin on southern California dairies. J. Econ. Entomol. 80: 636-640.

Meyer, J. A. & J. J. Petersen. 1983. Characterization and seasonal distribution of breeding sites of stable flies and house flies (Diptera: Muscidae) on eastern Nebraska feedlots and dairies. J. Econ. Entomol. 76: 103-108.

Meyer, J. A. & T. A. Shultz. 1990. Stable fly and house fly breeding sites on dairies. Calif. Agric. 44: 28-29.

Miller, R. W. 1970. Larvicides for fly control -- a review. Bull. Entomol. Soc. Amer. 16: 154-158.

Miller, R. W. 1993. The influence of dairy operations on the urban fly problem. *In* G. D. Thomas & S. R. Skoda (eds.), Rural Flies in the Urban Environment. Res. Bull. No. 317, Univ. of Nebraska, Lincoln, NB. pp. 25-33.

Miller, R. W. 1994. Inhibition of house flies and stable flies (Diptera: Muscidae) in field-spread dairy bedding from cattle treated with diflubenzuron boluses. J. Econ. Entomol. 87: 402-404.

Miller, R. W., D. A. Hall, R. E. Williams, L. G. Pickens & K. A. Doisy. 1991. Diflubenzuron boluses for fly control on dairy cattle. J. Agric. Entomol. 8: 117-126.

Miller, R. W. & J. A. Miller. 1994. Pyriproxyfen boluses for control of fly larvae. J. Agric. Entomol. 11:39-44.

Miller, R. W., L. G. Pickens & W. E. Potts. 1993a. Comparison of traps and an integrated program to manage house flies and stable flies on dairy farms. J. Agric. Entomol. 10: 189-196.

Miller, R. W. & D. A. Rutz. 1990. Survey of house fly pupal parasitoids on dairy farms in Maryland & New York, pp 59-66. *In* D. A. Rutz & R. S. Patterson (eds.), Biocontrol of Arthropods Affecting Livestock & Poultry. Westview Press, Boulder, CO. 316 pp

Miller, R. W., D. A. Rutz, L. G. Pickens & C. J. Geden. 1993b. Evaluation of traps and the parasitoid *Muscidifurax raptor* Girault and Sanders to manage house flies and stable flies on dairy farms. J. Agric. Entomol. 10: 9-19.

Miller, R. W. & E. T. Schmidtmann. 1985. Feeding of cyromazine (Larvadex¨) to dairy calves for the control of house flies. J. Agric. Entomol. 2: 313-316.

Miller, R. W. & E. C. Uebel. 1974. Juvenile hormone mimics as feed additives for control of the face fly and house fly. J. Econ. Entomol. 67: 69-70.

Morgan, N. O. & L. G. Pickens. 1978. *In* Surveillance and Collection of Arthropods of Veterinary Importance. USDA Handbook No. 518. pp. 72-76.

Morgan, N. O., L. G. Pickens & R. W. Miller. 1972. Doorway curtains help exclude flies from dairy farms. J. Econ. Entomol. 65: 1061-1063.

Morgan, N. O., L. G. Pickens & R. W. Thimijan. 1970. House flies and stable flies captured by two types of traps. J. Econ. Entomol. 63: 672-673.

Morgan, P. B., R. S. Patterson & G. C. Labrecque. 1976. Controlling house flies at a dairy installation by releasing a protelean parasitoid *Spalangia endius* (Hymenoptera: Pteromalidae). J. Ga. Entomol. 11: 39-43.

Morgan, P. B. & R. S. Patterson. 1977. Sustained release of *Spalangia endius* to parasitize field populations of three species of filth breeding flies. J. Econ. Entomol. 70: 450-452.

Mulla, Mir S., Y. S. Hwang & H. Axelrod. 1977. Attractants for synanthropic flies: chemical attractants for domestic flies. J Econ. Entomol. 70:644-648.

Mullens, B. A. 1985. Host age, sex, and pathogen exposure level as factors in the susceptibility of *Musca domestica* to *Entomophthora muscae*. Entomol. Exp. Appl. 37: 33-39.

Mullens, B. A. & J. L. Rodriguez. 1992. Effects of disodium actaborate tetrahydrate on survival, behavior, and egg viability of adult muscoid flies (Diptera: Muscidae). J. Econ. Entomol. 85: 137-143.

Pal, R. & L. E. LaChance. 1974. The operational feasibility of genetic methods for control of insects of medical and veterinary importance. Ann. Rev. Entomol. 19: 269-291.

Partridge, M. S., W. G. Smith & D. A. Rutz. 1991. Pesticide and pesticide use assessment for dairy cattle and cabbage production systems in New York State for 1991. NAPIAP Report. Cornell University. 65 pp.

Pawson, B. M. & J. J. Petersen. 1988. Dispersal of *Muscidifurax zaraptor* (Hymenoptera: Pteromalidae), a filth fly parasitoid, at dairies in eastern Nebraska. Environ. Entomol. 17: 398-402.

Petersen, J. J. & J. A. Meyer. 1983. Observations on the overwintering pupal parasites of filth flies associated with open silage in eastern Nebraska. Southwest. Entomol. 8: 219-225.

Petersen, J. J., J. A. Meyer, D. A. Stage & P. B. Morgan. 1983. Evaluation of sequential releases of *Spalangia endius* (Hymenoptera: Pteromalidae) for control of house flies and stable flies (Diptera: Muscidae) associated with confined livestock in eastern Nebraska. J. Econ. Entomol. 76: 283-286.

Pickens, L. G., E. T. Schmidtmann & R. W. Miller. 1994. How to control house and stable flies without using pesticides. USDA, ARS, Agric. Info. Bull. No. 673. 14 pp.

Pickens, L. G. & G. D. Mills, Jr. 1993. Solar-powered electrocuting trap for controlling house flies and stables flies (Diptera: Muscidae). J. Med. Entomol. 30: 872-877.

Pickens, L. G., N. O. Morgan, J. G. Hartsock & J. W. Smith. 1967. Dispersal patterns and populations of the house fly affected by sanitation and weather in rural Maryland. J. Econ. Entomol. 60: 1250-1255.

Pickens, L. G., N. O. Morgan & R. W. Miller. 1972. Comparison of traps and other methods for surveying density of populations of flies in dairy barns. J. Econ. Entomol. 65: 144-145.

Pickens, L. G. & R. W. Thimijan. 1986. Design parameters that affect the performance of UV-emitting traps in attracting house flies (Diptera: Muscidae). J. Econ. Entomol. 79: 1003-1009.

Plapp, F. W. 1976. Biochemical genetics of insecticide resistance. Ann. Rev. Entomol. 21: 179-197.

Roberts, D. W. & M. A. Strand. 1977. Bull. WHO 55 (Suppl. 1). 419 pp.

Schmidtmann, E. T. 1988. Exploitation of bedding in dairy outdoor calf hutches by immature house and stable flies (Diptera: Muscidae). J. Med. Entomol. 25: 484-488.

Schmidtmann, E. T. 1991. Suppressing immature house and stable flies in outdoor calf hutches with sand, gravel, and sawdust bedding. J. Dairy Sci. 74: 3956-3960.

Schmidtmann, E. T., R. W. Miller & R. Muller. 1989. Effect of experimental bedding treatments on the density of immature *Musca domestica* and *Stomoxys calcitrans* (Diptera: Muscidae) in outdoor calf hutches. J. Econ. Entomol. 82: 1134-1139.

Schmidtmann, E. T., D. W. Watson & P. A. W. Martin. 1993. Bacterial control of flies in livestock operations, pp 47-53. *In* R. D. Lumsden & J. L. Vaughn, (eds.), Pest Management: Biologically Based Technologies. Am. Chem. Soc.

Scott, J. C., R. T. Roush & D. A. Rutz. 1989. Insecticide resistance of house flies from New York dairies (Diptera: Muscidae). J. Agric. Entomol. 6: 53-64.

Six, D. L. & B. A. Mullens. 1993. Seasonal prevalence and attempted introduction of members of the *Entomophthora muscae* complex on southern California dairies. Livestock Insect Worker's Conf. Report.

Smith, L. & D. A. Rutz. 1991a. Seasonal and relative abundance of hymenopterous parasitoids attacking house fly pupae at dairy farms in central New York. Environ. Entomol. 20: 661-668.

Smith, L. & D. A. Rutz. 1991b. Relationship of microhabitat to incidence of house fly (Diptera: Muscidae) immatures and their parasitoids at dairy farms in central New York. Environ. Entomol. 20: 669-674.

Smith, L. & D. A. Rutz. 1991c. Microhabitat associations of hymenopterous parasitoids that attack house fly pupae at dairy farms in central New York. Environ. Entomol. 20: 675-684.

Smith, L., D. A. Rutz & G. A. Scoles. 1989. Influences of habitat and temperature on dispersal behavior of two pteromalid parasitoids of house flies during an inundative release at a dairy barn. Med. Vet. Entomol. 3: 169-178.

Steinkraus, D. C., C. J. Geden & D. A. Rutz. 1993. Prevalence of *Entomophthora muscae* (Cohn) *Fresenius* (Zygomycetes: Entomophthoracea) in house flies (Diptera: Muscidae) on dairy farms in New York and introduction of epizootics. Biological Control. 3: 93-100.

Thimijan, R. W., L. G. Pickens & N. O. Morgan. 1970. A trap for house flies. J. Econ. Entomol. 63: 1030-1032.

Thimijan, R. W., L. G. Pickens, N. O. Morgan & R. W. Miller. 1972. House fly capture as a function of number of traps in a dairy barn. J Econ. Entomol. 65: 876-877.

Uebel, E. C., P. E. Sonnet & R. W. Miller. 1976. House fly sex pheromone enhancement of mating strike activity by combination of (z)-9-tricosene with branched saturated hydrocarbons. Environ Entomol. 5: 905-908.

United States Department of Agriculture. 1978. Estimated losses and production costs attributed to insects and related arthropods-1976. *In* Coop. Plant Pest Report, March 17, 1978. 3(11): 91-117.

VanKirk, J. R., P. L. Sutton, D. W. Watson & D. A. Rutz. 1994. Demonstration of IPM for dairy barn fly pests. In. 1993 Reports: IPM Research, Development, and Implementation Projects, Dairy and Field Crops, NYS IPM Program 2/94, IPM Publication # 312.

Watson, D. W. & J. J. Petersen. 1993. Seasonal activity of *Entomophthora muscae* (Zygomycetes: Entomophthoracea) in *Musca domestica* L. (Diptera: Muscidae) with reference to temperature and relative humidity. Biological Control. 3: 182-190.

Watson, D. W., C. J. Geden, S. J. Long & D. A. Rutz. 1995. Efficacy of Beauveria bassiana for controlling the house fly and stable fly (Diptera: Muscidae). Biological Cont. 5: 405-411.

Watson, D. W., D. A. Rutz & S. J. Long. 1996. *Beauveria bassiana* and sawdust bedding for the management of the house fly, *Musca domestica* (Diptera: Muscidae) in calf hutches. Biological Cont. 7: 221-227.

eidhaas, D. E. & D. G. Haile. 1978. A theoretical model to determine the degree of trapping required for insect population control. Bull. Entomol. Soc. Amer. 24: 18-20.

West, L. S. & O. B. Peters. 1973. An Annotated Bibliography of *Musca domestica* Linn. Northern Michigan Univ.

Stable Fly

Description and Biology. The stable fly, *Stomoxys calcitrans (L)* isan important pest of confined livestock. Adults of both sexes are persistent biters, feeding on cattle and other warm-blooded animals. Sometimes known as the biting house fly or dog fly, this pest is not only found on livestock facilities, but miles from suitable larval habitat where adults often bite people and pets (Hogsette et al. 1987; Jones et al. 1987, 1991). Flies feed on the lower portions of animals, such as the legs of cattle, ankles of humans and ears of dogs. When stable flies attack humans or pets, livestock producers may presumptuously be given the blame, although large numbers of larvae can be found in suburban areas. Resulting economic and social consequences, including lawsuits, can be devastating (Hayes 1993).

Stable fly larvae feed and develop in fermenting, urine-soaked mixtures of manure and feed at feedlot perimeters, improperly composted vegetative materials, in moist feed spills, silage residues, and in large rolled hay bales (Scholl et al. 1981; Hall et al. 1982; Hogsette et al. 1987, Skoda et al. 1991). Historically recognized primarily as a pest around barns and stables, and subsequently in feedlot situations, stable flies have been found frequently on pastured livestock in recent years (Hogsette et al. 1989), possibly because of the increased hay residues from feeding or the increased use of large round bales of hay (Hall et al. 1982). However, in general, stored manure, soiled bedding and calf hutches are probably the most important sources of fly larvae under current dairy situations (Schmidtmann 1988, Meyer & Shultz 1990; Lysyk 1993a). At optimum temperatures of 28-32¡C, generation time is < 2 weeks (Larsen & Thompson 1940). Females may deposit ~ 500 eggs each, resulting in rapid increases in adult populations.

Adult stable flies require multiple blood meals to copulate and produce fertile eggs (Anderson 1978; Chia et al. 1981, Morrisson et al. 1982). Nectars may be used to supply energy for dispersal and host location but have no role in reproduction, although nectar sated flies may eschew blood meals (Jones et al. 1985, 1992). Stable flies may take three blood meals daily. Berry & Campbell (1985) discuss factors affecting feeding. Adults slash through the host's hide to create a pool of blood from which they feed for up to 4 minutes (Harris et al. 1974). The slashing process is very painful because the stable fly saliva contains no anesthetic. The stable fly is capable of transmitting several livestock diseases, notably anthrax, brucellosis, swine erysipelas, and equine infectious anemia.

Circumstantial evidence indicates that lumpy skin disease transmission to Israeli dairy cattle in 1989 may have been the result of wind-borne dispersal of infected flies from the Sinai or from Northern Egypt, following feeding on infected herds of cattle sheep or goats.

Economic Importance. Bovine responses to stable fly feeding range from behaviors that denote pain and irritation such as bunching, head throwing, and foot stamping (Wieman et al. 1992) to physiological changes including increased body temperatures, increased urinary nitrogen output, and elevated cortisol levels (Schwinghammer et al. 1986). Beef calves exposed to stable flies for 98 days weighed an average of 9 kg less than their unexposed counterparts (Campbell et al. 1977) and feed efficiency was decreased by 12.9%. Wieman et al. (1992) showed that decreased gain was attributable directly to fly activity (blood loss and defensive actions) and indirectly to host activity (bunching and heat stress). Drummond et al. (1981) estimated that milk production decreases 5% for a 6 month fly season, but losses probably vary widely depending on a variety of environmental factors as well as bovine behavior. Milk production increases attributed to fly control have been reported (Bruce & Decker 1958, Morgan & Bailie 1980; Block & Lewis 1986), but on well managed farms nutritional supplementation may obscure the damage potentially caused by stable flies (Chang & Kessler 1961; Miller et al. 1973). Unlike house flies, transmission of Cornnebacterium pseudotuberculosis in Israel (Braverman et. Al. 1998) is not accomplished be stable flies following membrane feeding on a mixture of bacterial broth and dairy cattle blood.

Dispersal Stable flies are relatively strong fliers that stay close to the ground during short distance dispersal. Flight speed of stable flies has been estimated at a mean of 9.4 km per 24 h in tethered 3 day-old laboratory flies, with maximum flight speed of approximately 29 km/24 h for both male and female flies (Bailey et al. 1973). Most flights are of short duration, generally downwind, with less than 10% of marked flies on traps as far as 0.9 km from their release site (Gersabeck & Merritt 1985). In studies intended to assess the flight of stable flies moving short distances, Gersabeck & Merritt (1983) found that 70% of the flies captured on traps (2:1 females:males) were below 0.6 m. Flies caught 3-8 km from the nearest known bloodmeal source, in Kansas, were chronologically older than flies of the same physiological age caught near cattle feedlots (Jones et al. 1998) indicating that dispersal may not be the result of cues related to reproduction. In Florida, Hogsette and Ruff (1985) demonstrated movement of 225 km by marked wild flies associated with a frontal system. Whether their work demonstrates directed movement or inadvertent movement via air currents is unknown. Krafsur et al., studying stable flies collected from Minnesota and Iowa showed that substantial gene flow occurs among populations in these contiguous states. Similar data was developed by Szlanski et al (1996). Analysis of isozymes from dispersing stable flies indicates that these insects may move with frontal systems for even greater distances than the 225 km previously mentioned (Jones et al. 1991). Wind from local fronts may be responsible for rapid increases of stable fly populations on the southern shores of Lake Superior (Broce 1993).

Methods of Control. The stable fly is a difficult insect to control under many situations because its larval habitats can be so diverse and so widely spread. Little work on the effect of current manure and bedding removal and disposal systems on stable fly population dynamics has been completed, except as noted below. Control of flies on cattle and in barns is relatively easy on a daily basis, but long term control is much more difficult. Stable flies are also pests of dairy cattle on pastures, especially in areas where soiled and moist hay residues allow fly larvae to develop. There are no good control methods available under these circumstances. Pickens et al. (1994) describe a variety of unconventional techniques that work under some circumstances. Many of these do not have producer confidence, and are not used except under experimental conditions.

(1) Conventional Insecticides. In general, chemical pesticides have the greatest acceptance for control of stable flies by dairy farmers because of the low time required to use them and because dead flies can be readily seen after spraying or fogging. Current chemical control measures for the stable fly on lactating dairy cattle include the use of insecticides either directly to cattle or on surfaces where the flies rest. Pyrethroids, organophosphates, insect growth regulators are all available for use. These treatments are generally effective for confined (as opposed to grazed) cattle if treatments are practiced on a regular basis. Ear tags are less effective on stable flies than on non-resistant horn fly populations. Organophosphate resistance has been reported in stable fly populations in Kansas, but the extent of the phenomenon is still unknown (Cilek & Greene 1994). Sanitation and larvicidal treatments are practiced to a limited extent to control stable fly larval breeding. These practices result in lower stable fly populations if carried out regularly and conscientiously. Cyromazine, administered orally at 0.5 and 1.0 mg/kg, is excreted from dairy calves primarily through the urine. At these rates it prevents the development of stable fly and house fly larvae to pupation.

An attractant-toxicant system-using fiberglass (Alsynite) panels treated with permethrin for stable fly control was developed by Meifert et al. (1978), but is rarely used.

(2) Area Wide Control. Most stable fly control on dairy cattle has been practiced by individual farmers. Area-wide control of stable flies can be accomplished, however, as demonstrated by a USDA pilot sterile-male release, selective insecticide treatment program conducted on the island of St. Croix (Patterson et al. 1981) in which 99.9% of wild flies were eliminated. Unfortunately, it has not been possible to extend this type of program to other regions due to the ability of males to take blood-meals.

(3) Trapping Methods. Rugg (1982) successfully reduced stable fly populations at a zoo, using Williams traps (Williams 1973). Unfortunately the procedure was too time consuming and probably too costly for practical use. Modifications of this trap (Broce 1988) are now sold commercially and generally used for sampling. Phenyl propanoid treated sticky traps in corn fields adjacent to dairy barns had significantly more stable flies on them than other traps (Hammack and Hesler. 1996). These compounds are in floral nectaries, again raising the idea of using floral attractants as an adjunct to other methods used to attract stable flies.

(4) Insect Growth Regulators(. Diflubenzuron has shown limited effectiveness in dairy situations (Miller 1994), leaving methoprene as the only available IGR.

(5) Sanitation. Sanitation is by far the soundest method of stable fly control, but also one of the most difficult in practice. Because of the labor required, farmers tend not to clean out bedding in cattle holding pens, especially calf pens. Spilled silage and hay are also prime sites for stable fly oviposition. Lysyk (1993a) found that manure mounds, general lots and barn interiors were most likely to be utilized by fly larvae in Alberta. Calf pens, a consistent source of stable fly larvae (Schmidtmann 1988), can be managed with alternative bedding sources (Schmidtmann 1991) to decrease the production of stable flies.

(6) Biological Control. Petersen (1989) reviewed the literature on pathogens, predators and parasites of the stable fly. Except for a few of the pathogens most of this work was done prior to the 1930s with little success. Although *Bacillus thuringiensis* has been shown to be active against the stable fly, it is not available for use, although under certain circumstances, it might be useful (Gingrich 1965). Generally stable flies are considered aberrant hosts for known pathogens. Although predators almost certainly have the greatest effect of all biological control agents on larval populations of stable flies, with larval mortality approaching 90% in California and Missouri (Legner & Brydon 1966; Smith et al. 1985, respectively), with about half resulting from predator activity. However, because of the difficulties in raising sufficient predators, and the often transient nature of the substrate through which they need to search, their potential is viewed as limited. Greater attention is currently being placed on parasites, or more properly parasitoids, primarily Pteromalid wasps (Meyer et al. 1990). Although certain species show great promise for house fly control, there is little evidence that a single species will suffice for stable fly control throughout North America. Anecdotal evidence for this indicates that releases of *Spalangia nigroaenea, S. cameroni,* and *S. endius* all are effective under some circumstances. Currently, experimental tests with several species are being carried out on beef cattle feedlots, but the only commercial scale operation based strictly on parasitoids is underway in Kansas (G. Greene, KSU), associated with a feedlot IPM demonstration.

(7) Integrated Control Programs. Currently, a large scale IPM program for dairies exists in New York State, where the economic aspects have been proven to be favorable (Lazarus et al. 1989). Elsewhere, integrated programs have proven successful on an experimental basis (Miller et al. 1993), but have not been continued without support from scientists.

Current Support. There are no SYs (CSRS) assigned to stable fly control on dairies in the United States at this time. In Canada, Lysyk has a 25% commitment in this area. Although there is some overlap in biology and behavior, the Canadian research is in Alberta, and has limited seasonal relevance to California and Florida dairies.

Summary of Research and Extension Needs

With increasing urban encroachment into farm regions has come the familiar portrait of a grower suddenly under legal constraints to keep fly populations low under a locally defined nuisance level. Unlike economic injury thresholds, these are based on peoples perceptions or emotions rather than a strictly defined set of figures developed by observing the effects of flies on cattle. Instead, what we now must learn to develop are action levels based on how flies of a given population will disperse to areas not associated with cattle. It can safely be assumed that this level will be reached before a biologically determined economic threshold has.

Most of the tools required for studying these action levels have been available for some time. Sampling procedures, the basic biology of the stable fly, and control components are all reasonably well defined. Less well defined for dairies in much of the United States are models for population development on dairies. Even more crucial, is the lack of sufficient information on stable fly dispersal. It will be very difficult for dairy farmers to remain viable in periurban situations without data on: 1) The speed of movement with and against winds. 2) The relationship of short and long distance dispersal to weather, population pressure, and season. 3) Possible barriers to directed or random dispersal.

References Cited

Anderson, J. R. 1978. Mating behavior of *Stomoxys calcitrans*: effects of a blood meal on the mating drive and its necessity as a prerequisite for proper insemination of females. J. Econ. Entomol. 71: 379-386.

Bailey, D. L., T. L. Whitfield & B. J. Smittle. 1973. Flight and dispersal of the stable fly. J. Econ. Entomol. 66: 410-411.

Berry, I. L. & J. B. Campbell. 1985. Time and weather effects on daily feeding patterns of stable flies (Diptera: Muscidae). Environ. Entomol. 14:336-342.

Block, E. & D. J. Lewis. 1986. Efficacy of insecticidal ear tags on fly control and mile production of dairy cows. Can. J. Anim. Sci. 66: 47-51.

Braverman Y, Chizov-Ginzburg A, Saran A, Winkler M. 1998. The role of houseflies (*Musca domestica*) in transmitting the bacterium *Corynebacterium pseudotuberculosis* in Israeli dairy herds. Israel Journal of Veterinary Medicine 53(1):7-15.

Broce, A. B. 1988. An improved trap for stable flies, *Stomoxys calcitrans* (Diptera: Muscidae). J. Med. Entomol. 25: 406-409.

Broce, A. B. 1993. Dispersal of house and stable flies. *In* G. D. Thomas & S. R. Skoda (eds.), Rural Flies in the Urban Environment. North Central Regional Research Publ. # 335.

Bruce, W. N. & G. C. Decker. 1958. The relationship of stable fly abundance to milk production in dairy cattle. J. of Econ. Entomol. 51:269-274.

Campbell, J. B., R. G. White, J. E. Wright, R. Crookshank & D. C. Clanton. 1977. Effects of stable flies on weight gains and feed efficiency of calves on growing or finishing rations. J. Econ. Entomol. 70: 592-594.

Chang, T. H. & E. M. Kessler. 1961. A three year study on the effect of fly control on milk production by selection and randomized dairy herds. J. Econ. Entomol. 54: 751-757.

Chia, L. S., J. A. Baxter & P. E. Morrison. 1981. Quantitative relationship between ingested blood and follicular growth in the stable fly, *Stomoxys calcitrans*. Can. J. Zool. 60: 1917-1921.

Cilek, J. E. & G. L. Greene. 1994. Stable fly (Diptera: Muscidae) resistance in Kansas cattle feedlots. J. Econ. Entomol. 87:275-79.

Drummond, R.O., G. Lambert, H. E. Smalley, Jr. & C. E. Terrill. 1981. Estimated losses of livestock to pests. *In* D. Pimentel (ed.), Handbook of Pest Management in Agriculture. Vol. 1. CRC Press, Inc. Boca Raton, FL.

Gersabeck, E.F. & R. W. Merritt. 1983. Verical and temporal aspects of Alsynite panel sampling for adult *Stomoxys calcitrans* (L.) (Diptera: Muscidae). Fla. Entomol. 66: 222-227.

Gersabeck, E. F. & R. W. Merritt. 1985. Dispersal of adult *Stomoxys calcitrans* (L.) (Diptera: Muscidae) from known immature developmental areas. J. Econ. Entomol. 78: 617-621.

Gingrich, R. E. 1965. *Bacillus thuringiensis* as a feed additive to control dipterous pests of cattle. J. Econ. Entomol. 58: 363-4.

Greene, G. G., J. A. Hogsette & R. S. Patterson. 1989. Parasites that attack stable fly and house fly (Diptera: Muscidae) puparia during the winter on dairies in northwestern Florida. J. Econ. Entomol. 82: 412-415.

Hall, R. D., G. D. Thomas & C. E. Morgan. 1982. Stable fly, *Stomoxys calcitrans* (L.) breeding in large round hay bales: initial associations (Diptera: Muscidae). J. Kans. Entomol. Soc. 55: 617-620.

Hammack L, Hesler LS. 1996. Phenylpropanoids as attractants for adult *Stomoxys calcitrans* (Diptera: Muscidae). The Entomological Society of America 33(5):859-862

Harris, R. L., J. A. Miller & E. D. Frazar. 1974. Horn flies and stable flies: feeding activity. Ann. Ent. Soc. Am. 67: 891-894.

Hayes, D. K. 1993. Legal aspects of rural flies in the urban environment. *In* G. D. Thomas & S. R. Skoda (eds.), Rural Flies in the Urban Environment. North Central Regional Research Publ. # 335.

Hogsette, J. A., J. P. Ruff & C. J. Jones. 1989. Dispersal behavior of stable flies. *In* J. J. Petersen & G. L. Greene (eds.), Current Status of Stable Fly Research. Misc. Publ. Entomol. Soc. Am. #74.

Hogsette, J. A., J. P. Ruff & C. J. Jones. 1987. Stable fly biology and control in northwest Florida. J. Agric. Entomol. 4: 1-11.

Hogsette, J. A. & J. P. Ruff. 1985. Stable fly (Diptera: Muscidae) migration in northwest Florida. Environ. Entomol. 14: 170-175.

Jones, C. J., R. S. Patterson & D. E. Milne. 1987. Electrophoretic comparisons of isozymes from selected populations of *Stomoxys calcitrans* (Diptera: Muscidae). J. Med. Entomol. 24: 54-60.

Jones, C. J., J. A. Hogsette, R. B. Patterson & D. E. Milne. 1985. Effects of natural saccharide and pollen extract feeding on stable fly (Diptera: Muscidae) longevity. Environ. Entomol. 14: 223-227.

Jones, C. J., D. E. Milne, R. S. Patterson, E. T. Schreiber & J. A. Milio. 1992. Nectar feeding by *Stomoxys calcitrans* (Diptera: Muscidae): Effects on egg production and survival. Environ. Entomol. 21: 141-147.

Jones, C. J., J. A. Hogsette, R. B. Patterson, D. E. Milne, G. D. Propp, J. F. Milio, L. G. Rickard & J. P. Ruff. 1991. Origin of stable flies (Diptera: Muscidae) on West Florida beaches: electrophoretic analysis of dispersal. J. Med. Entomol. 28: 787 795.

Jones, C. J., J. A. Hogsette, S. A. Isard, Y-J. Guo, G. Greene and A. Broce. 1998**.** Using phenology to detect dispersal of stable flies in Western Kansas. Proc. Am. Meteorological Soc. 13th Conference on Biometeorology and Aerobiology. pp348-351.

Larsen, E. B. & M. Thomsen. 1940. The influence of temperature on the development of some species of Diptera. Vidensk. Medd. fra Dansk naturh. Foren. Bd. 104: 1 75.

Lazarus, W. F., D. R. Rutz, R. W. Miller & D. A. Brown. 1989. Costs of existing and recommended manure management practices for house fly and stable fly (Diptera: Muscidae) control on dairy farms. J. Econ. Entomol. 82: 1145-1151.

Legner, E. F. & H. W. Brydon. 1966. Suppression of dung inhabiting fly populations by pupal parasites. Ann. Entomol. Soc. Am. 59: 638-651.

Lysyk, T. J. 1993a. Adult resting and larval developmental sites of stable flies and house flies (Diptera: Muscidae) on dairies in Alberta. J. Econ. Entomol. 86: 1746 1753.

Meifert, D. W., R. S. Patterson, T. Whitfield, G. C. Labrecque & D. E. Weidhaas. 1978. A unique attractant-toxicant system to control stable fly populations. J. Econ. Entomol. 71: 290-292.

Meyer, J. A., B. A. Mullens, T. L. Cyr & C. Stokes. 1990. Commercial and naturally occurring fly parasitoids (Hymenoptera: Pteromalidae) as biological control agents of stable flies and house flies (Diptera: Muscidae) on California dairies. J. Econ. Entomol. 83: 799-806.

Meyer, J. A. & T. A. Schultz. 1990. Stable fly and house fly breeding sites on dairies. Cal. Agr. 44 (1): 28-29.

Miller, R. W., L. G. Pickens, N. O. Morgan, R. W. Thimijan & R. L. Wilson. 1973. Effect of stable flies on feed intake and milk production of dairy cows. J. Econ. Entomol. 66: 711-713.

Miller, R. W., L. G. Pickens & W. E. Potts. 1993. Comparisons of traps and an integrated program to manage house flies and stable flies on dairy farms. J. Agric. Entomol. 10: 189-196.

Miller, R. W. 1994. Inhibition of house flies and stable flies (Diptera: Muscidae) in field-spread dairy bedding from cattle treated with diflubenzuron boluses. J. Econ. Entomol. 87: 402-404.

Miller RW, Schmidtmann ET, Wauchope RD, Clegg CM, Herner AE, Weber H. 1996. Urine delivery of cyromazine for suppressing house and stable flies (Diptera: Muscidae) in outdoor dairy calf hutches. J. Econ. Entomol. 89(3):689-694.

Morgan, D. W. T. & H. D. Bailie. 1980. A field trial to determine the effect of fly control using permethrin on milk yields in dairy cattle in the UK. Vet. Record. 106: 121 123.

Morrison, P. E., K. Venkatesh & B. Thompson. 1982. The role of male accessory gland substance on female reproduction with some observations of spermatogenesis in the stable fly. J. Insect Physiol. 28: 607-614.

Patterson, R. S., G. C. Labrecque, D. F. Williams & D. E. Weidhaas. 1981. Control of the stable fly, *Stomoxys calcitrans* (Diptera: Muscidae), on St. Croix, U.S. Virgin Islands, Using integrated pest management measures. III. Field techniques and population control. J. Med. Entomol. 18: 203-210.

Petersen, J. J. 1989. Potential for biological control of stable flies associated with confined livestock, pp. 41-45 *In* J. J. Petersen & G. L. Greene (eds.), Current Status of Stable Fly (Diptera: Muscidae) Research. Misc. Publ. Etomol. Soc. America. 74. Lanham, MD.

Pickens, L. G., E. T. Schmidtmann & R. W. Miller. 1994. How to control house flies and stable flies without using pesticides. U. S. Department of Agriculture. Agriculture Information Bulletin # 673. 14 pp.

Rugg, D. 1982. Effectiveness of Williams traps in reducing the numbers of stable flies (Diptera: Muscidae). J. Econ. Entomol. 75: 857-859.

Schmidtmann, E. T. 1988. Exploitation of bedding in dairy outdoor calf hutches by immature house and stable flies (Diptera: Muscidae). J. Med. Entomol. 25: 484 488.

Schmidtmann, E. T. 1991. Suppressing immature house and stable flies in outdoor calf hutches with sand, gravel, and sawdust bedding. J. Dairy Sci. 74: 3956 3960.

Scholl, P. J., J. J. Petersen, D. A. Stage & J. A. Meyer. 1981. Open silage as an overwintering site for immature stable flies in eastern Nebraska. Southwest Entomol. 6: 253-258.

Schwinghammer, K. A., F. W. Knapp, J. A. Boling & K. K. Schillo. 1986. Physiological and nutritional response of beef steers to infestations of the stable fly (Diptera: Muscidae). J. Econ. Entomol. 79: 1294-1298.

Skoda, S. R., G. D. Thomas & J. B. Campbell. 1991. Developmental sites and relative abundance of immature stages of the stable fly (Diptera: Muscidae) in beef cattle feedlot pens in eastern Nebraska. J. Econ. Entomol. 84: 191-197.

Smith, J.P., R. D. Hall & G. D. Thomas. 1985. Field studies on mortality of the immature stages of the stable fly (Diptera: Muscidae). Environ. Entomol. 14: 881 890.

Szalanski, A.L., D.B. Taylor and R.D. Peterson II. 1996**.** Population genetics and gene variation of stable fly populations (Diptera: Muscidae) in Nebraska. J. Med. Entomol. 33: 413-42.

Wieman, G. A., J. B. Campbell, J. A. Deshazer & I. L. Berry. 1992. Effect of stable flies (Diptera: Muscidae) and heat stress on weight gain and feed efficiency of feeder cattle. J. Econ. Entomol. 85: 1835-1842.

Williams, D. F. 1973. Sticky traps for sampling populations of *Stomoxys calcitrans*. J. Econ. Entomol. 66: 1279-1280.

Yeruham I, Nir O, Braverman Y, Davidson M, Grinstein H, Haymovitch M, Zamir O. 1995. Spread of lumpy skin disease in Israeli dairy herds. Veterinary Record 137(4):91-93.

Face Fly

Description and Biology. The face fly, an introduced pest from Canada to the United States in 1952, has generated considerable interest and research during its 43-year stay in this country. The life cycle is similar to that of other dung inhabiting flies. The female lays its eggs in fresh bovine manure pats. The condition and type of manure is important in the development of the face fly. A number of studies have been done on larval competition and what effect the animals' diet has on subsequent fly production (Meyer et al. 1978, Moon 1980, D'amato et al. 1980). The egg is large with a respiratory mast that extends out of the manure pat. The eggs hatch and the larvae develop in manure pats. The mature third stage larvae migrate from the pat to pupate in the surrounding soil and the adults subsequently emerge from the pupae. An interesting set of papers on calcium and its role in defense from parasitoids in the pupal stage have been published. There is speculation that the hardness may play a role in a defense mechanism (Darlington et al. 1983, Darlington et al. 1984, Grodowitz & Broce 1983, Grodowitz et al. 1987, Grodowitz et al. 1987a, Grodowitz et al. 1987b, Krueger et al. 1987, Broce et al. 1988). The age structure of the face fly along with imaginal diapause has been studied by numerous workers (Easton & Lysyk 1986, VanGeem et al. 1983, VanGeem & Broce 1985, VanGeem & Broce 1986, Evans & Krafsur 1990, Krafsur et al. 1985, Krafsur et al. 1986, Moon & Kaya 1981, Moon et al. 1986, Schmidtmann & Pickens 1986, Schmidtmann & Redfern 1985). These studies along with Moon 1983, Moon 1986, have provided the basis for some simulation modeling. Several general papers have been published on the biology of the face fly (Yonggyun & Krafsur 1993, Meyer & Kopp 1984, Lysyk & Easton 1985).

Face flies do not have mouthparts capable of piercing the skin of their hosts so they are not primary blood feeders, however, studies have shown that the prestomal teeth are capable of predisposing the eye to infection by bacteria (Broce & Elzinga 1984), such as *Morexaella bovis.* Transmission of this and other bacteria by the face fly has been studied (Arends et al. 1982, Arends et al. 1984, Berkebile et al. 1981a, Berkebile et al. 1981b, Cheville et al. 1989, Coleman & Gerhardt 1987, Gerhardt et al. 1982, Glass & Gerhardt 1994, Glass et al. 1982, Hall 1984, Johnson et al. 1991, Schugart et al. 1979). These studies indicate that the face fly is capable of transmitting pinkeye, *Brucella abortus* and bovine Herpessvirus-1 in cattle.

Economic Importance. The threshold of one face fly is still the theoretical threshold but practically not attainable (Schugart et al. 1979). The combination of pinkeye and large numbers of face flies remain an economic concern. Financial losses due to pinkeye in feeder cattle have been demonstrated in Tennessee. Percent market price discounts and premiums for unusual characteristics were used as the measure to determine losses. Total face fly losses of $150 million still remain our best estimate of actual losses (Anonymous 1975). Schmidtmann conducted several studies in which he looked at behavioral stress but did not show significant losses (Schmidtmann et al. 1984, Schmidtmann & Berkebile 1985, Schmidtmann 1985a, 1985b).

Methods of Control. Chemical control has been the main tool used to manage the face fly. This is supported by the large number of chemical control-based studies using the latest compounds available. The approaches are mainly ear tags, boluses and residual activity or some variation of these methods (Hall & Foehse 1980, Herald & Knapp 1980, Herald & Knapp 1981, Hogsette & Ruff 1986, Knapp & Herald 1982, Knapp & Herald 1983, Knapp 1984, Knapp et al. 1985, Knapp & Herald 1980, Krafsur 1984, Miller et al. 1981, Miller et al. 1986, Miller & Miller 1994, Miller et al. 1984, Miller et al. 1984a, Miller et al. 1984b, Miller et al. 1984c, Miller et al. 1991, Moon et al. 1991, Pickens & Miller 1980, Scott et al. 1986, Skoda et al. 1987b, Williams & Westby 1980, Williams et al. 1981, Broce & Gonzaga 1987, Miller et al. 1990). Some work has been done on the use of traps as both sampling devises and population reduction tools. Traps have been better at sampling rather than population reduction (Easton 1979, Johnson & Campbell 1987, Pickens 1981, Pickens & Nafus 1982, Pickens 1990).

Biocontrol of the face fly has not been too successful probably due to the fact that it is an introduced pest and does not have many natural enemies. However, Skoda et al. (1987a) list a number of parasitoids of the face fly. *Heterotylenchus autumnalis* does show up periodically in epizootic proportions (Krafsur et al. 1983, Kaya & Moon 1980). Limited work has been done on utilizing dung beetles for the control of face flies. Moon et al. (1980) looked at the effects of dung beetles on face fly development. These approaches give moderate success but we are still looking for the non chemical approach to give us good levels of control.

Genetic manipulation is in its infancy in face fly research. A few workers are working at the molecular level but more resources are needed to make a major impact (Bryant et al. 1981, Krafsur & Black 1992, Mansour & Krafsur 1991, Darlington & Meyer 1988).

Current Support. There is very little research being conducted on the face fly. The total SY is less than 0.5.

Needs for Insect Pest Management

(1) Economic Injury Levels. There is a need for dairymen to keep face fly populations as low as possible mainly because of eye irritation and pinkeye. This is based on the threshold of 1.

(2) Sampling. Current sampling methods allow for estimation of face flies per head. With the threshold of one, the sampling method does not have to be extremely accurate.

(3) Control Components. These are adequate but more emphasis must be placed on resistance management of the face flies.

(4) Biological Control. Continue to search for good parasitoids and predators of the face fly for possible augmentation of existing populations.

(5) Systems Models. Join efforts with beef in the modeling efforts.

(6) Area Control vs. Individual Control. Some effort should continue on area control of the face fly. The aggregation for overwintering still offers potential control if the mechanism of aggregation was understood.

(7) Legal Barriers Involved. The biggest legal barrier is the limited number of pesticides available due to possible residues in milk.

(**8) Eradication vs. Regulation.** Not feasible.

References Cited

Anonymous. 1975. USDA 25:298.

Arends, J. J., P. B. Barto & R. E. Wright. 1982. Transmission of *Moraxella bovis* in the laboratory by the face fly (Diptera: Muscidae). J. Econ. Entomol. 75:816-818.

Arends, J. J., R. E. Wright, P. B. Barto & K. S. Lusby. 1984. Transmission of *Moraxella bovis* from blood agar cultures to hereford cattle by face flies (Diptera: Muscidae). J. Econ. Entomol. 77(2): 394-398.

Berkbile, D. R., R. D. Hall & J. J. Webber. 1981a. Field association of female face FLies with *Moraxella bovis*, an etiological agent of bovine pinkeye. J. Econ. Entomol. 74(4): 475-477.

Berkebile, D. R., R. D. Hall & J. J. Webber. 1981b. *Moraxella bovis* (Hauduroy) survival on female face flies: laboratory studies. Trans. Mo. Acad. Sci. 15: 83-88.

Broce, A. B. & V. G. Gonzaga. 1987. The effects of substituted benzylphenols and trislumuron on reproduction in the face fly (Diptera: Muscidae). J. Econ. Entomol. 80: 73-76.

Broce, A. B., M. J. Grodowitz & J. G. Riley. 1988. Effect of the Ionophore Lasalocid on face fly (Diptera: Muscidae) larval survival and physical and chemical parameters of cattle feces. J. Kansas Entomol. Soc. 61(4): 471-476.

Broce, A. B. & R. J. Elzinga. 1984. Comparison of prestomal teeth in the face fly (*Musca autumnalis*) and the house fly (*Musca domestica*) (Diptera: Muscidae). J. Med. Entomol. 21:82-85.

Brown, A.H. Jr.; Johnson, Z.B.; Simpson, R.B.; Brown, M.A.; Steelman, C.D.; Rosenkrans, C.F. Jr. 1994. Relationship of horn fly to face fly infestation in beef cattle. J. Anim. Sci.. 72 (9): 2264-2269.

Bryant, E. H., H. van Dijk & W. van Delden. 1981. Genetic variability of the face fly, *Musca autumnalis* De Geer, in relation to a population bottleneck. Evolution. 35: 872.

Burks, C.S.; Hagstrum, D.W.; Hampton, K.E.; Broce, A.B. 1997. Crystallization temperature and chilling injury during overwintering in a feral face fly (Diptera: Muscidae) population. Environ. Entomol. 26 (5): 1124-1130.

Burt, E.; Darlington, M.V.; Graf, G.; Meyer, H.J. 1992. Isolation, purification and characterization of an insect carbonic anhydrase. Insect Biochem Mol Biol. 22 (3): 285-291.

Cheville, N. F., D. G. Rogers, W. L. Deyoe, E. S. Krafsur & J. C. Cheville. 1989. Uptake and excretion of *Brucella abortus* in tissues of the face fly (*Musca autumnalis*). Am. J. Vet. Res. 50: 1302-1306.

Chirico, J. 1996. Transmission of *Heterotylenchus autumnalis* nematodes into field populations of *Musca autumnalis* by release of laboratory-reared specimens. Med-vet-entomol. Oxford. 10 (2): 187-189.

Cilek, J.E.; Knapp, F.W. 1994. Face fly (Diptera: Muscidae) gynandromorph. J Med Entomol. 31 (5): 760-762.

Coleman, R. E. & R. R. Gerhardt. 1987. Isolation of *Moraxella bovis* from the crops of field-collected face flies. J. Agric. Entomol. 4: 92-94.

D'amato, L. A., F. W. Knapp & D. L. Dahlman. 1980. Survival of the face fly in feces from cattle fed alfalfa hay or grain diets: Effect of fermentation and microbial changes. Environ. Entomol. 9: 557-560.

Darlington, M. V. & H. J. Meyer. 1988. Polytene chromosomes of the face fly, *Musca autumnalis* DeGeer. Bionature 8(1): 70-73.

Darlington, M. V., H. J. Meyer & G. Graf. 1984. The location, purification and partial characterization of carbonic anhydrase in the face fly, *Musca autumnalis*. Ann. N.Y. Acad. Sci. 429: 219-221.

Darlington, M. V., H. J. Meyer, G. Graf & T. P. Freeman. 1983. The calcified puparium of the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Insect Physiology 29: 157.

DeBower, D.; Thompson, J. R. 1997. Infectious bovine keratoconjunctivitis. Iowa State University Veterinarian. 59(1): 20-24.

Dougherty, C.T.; Knapp, F.W.; Burrus, P.B.; Willis, D.C.; Bradley, N.W. 1993. Face flies (*Musca autumnalis* De Geer) and the behavior of grazing beef cattle. Appl Anim Behav Sci. Amsterdam ; New York : Elsevier, 1984-. Feb 1993. v. 35 (4) p. 313-326.

Easton, E. R. & T. J. Lysyk. 1986. Response of a laboratory strain of the face fly to seasonal changes in manure quality. J. Agric. Entomol. 3: 179-185.

Easton, E. R. 1979. The value of the pyramid sticky trap for sampling face flies, *Musca autumnalis*, under field conditions in South Dakota. Environ. Entomol. 8: 1161-1164.

Evans, L. D. & E. S. Krafsur. 1990. Light intensity, photoperiod, and diapause induction in face flies (Diptera: Muscidae). Environ. Entomol. 19(5): 1217-1222.

Frishman, Austin M.; Matthysse, John George. 1966. Olfactory responses of the face fly Musca autumnalis De Geer and the housefly Musca domestica Linn. Memoir Cornell University. Agricultural Experiment Station, Ithaca, N.Y No. 394: 91p

Gerhardt, R. R., J. W. Allen, W. H. Greene & P. C. Smith. 1982. The role of face flies in an episode of infectious bovine keratoconjunctivitis. J. Am. Vet. Med. Assoc. 180:156-159.

Glass, H. W. & R. R. Gerhardt. 1994. Transmission of *Moraxella bovis* by regurgitation from the crop of the face fly (Diptera: Muscidae). J. Econ. Entomol. 77: 399-401.

Glass, H. W., Jr., R. R. Gerhardt & W. H. Greene. 1982. Survival of *Moraxella bovis* in the alimentry tract of the face fly. J. Econ. Entomol. 75: 545-546.

Grodowitz, M. J. & A. B. Broce. 1983. Calcium storage in face fly (Diptera: Muscidae) larvae for puparium formation. Ann. Entomol. Soc. Am. 76: 418-424.

Grodowitz, M. J., A. B. Broce & K. J. Kramer. 1987a. Morphology and biochemical composition of mineralized granules for the malpighian tubules of *Musca autumnalis* DeGeer larvae. Insect Biochem. 17: 335-345.

Grodowitz, M. J., A. B. Broce & L. H. Harbers. 1987b. Characteristics of dung that affect face fly, *Musca autumnalis* (Diptera: Muscidae), larvae survival and puparial mineralization. Environ. Entomol. 16(3): 722-730.

Grodowitz, M. J., C. R. Roseland, K. K. Hu, A. B. Broce & K. J. Kramer. 1987. Mechanical properties of mineralized and sclerotized puparial cuticles of the flies *Musca autumnalis* and *M. domestica*. J. Exp. Zool. 243: 201-210.

Hall, R. D. & M. C. Foehse. 1980. Laboratory and field tests of CGA-72662 for control of the house fly and face fly in poultry, bovine or swine manure. J. Econ. Entomol. 73(3): 564-569.

Hall, R. D. 1984. Relationship of the face fly (Diptera: Muscidae) to pinkeye in cattle: a review and synthesis of the relevant literature. J. Med. Entomol. 21: 361-365.

Herald F. & F. W. Knapp. 1980. Effects of monensin on the development of the face fly and the horn fly. J. Econ. Entomol. 73(6): 762-763.

Herald, F. & F. W. Knapp. 1981. Cattle, evaluation of fenvalerate and tetrachlorvinphos ear tags for horn fly and face fly control. Insecticide-Acaracide Tests. 5: 220-221.

Hogsette, J. A. & J. P. Ruff. 1986. Evaluation of flucythrinate- and fenvalerate impregnated ear tags and permethrin ear tapes for fly (Diptera: Muscidae) control on beef and dairy cattle in northwest Florida. J. Econ. Entomol. 79(1): 152-157.

Johnson, G. D. & J. B. Campbell. 1987. Evaluation of sticky traps for collection face flies (Diptera: Muscidae) from different locations in an irrigated pasture. Environ. Entomol. 16(1): 190-194.

Johnson, G. D., J. B. Campbell, H. C.Minocha & A. B. Broce. 1991. Ability of *Musca autumnalis* (Diptera: Muscidae) to acquire and transmit bovine Herpesvirus-1. J. Med Entomol. 28(6): 841-846.

Jurenka, R.A.; Holland, D.; Krafsur, E.S. 1998. Hydrocarbon profiles of diapausing and reproductive adult face flies (Musca autumnalis). Arch Insect Biochem Physiol. 37 (3): 206-214.

Kaya, H. K. & R. D. Moon. 1980. The influence of protein in the diet of face fly on the development of its nematode parasite, *Heterotylenchus autumnalis*. Ann. Entomol. Soc. Am. 73: 547-552.

Kennedy,-M.J. 1994. The effect of treating beef cattle on pasture with ivermectin on the prevalence and intensity of *Thelazia* spp. (Nematoda: Thelazioidea) in the vector, *Musca autumnalis* (Diptera: Muscidae). J-parasitol. 80 (2): 321-326.

Kim, Y.; Krafsur, E.S. 1994. Temperature and photoperiod affects diapause induction in immature stages of face flies (Diptera: Muscidae). Environ Entomol. 23 (3): 744-748.

Kim, Y.; Krafsur, E.S. 1995. In vivo and in vitro effects of 20-hydroxyecdysone and methoprene on diapause maintenance and reproductive development in *Musca autumnalis*. Physiol Entomol. 20 (1): 52-58.

Kim, Y.; Krafsur, E.S.; Bailey, T.B.; Zhao, S. 1995. Mode of inheritance of face fly diapause and its correlation with other developmental traits. Ecol Entomol. 20 (4): 359-366.

Kim, Y.G.; Krafsur, E.S. 1993. Adult emergence patterns in *Musca autumnalis* (Diptera: Muscidae). J-med-entomol. 30 (4): 816-819.

Knapp, F. W. & F. Herald. 1982. Congenitally induced mortality in face flies (Diptera: Muscidae) following adult exposure to diflubenzuron treated surfaces. J. Med. Entomol. 19(2): 191-194.

Knapp, F. W. & F. Herald. 1983. Mortality of eggs and larvae of the face fly (Diptera: Muscidae) after exposure of adults to surface treated with BAY SIR 8514 and Penfluron. J. Econ. Entomol. 76(6): 1350-1352.

Knapp, F. W. 1984. Effects of application date and selective tagging of cows and calves with fenvalerate ear tags for the control of the horn fly and face fly (Diptera: Muscidae). J. Agric. Entomol. 1(2): 126-136.

Knapp, F. W. & F. Herald. 1980. Efficacy of permethrin ear tags against face flies and horn flies on pastured cattle. Southwest. Entomol. 5: 183-186.

Knapp, F.W.; Webb, J.D. 1992. Use of dichlorvos in a portable automatic sprayer-mineral station to control face flies (Diptera: Muscidae) and pyrethroid-resistant horn flies (Diptera: Muscidae) on beef cattle. J Agric Entomol. 9 (4): 273-281.

Knapp, F. W., F. Herald & K. A. Schwinghammer. 1985. Comparative toxicity of selected insecticides to laboratory-reared and field-collected face flies (Diptera: Muscidae). J. Econ. Entomol. 78(4): 860-862.

Krafsur, E. S. 1984. Use of age structure to assess insecticidal treatments of face fly populations, *Musca autumnalis* DeGeer (Diptera: Muscidae). J. Econ. Entomol. 77: 1364-1366.

Krafsur, E.S. 1995. Analysis of gene flow in North American face fly (Musca autumnalis) populations. Med Vet Entomol. 9 (3): 229-234.

Krafsur, E. S. & W. C. Black IV. 1992. Analysis of isozyme loci in the face fly, *Musca autumnalis* DeGeer. Biochemical Genetics. 30(11&12): 625-634.

Krafsur,-E.S.; Moon,-R.D. 1997. Bionomics of the face fly, Musca autumnalis. Annu-rev-entomol. 42: 503-523.

Krafsur, E. S., C. J. Church, M. K. Elvin & C. M. Ernst. 1983. Epizootiology of *Heterotylenchus autumnalis* among face flies in central Iowa. J. Med. Entomol. 20(3): 318-324.

Krafsur, E. S., L. D. Evans & W. C. Black. 1986. Autumn phenology of face fly (Diptera: Muscidae) populations in Iowa. J. Med. Entomol. 23: 383-389.

Krafsur, E. S., R. D. Moon & C. J. Church. 1985a. Age structure and reproductive history of some overwintering face fly (Diptera:Muscidae) populations in North America. Ann. Entomol. Soc. Am. 78: 480-487.

Krafsur, E. S., Moon, R. D. & H. K. Kaya. 1985b. Age structure of breeding face fly populations (Diptera: Muscidae). Environ. Entomol. 15: 590.

Krafsur, E.S.; Moon, R.D.; Kim, Y. 1995. Age structure and reproductive composition of summer Musca autumnalis (Diptera: Muscidae) populations estimated by pterin concentrations. J Med Entomol.. 32 (5): 685-696.

Krueger, R. A., A. B. Broce & T. L. Hopkins. 1987. Dissolution of granules in the malpighian tubules of *Musca autumnalis* DeGeer, during mineralization of the puparium. J. Insect Physiol. 33: 255-263.

Lysyk, T. J. & E. R. Easton. 1985. Relationships between body size and fecundity in the face fly. Proc. S.D. Acad. Sci. 64: 171-176.

Mansour, Y. M. & E. S. Krafsur. 1991. Induction of dominant lethal mutations for control of the face fly, *Musca autumnalis* DeGeer. Med & Vet Ent. 5: 175-181

Mansour, M.Y.; Krafsur, E.S.; Mutchmor, J.A. 1994. Effects of anoxia during gamma irradiation on face fly fertility and viability (Diptera: Muscidae). J Med Entomol. 31 (1): 62-66.

McLemore, D. L., E. L. Rawls and D. E. Wells. 1993. Factors affecting price differences for feeder cattle sold through Tennessee auctions. Univ. of Tn. Ag. Exp. Stat., Res. Rpt. 93-18. De. 1993.

McLeod, M.; Zulesky, D. 1993. Control of horn flies and face flies. Ext-extra. Brookings, SD : Cooperative Extension Service, (8092) 5 p.

Meyer, H. J, & D. D. Kopp. 1984. The face fly *Musca autumnalis* DeGeer, in North Dakota. North Dakota Cooperative Extension Service EB36.

Meyer, J. A., C. M. Christensen & F. W. Knapp. 1978. The Influence of various levels of ground ear corn and alfalfa hay in the bovine diet on the development of the face fly. Environ. Entomol. 7(6): 829-830.

Miller, R. W. & J. A. Miller. 1994. Pyriproxyfen bolus for control of fly larvae. J. Agric. Entomol. 11(1): 39-44.

Miller, J. A., S. E. Kunz, D. D. Oehler & R. W. Miller. 1981. Larvicidal activity of Merck MK-933, an avermectin, against the horn fly, stable fly, face fly and house fly. J. Econ. Entomol. 74:608-611.

Miller, R. W., L. G. Pickens & D. M. Nafus. 1984a. Use of white tetrahedral traps and stirofos oral larvicide for area-wide control of the face fly. J. Agric. Entomol. 1(2): 126-136.

Miller, R. W., L. G. Pickens, E. T. Schmidtmann & W. E. Kunkle. 1984b. Fenvalerate ear tags with and without stirofos oral larvicide for control of horn flies and face flies. The Southwest. Entomol. 9(1): 79-83.

Miller, R. W., R. D. Hall, F. W. Knapp, R. E. Williams, K. E. Doisy, F. Herald & C. A. Towell. 1984c. Permethrin ear tags evaluated in four states for control of the horn fly and face fly. J. Agric. Entomol. 1(3): 264-268.

Miller, J. A., F. W. Knapp, R. W. Miller, C. W. Pitts & J. Weintraub. 1986. Diflubenzuron bolus for control of fly larvae. J. Agric. Entomol. 3(1): 48-55.

Miller, R. W., F. W. Knapp, R. D. Hall, R. E. Williams, K. E. Doisy & J. Webb. 1990. Field evaluation of diflubenzuron boluses for fly control on pastured cattle. J. Agric. Entomol. 7(4): 305-319.

Miller, R. W., R. D. Hall, R. E. Williams, L. G. Pickens & K. A. Doisy. 1991. Diflubenzuron boluses for fly control on dairy cattle. Agric. Entomol. 8(2): 117-126.

Minnifield, N.M.; Hayes, D.K. 1992. Partial purification of a receptor for the adipokinetic hormones from *Musca autumnalis* face flies. Prep Biochem. 22 (3/4): 215-228.

Moon, R. D. 1980. Effects of larval competition on face fly. Environ. Entomol. 9: 325 330.

Moon, R.D. 1983. Simulating developmental time of preadult face flies (Diptera: Muscidae) from air temperature records. Environ. Entomol. 12: 943-948.

Moon, R. D. 1986. Structure and validation of Flypop: a simulation model of face fly populations, pp. 18-27. In J, A. Miller (ed.), Modeling and simulation tools for management of veterinary pests, USDA, Agricultural Research Service, AARS-46, 54 pp.

Moon, R. D. & H. K. Kaya. 1981. A comparison of methods for assessing age structure and abundance in populations of nondiapausing female *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. 18: 289-297.

Moon, R. D., E. C. Loomis & J. R. Anderson. 1980. Influence of two species of dung beetles on larvae of face fly. Environ. Entomol. 9: 607-612.

Moon, R. D., K. Schwinghammer, F. W. Knapp, H. J. Meyer, E. T. Schmidtmann & R. D. Hall. 1986. Sampling bias and habitat variation in gonotrophic age of female *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. 23: 269-273.

Moon, R. D., R. A. Weinzierl, F. W. Knapp, R. W. Miller & R. D. Hall. 1991. Date of ear tag application and control of face fly, *Musca autumnalis* DeGeer, and horn fly, *Haematobia irritans* (L.), on pastured cattle. J. Agric. Entomol. 8(3): 199-208.

Peitzmeier,-B.A.; Campbell,-J.B.; Thomas,-G.D. 1992. Insect fauna of bovine dung in northeastern Nebraska and their possible effect on the face fly, Musca autumnalis (Diptera: Muscidae). J-Kans-Entomol-Soc. 65 (3): 267-274.

Pickens, L. G. 1981. Relationship between the capture of marked face flies and the number of sticky traps used. Southwest. Entomol. 6:326-330.

Pickens, L. G. 1990. Colorimetric versus behavioral studies of face fly (Diptera: Muscidae) vision. Environ. Entomol. 19(5): 1242-1252.

Pickens, L. G. & D. M. Nafus. 1982. Relationships between patterns of face fly capture and pasture topography, vegetation, and cattle movements. Environ. Entomol. 11(4): 843-846.

Pickens, L. G. & R. W. Miller. 1980. Biology and control of the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. 17: 195-210.

Raman, D.R.; Gerhardt, R.R. 1997. An insect-based detector of bovine manure odors. Trans ASAE. 40 (6): 1699-1707.

Rosales, A.L.; Krafsur, E.S.; Kim, Y. 1994. Cryobiology of the face fly and house fly (Diptera: Muscidae). J Med Entomol. 31 (5): 671-680.

Schmidtmann, E. T. 1985a. Face flies, *Musca autumnalis* DeGeer, and ear flap behavior in holstein cows. J. Agric. Entomol. 2: 69-72.

Schmidtmann, E. T. 1985b. The face fly, *Musca autumnalis* De Geer, and aggregation behavior in holstein cows. Vet. Para. 18:203-208.

Schmidtmann, E. T. & D. R. Berkebile. 1985. Relationship between numbers of face flies (Diptera: Muscidae) and nearest-neighbor distance among holstein cows. J. Econ. Entomol. 78(1): 102-105.

Schmidtmann, E. T. & L. G. Pickens. 1986. Phenology of diapause induction in a Maryland population of the face fly, *Musca autumnalis* De Geer. J. Agric. Entomol. 3(4): 297-303.

Schmidtmann, E. T. & R. E. Redfern. 1985. Physiological condition of face flies, *Musca autumnalis*, captured from cattle during the fall months. Canad. Entomol. 117: 225-226.

Schmidtmann, E. T., D. Berkebile, R. W. Miller & L.W. Douglass. 1984. The face fly (Diptera: Muscidae): Effect on holstein milk secretion. J. Econ. Entomol. 77(5): 1200-1205.

Schugart, J. I., J. B. Campbell, D. B. Hudson, C. M. Hibbs, R. G. .White & D. C. Clanton. 1979. Ability of the face fly to cause damage to the eyes of cattle. J. Econ. Entomol. 72: 633-635.

Scott, T. W. , R. W. Miller & F. W. Knapp. 1986. Field evaluation of diflubenzuron boluses with and without flucythrinate ear tags for control of horn flies, *Haematobia irritans*, and face flies, *Musca autumnalis*, on pastured cattle. J. Agric. Entomol. 3: 105-113.

Skoda, S. R., J. B. Campbell & G. D. Thomas. 1987a. Parasites of face flies (Diptera: Muscidae) in south central Nebraska. Environ. Entomol. 16(4): 902-904.

Skoda, S. R., J. B. Campbell & S. E. Kunz. 1987b. Wide-area treatment of cattle for horn flies and face flies (Diptera: Muscidae) in south-central Nebraska. J. Econ. Entomol. 80(4): 811-816.

Sowig, P. 1997. Predation among *Sphaeridium* larvae: the role of starvation and size differences (Coleoptera: Hydrophilidae). Ethology Ecology & Evolution. 9(3): 241-251.

Sowig, P.; Himmelsbach, R.; Himmelsbach, W. 1997. Predator-prey relationship between insect larvae: growth of *Sphaeridium* larvae (Coleoptera: Hydrophilidae) under time constraints through predation on *Musca autumnalis* maggots (Diptera: Muscidae). Canadian Journal of Zoology. 75(12): 2069-2076.

Sowig, P.; Himmelsbach, R.; Himmelsbach, W. 1997. Predator-prey relationship between insect larvae: growth of *Sphaeridium* larvae (Coleoptera: Hydrophilidae) under time constraints through predation on *Musca autumnalis* maggots (Diptera: Muscidae). Can J Zool. 75 (12): 2069-2076.

Steelman, C.D.; Gbur, E.E.; Tolley, G.; Brown, A.H. Jr. 1993. Variation in population density of the face fly, *Musca autumnalis* De Geer, among selected breeds of beef cattle. J-Agric-Entomol. 10 (3): . 97-106.

Van Geem, T. A. & A. B. Broce. 1985. Significance of cattle discharges and secretions as protein sources for ovarian development in the face fly (Diptera: Muscidae). Environ. Entomol. 14: 60-64.

VanGeem, T. A. & A. B. Broce. 1986. Fluctuations in the protein and carbohydrate content of the crop correlated to periodicities in ovarian development of the female face fly (Diptera: Muscidae). Ann. Entomol. Soc. Am. 79: 1-6.

VanGeem, T. A., A. B. Broce & R. D. Moon. 1983. A system for physiological age grading of female face flies *Musca autumnalis* DeGeer. Kans. Agric. Exp. Stn. Res. Bull. 643. 23 pp.

Williams, R. E. & E. J. Westby. 1980. Evaluation of pyrethroids impregnated in cattle ear tags for control of face flies and horn flies. J. Econ. Entomol. 73: 791-792.

Williams, R. E., E. J. Westby, K. S. Hendrix & R. P. Lemenager. 1981. Use of insecticide-impregnated ear tags for the control of face flies and horn flies on pastured cattle. J. An. Sci. 53: 1159-1165.

Yonggyun, Kim & E. S. Krafsur. 1993. Adult emergence patterns in *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. 30(4): 816-819.

Horn Fly

Description and Biology. The horn fly, *Haematobia irritans,* is an obligate blood-sucking parasite of cattle that was first reported in the U.S. in 1887. Adult female horn flies lay eggs in fresh cattle dung, usually within minutes after the pat is deposited on pasture. The eggs hatch in about 16 hours and ensuing larvae burrow into the pat where development continues through three instars. Pupation takes place either in the pat or in the soil immediately beneath or around the pat. The generation time for horn flies ranges from 8 to 30 days, depending on temperature (Kunz & Cunningham 1977). Adult females feed 24-38 times per day and leave the host only for short periods of time to oviposit on fresh cattle dung (Harris et al. 1974). In some areas, several thousand flies per animal may be found on cattle during the peak horn fly season. Horn flies are active 3-4 months of the year in the northern part of the country with some fly activity present during most of the year in the southernmost areas.

Economic Importance. Bruce and Decker (1958) suggested several ways in which biting flies might cause losses (decrease in milk production or decreases in body weight gain) in dairy cattle. These include annoyance from pain, blood losses, possible anaphylaxis from fly-derived substances left in the animal, interference with normal grazing habits, and increased energy utilized by the animal in its effort to ward off the flies. The use of insecticidal ear tags in Holstein cows for horn fly control resulted in an overall increase in milk yield by 1.06 kg/d (Block & Lewis 1986).

Methods of Control.

(1) Insecticides. Organophosphates, pyrethroids, and avermectins are available for horn fly control on dairy cattle. Various delivery systems include backrubbers, sprays, dusts, ear tags, pour-ons, spot-ons, feed additives, and mineral blocks. Although the avermectins are not recommended for lactating cattle, they can be used for horn fly control on non-lactating cows. However, the avermectins have been reported to be detrimental to beneficial insects such as dung beetles (Wardhaugh & Rodriguez-Menendez 1988, Fincher 1992, Ridsdill-Smith 1993) and prolong the degradation of cattle dung pats dropped on pasture (Wall & Strong 1987, Madsen et al. 1990).

(2) Insect Growth Regulators. Insect growth regulators (IGR's) are also used to control horn flies on dairy cattle. Methoprene and diflubenzuron have been used in bolus formulations for horn fly control and methoprene is registered for use in mineral mixes. Little damage to other arthropods within the manure pat has been demonstrated when animals were treated with methoprene (Pickens & Miller 1975, Fincher 1991). Cook and Gerhardt (1977) found a significant reduction of only one nontarget species in pats decomposed after eight weeks following diflubenzuron treatment. However, the emergence of two species of dung beetles was reduced for seven weeks when brood balls were made with dung from cattle containing a diflubenzuron bolus (Fincher 1991). Methoprene and diflubenzuron are of little use to individual producers because adult flies are not killed and migration from outside premises will be sufficient to maintain damaging adult populations (Kunz et al. 1972).

(3) Sanitation. Because the horn fly breeds only in fresh cattle dung dropped on pasture, sanitation will play no role in the control of this pest species.

(4) Biological Control. The insect fauna of cattle dung pats has been studied extensively during the past 15 years to determine the parasites, predators and competitors of immature stages of horn flies (Summerlin et al. 1982a,b, Figg et al. 1983, Roth et al. 1983, Harris & Summerlin 1984, Hunter et al. 1986, 1989, Schreiber & Campbell 1986, Blume 1986, 1987, Roth 1989, Cervenka & Moon 1991). A total of 457 species of insects have been reported to be associated with cattle dung on pasture in America north of Mexico (Blume 1985). In central Texas, 103 species of insects were associated with cattle dung on pasture (Blume 1970). Naturally deposited dung pats exposed to the existing dung-inhabiting fauna resulted in 88% mortality to immature horn flies (Roth 1989).

Most emphasis on the biological control of horn flies in recent years has been on the use of dung-burying beetles to reduce the breeding habitat of the pest. Fifteen exotic species of dung-burying scarabs have been released in various states and five species are known to be established, mostly in the southern tier of states (Fincher 1990). The effect of these dung-burying beetles on populations of horn flies has yet to be fully evaluated, but a decrease in horn fly populations has been noted on cattle in several states when dung beetle populations were sufficient to bury most cowpats within 24 hours after deposition. Scarab competition was reported to be a significant mortality factor of the horn fly in east-central Texas (Roth et al. 1983). A significant reduction in the number of horn flies emerging from individual dung pats due to the dung-burying activity of scarabs has also been reported (Roth 1989).

The family Staphylinidae contains the most important predators of dung-breeding flies because of their diversity and high populations in pastures. Both the adult and larval stages of staphylinids prey on immature stages of flies in dung deposits and several species have been documented as effective predators (Thomas & Morgan 1972, Roth et al. 1983, Thomas et al. 1983, Hunter et al. 1989). Two exotic staphylinid species, *Philonthus flavocinctus* from southeast Asia and *P. minutus* from Australia have recently been released in Texas, but it is unknown if they will become established. Additional species of staphylinids from Europe and South America will soon be released.

Beetles in the family Histeridae are also effective predators on dung-breeding flies, but the number of histerids occurring in cattle dung pats on pasture is usually low (Summerlin et al. 1982a,b). Three exotic species of histerids have been evaluated as horn fly predators in the laboratory, but only one species, *Atholus rothkirchi* from South Africa, has been released. A histerid commonly found in cattle dung in Argentina is currently being evaluated for potential release in Texas.

Twenty-two species of wasp parasitoids and one species of beetle parasitoid (Staphylinidae) have been reported from horn fly pupae (Fincher 1990). No parasitoids are known to be host specific for the horn fly.

(5) Mechanical Control. Fly traps are effective under certain conditions in reducing horn fly populations (Bruce 1940, Hall & Doisy 1989). The installation of such traps is recommended only for locations where the cattle will be forced to pass through it daily to drink water or enter the dairy barn.

(6) Genetic Control. None being done.

(7) Environmental Modification and Husbandry Practices. Environmental modifications will be of little or no help in the control of horn flies since they are a pest of pastured cattle.

(8) Host Resistance. No work will probably be done in this area with dairy cattle.

Current Support. Support for research on horn flies is directed toward beef cattle. Most results obtained are applicable to dairy cattle.

Summary of Research and Extension Needs

(1) Economic Injury Levels. Controlling horn flies on dairy cattle is a producer's decision. However, dairymen need to keep horn fly populations on dairy cattle as low as possible on each animal. There is no doubt that under certain conditions, horn flies may cause a significant reduction in milk production, but the exact amount is unknown.

(2) Sampling. Current sampling methods allow for the estimation of horn flies per head or on a per side basis. This method is acceptable as long as the counts are below 500 per head; populations above this number become more difficult to estimate. An experienced technician can, however, provide an accurate relative estimate. The estimates are sufficient as long as the same person is making the counts throughout the test. A total body count is more accurate than making a side count. Light conditions, the degree of animal irritation, etc., are too variable to make this a reliable estimate. At least 10 animals or 10% of a herd should be examined to establish an average fly count per head for the entire herd.

(3) Control Components. (a) Insecticides - direct animal applications by hand, dust bags, back rubbers, ear tags, injections or used as feed additives. (b) Insect growth regulators - methoprene as a feed additive, diflubenzuron as a bolus. (c) Biological - parasites, predators, competitors, pathogens.

(4) Biological and Ecological Information. Same as for beef cattle.

(5) Systems Models. Same as for beef cattle.

(6) Area Control vs. Individual Control. Individual control offers better control of the horn fly than for other pest species such as the face fly.

(7) Legal Barriers Involved. No legal barriers known.

(8) Eradication vs. Regulation. Same as for beef cattle.

References Cited

Block, E. & D. J. Lewis.1986. Efficacy of insecticidal ear tags on fly control and milk production of dairy cows. Can. J. Anim. Sci. 66: 47-51.

Blume, R. R. 1970. Insects associated with bovine droppings in Kerr and Bexar Counties, Texas. J. Econ. Entomol. 63: 1023-1024.

Blume, R. R. 1985. A checklist, distributional record, and annotated bibliography on the insects associated with bovine droppings on pastures in America north of Mexico. Southwest. Entomol. (Suppl. No. 9) 55 p.

Blume, R. R. 1986. Parasites of Diptera associated with bovine droppings on a pasture in east central Texas. Southwest. Entomol. 11: 215-222.

Blume, R. R. 1987. Bionomics of some species of the genus *Spalangia* Latreille. Southwest. Entomol. 12: 191-195.

Bruce, W. G. 1940. A cattle fly trap for the control of horn flies. U.S. Dept. Agri. Circ. E 498.

Bruce, W. N. & G. C. Decker. 1958. The relationship of stable fly abundance to milk production in dairy cattle. J. Econ. Entomol. 51: 269-274.

Cervenka, V. J. & R. D. Moon. 1991. Arthropods associated with fresh cattle dung pats in Minnesota. J. Kans. Entomol. Soc. 64: 131-145.

Cook, C. W. & R. R. Gerhardt. 1977. Selected mortality of insects in manure from cattle fed rabon and dimilin. Environ. Entomol. 6: 589-590.

Figg, D. E., R. D. Hall, & G. D. Thomas. 1983. Insect parasites associated with Diptera developing in bovine dung pats on central Missouri pastures. Environ. Entomol. 12: 961-966.

Fincher, G. T. 1990. Biological control of dung-breeding flies: Pests of pastured cattle in the United States, pp. 137-151. *In* D. A. Rutz & R. S. Patterson (eds.), Biocontrol of arthropods affecting livestock and poultry. Westview Press, Boulder, CO. 316 pp.

Fincher, G. T. 1991. Sustained-release bolus for horn fly (Diptera: Muscidae) control: Effects of methoprene and diflubenzuron on some nontarget species. Environ. Entomol. 20: 77-82.

Fincher, G. T. 1992. Injectible ivermectin for cattle: Effects on some dung-inhabiting insects. Environ. Entomol. 21: 871-876.

Hall, R. D. & K. E. Doisy. 1989. Walk-through trap for control of horn flies (Diptera: Muscidae) on pastured cattle. J. Econ. Entomol. 82: 530-534.

Harris, R. L. & J. W. Summerlin. 1984. Parasites of horn fly pupae in east central Texas. Southwest. Entomol. 9: 169-173.

Harris, R. L., J. A. Miller & E. D. Frazar. 1974. Horn flies and stable flies: feeding activity. Ann. Entomol. Soc. Am. 67: 891-894.

Hunter, J. S., D. E. Bay & G. T. Fincher. 1986. A survey of Staphylinidae associated with cattle droppings in Burleson County, Texas. Southwest. Entomol. 11: 83-89.

Hunter, J. S., D. E. Bay & G. T. Fincher. 1989. Laboratory and field observations on the life history and habits of *Philonthus cruentatus* and *P. flavolimbatus* (Coleoptera: Staphylinidae). Southwest. Entomol. 14: 41-47.

Kunz, S. E. & J. R. Cunningham. 1977. A population prediction equation with notes on the biology of horn fly in Texas. Southwest. Entomol. 2: 79-87.

Kunz, S. E., B. F. Hogan, R. R. Blume & J. L. Eschle. 1972. Some bionomical aspects of horn fly populations in central Texas. Environ. Entomol. 1: 565-568.

Madsen, M., B. Overgaard Nielsen, P. Holter, O. C. Pedersen, J. B. Jespersen, K. M. Vagn Jensen, P. Nansen & J. Gronvold. 1990. Treating dung with ivermectin: Effects on the fauna and decomposition of dung pats. J. Appl. Ecol. 17: 1-15.

Pickens, L. G. & R. W. Miller. 1975. Growth regulating chemicals tested against nontarget insect fauna in bovine fecal pats. Environ. Entomol. 4: 46-48.

Ridsdill-Smith, T. J. 1993. Effects of avermectin residues in cattle dung on dung beetle (Coleoptera: Scarabaeidae) reproduction and survival. Vet. Parasitol. 48: 127-137.

Roth, J. P. 1989. Field mortality of the horn fly on unimproved central Texas pasture. Environ. Entomol. 18: 98-102.

Roth, J. P., G. T. Fincher &. J. W. Summerlin. 1983. Competition and predation as mortality factors of the horn fly, *Haematobia irritans* (L.) (Diptera: Muscidae), in a central Texas pasture habitat. Environ. Entomol. 12: 106-109.

Schreiber, E. T. & J. B. Campbell. 1986. Parasites of the horn fly in western Nebraska. Southwest. Entomol. 11: 211-215.

Summerlin, J. W., D. E. Bay & R. L. Harris. 1982a. Seasonal distribution and abundance of *Histeridae* collected from cattle droppings in south central Texas. Southwest. Entomol. 7: 82-86.

Summerlin, J. W., D. E. Bay, R. L. Harris, D. J. Russell & K. C. Stafford. 1982b. Predation by four species of Histeridae (Coleoptera) on horn fly (Diptera: Muscidae). Ann. Entomol. Soc. Am. 75: 675-677.

Thomas, G. D. & C. E. Morgan. 1972. Field mortality studies on the immature stages of the horn fly in Missouri. Environ. Entomol. 1: 453-459.

Thomas, G. D., R. D. Hall, C. W. Wingo, D. B. Smith & C. E. Morgan. 1983. Field mortality studies on the immature stages of the face fly (Diptera. Muscidae) in Missouri. Environ. Entomol. 12: 823-830.

Wall, R. & L. Strong. 1987. Environmental consequences of treating cattle with the antiparasitic drug ivermectin. Nature. 327: 418-421.

Wardhaugh, K. G. & H. Rodriguez-Mendendez. 1988. The effects of the antiparasitic drug, ivermectin, on the development and survival of the dung breeding fly, *Orthelia cornicina* (F.) and the scarabaeine dung beetles, *Copris hispanus* L., *Bubas bubalus* (Oliver) and *Onitis belial* F. J. Appl. Entomol. 106: 381-389.

Horse Flies And Deer Flies

Description and Biology. The tabanids that usually attack dairy cattle are called horse flies and range from 27 to 10 mm in length. Deer flies (*Chrysops* spp.) are smaller (9 to 6 mm) and frequently attack humans in addition to pastured cattle. All tabanids have a similar life cycle that starts with gravid females depositing eggs on objects over a larval habitat that is suitable for that particular species. Eggs are laid in single or multi-tiered masses of ca. 100-1000 eggs/mass. The eggs hatch in 5-7 days and the larvae move into the habitat below. Larval habitats are usually aquatic or semi-aquatic and are generally species specific. However, they range from fast moving streams to moist sod that can be considered semi-aquatic only in the loosest sense.

Larvae are predatory and cannibalistic and may reach maturity in 2 mos. to 2 years depending on species, with 1 year being the most common. Prepupae usually move to a dryer situation to pupate and adults emerge 1 to 3 wks. later.

Dairy cattle may be attacked by one species for 1 mo. to 6 weeks, but are subjected to attack by several species in succession. Attacks occur in all the warm months, but are most numerous in the spring and early summer. There are about 300 species in North

America, but in any geographical area 3-6 species are responsible for the majority of attacks on cattle.

Adult tabanids are pool-feeders that cause the loss of blood from both feeding and subsequent bleeding from the wound. Free bleeding wounds attract facultative blood feeders (*Hippellates*, spp. and *Muscaautumnalis*). Horse and deer flies are also vectors of several important livestock diseases.

Economic Importance. Pastured cattle may suffer severely from heavy attacks of tabanids, and nervous activity may supplant normal grazing. Tashiro and Schwardt (1949) estimated that 40 *T. sulcifrons* would cause a cow to lose 115 cc of blood in 1 hour and Hollander and Wright (1980) estimated a total blood loss of 200 cc/day in Oklahoma. Dense fly populations are thought to contribute to slower weight gain and lower milk production in cattle (Bruce & Decker 1951; Granett & Hansens 1957), and blood oozing from the wound inflicted by tabanids may be fed upon by other flies (Garcia & Radovsky 1962) thus increasing the risk of infection. Perich et al. (1986) demonstrated that beef cattle exposed to an average of 66-90 horse flies per animal per day gained 0.08-0.10 kg per day less than protected cattle, and feed efficiency decreased 16.9 percent.

Methods of Control. No satisfactory control methods are currently available. However, application of high pressure (100-200 psi) pyrethroid sprays can impact tabanid feeding success. The feeding time was significantly lower on treated cows for the three predominant horse fly species. The amount of blood consumed by *T. fascicostatus* was significantly reduced by 29.3% for flies feeding on treated cows. Reducing the feeding time of a population of horse flies feeding on livestock by 35% would result in a 35% reduction of flies on the animal at any time. A 44% reduction in daily blood loss to tabanid feeding for cattle treated with fenvalerate would be predicted from the combination of reduced subsequent feeding and reduced blood meal size (Foil et al. 1990; Leprince et al. 1991).

Current Support. There is no tabanid research being conducted by USDA. SAES support is approximately 0.5 SY.

Summary of Research and Extension Needs

(1) Economic Injury Threshold. Few data are available relative to the effects of horse fly and deer fly attack on dairy cattle. Research is needed to show whether milk production decreases due to tabanid attacks and whether compensatory gains in cattle body weight occur after periods of attack have ended. Studies are also needed to determine the effects of tabanid attack on animals raised on high and low energy rations.

(2) Control. Two types of control methodologies need to be developed. These are (1) area-wide controls, and (2) individual animal protection techniques.

(3) Sampling. Sampling techniques for adults have been developed. However, the efficiency of these methods needs to be examined. Egg and larval sampling techniques need development. Population estimates of larval and adult sampling strategies should be compared. Basic biological and ecological data on larval habitat, adult flight and feeding behavior are needed for the majority of species which may be pests of dairy cattle.

(4) Disease Organism Transmission. Horse flies can transmit several disease organisms of dairy cattle (Krinsky 1976). *Anaplasma marginale*, bovine leukemia virus and vesicular stomatitis virus are disease organisms of dairy cattle that can be transmitted by tabanids.

(5) Research Classification
(a) Basic Research.
(1) For most of the common species attacking dairy cattle their basic biology and ecology needs to be studied. Seasonal activity, daily feeding cycles, larval habitats, flight activity and responses to visual and chemical cues, etc. need to be known in order to develop control strategies.
(2) Laboratory rearing techniques, if only for 1 or 2 selected species, are needed to allow for basic physiology studies and the development of more efficient insecticide screens for chemical control development.
(3) The role of tabanids in transmission of different diseases needs to be elucidated to help determine the economic impact of these pests.

(b) Control Components Research.
(1) Development of control technology including insecticide and application techniques needs to be developed. The pyrethroids have been shown to reduce feeding time and blood meal size of tabanids, but there are no accepted effective controls for tabanids. Techniques to evaluate potential repellents of tabanids as well as other hematophagous Diptera should be developed.
(2) The entire area of biological, ecological and genetic control needs to be investigated. These areas offer the most promise for the control of these pests.
(3) The economic impact of tabanids from both the annoyance and disease transmission needs to be determined.

(c) IPM Systems Research. The use of insecticide - impregnated targets in combination with semiochemicals is currently used in tsetse control. There may be odors used by multiple dipteran pests of range cattle to identify hosts. Techniques to screen for visual and olfactory ques used by dipteran pests of cattle should be developed. Tabanids will be an important group to use in these studies due to the similarity of host-seeking behavior of tsetse and certain tabanids.

Basic biological and ecological data on larval habitat, adult flight and feeding behavior are needed for the majority of species which may be pests of dairy cattle.

References Cited

Bruce, W. N. & G. C. Decker. 1951. Tabanid control on dairy and beef cattle with synergized pyrethrins. J. Econ. Entomol. 44: 154-9.

Foil, L. D., D. J. Leprince & R. L. Byford. 1990. Sublethal effects and mortality of tabanids (Diptera) induced by fenvalerate treatment of cattle. J. Entomol. Sci. 25: 294-302.

Garcia, R. & F. J. Radovsky. 1962. Haematophagy by two non-biting muscid flies and its relationship to tabanid feeding. Can. Entomol. 94: 1110-16.

Granett, P. & E. J. Hansens. 1957. Further observations on the effect of biting fly control on milk production in cattle. J. Econ. Entomol. 50: 332-6.

Hollander, A. L. & R. E. Wright. 1980. Impact of tabanids on cattle: blood meal size and preferred feeding sites. J. Econ. Entomol. 73: 431-3.

Krinsky, W. L. 1976. Animal disease agents transmitted by horse flies and deer flies. J. Med. Entomol. 13: 225-75.

Leprince, D. J., L. D. Foil & R. L. Byford. 1991. Evaluation of pyrethroid ear tag and spray treatment of cattle against horse flies (Diptera: Tabanidae). J. Entomol. Sci. 26(2): 271-280.

Perich, M. J., R. E. Wright & K. S. Lusby. 1986 . Impact of horse flies (Diptera: Tabanidae) on beef cattle. J. Econ. Entomol. 79: 128-31.

Tashiro, H. & H. H. Schwardt. 1949. Biology of the major species of horse flies of central New York. J. Econ. Entomol. 42: 269-72.

Biting Midges

Description and Biology. *Culicoides* and *Leptoconops* are small (0.6 to 5.0 mm) blood sucking flies known variously as biting midges, punkies, no-see-ums, or biting gnats. The wings at rest are positioned flat over the abdomen; in *Culicoides* they are patterned in species-specific light and dark patterns. Female *Leptoconops* have white, milky wings and a shiny black notum.

Immature *Culicoides* are found in semi-aquatic to aquatic habitats consisting of water-saturated mud, sand and detritus (Jamnback 1965). Larval populations of the *C. variipennis* complex commonly attain high densities in substrates contaminated by animal manure or high-level dissolved salts (Wirth & Jones 1957, Schmidtmann et al. in press). Dairy farms in particular provide numerous habitats for the immature stages of this economically important insect. Cattle with access to ponds and streams trample the margins, mix mud with manure and open shorelines to sunlight, creating favorable micro-habitats for the immature stages (Schmidtmann et al. 1983, Mullens & Rodriquez 1988). Dairy waste water lagoons also support high larval densities in southern and western regions of the U.S. (O'Rourke et al. 1983, Mullens & Lii 1987, Schmidtmann et al. 1998). *Leptoconops* larvae develop in sandy-saline or alkali-clay substrates (Whitsel & Schoeppner 1966; Wirth and Atchley 1973). Females of both *Culicoides* and *Leptoconops* seek blood meals to develop their eggs; males do not take blood. *Leptoconops* seek blood hosts during daylight and are univoltine. *Culicoides* generally seek hosts during crepuscular and nocturnal periods. Many species produce a single generation annually, but the *C.variipennis* complex produces repeated generations during the warmer months.

Economic Importance. *Culicoides* spp. are important vectors of arboviruses worldwide, where they serve as biological vectors for ca. 25 viruses, including exotic strains of bluetongue, akabane, Ibaraki, bovine ephemeral fever and epizootic hemorrhagic disease viruses. In the U.S., the biting midge, *C. sonorensis*, is the primary vector of bluetongue viruses. A bluetongue disease outbreak in Kentucky resulted in $35,000 in losses to 194 herds, and an estimated $6 million statewide (Metcalf et al. 1980). Export restrictions that prevent marketing of U. S. dairy cattle and their germplasm to bluetongue-free countries are estimated to cost producers 125 million in lost revenue annually (see Holbrook, 1988). Other species, such as *C. insignis, C. lahillei,* and *C. stellifer*, also may transmit bluetongue viruses in the southeastern region (Mullen et al. 1985, Tanya et al. 1992, Weiser-Schimpf et al. 1993). The *C. variipennis* complex consists of a complex of three species that are widespread and abundant throughout the U.S. (Holbrook et al. in press). The species, *C. sonorensis,* is primarily responsible for transmitting bluetongue viruses in the U. S. The transmission of bluetongue viruses occurs largely in western, central and southern regions where *C. sonorensis* is common. Major dairy economies in North-central and Northeastern regions are free of bluetongue (Metcalf et al. 1981, Pearson et al. 1992), despite the widespread presence of the closely related species, *C. variipennis* (Schmidtmann et al. 1983). This species is an inefficient vector of Bluetongue viruses (Tabachnick and Holbrook 1992). Because of the absence of bluetongue, the Northeastern region is being considered for special status as a free trade zone for the international movement of dairy cattle and germplasm (Walton et al. 1992). Species of the *C. variipennis* complex also are incriminated as vectors of epizootic hemorrhagic disease virus, an orbivirus closely related to bluetongue viruses, that causes epizootics with mortality in white-tailed deer (Gibbs & Greiner 1983). *Culicoides* have also been associated vesicular stomatitis virus during an outbreak in Colorado (Kramer et al. 1990). The role of *Culicoides* as vectors on non viral animal pathogens was reviewed by Linley (1985).

In Denmark, blood feeding *Culicoides* induce a dermatitis condition of the udder and teats of dairy cattle known as "summer mastitis" (Nielsen 1971). This condition is not recognized in the U.S., but blood feeding from dairy cattle and other large mammals by *Culicoides* is widespread and common (Jones & Akey 1977; Schmidtmann et al. 1980, Mullen & Hribar 1988, Anderson & Holloway 1993).

Current Control Methods. Females seeking blood from animals may be killed by contact or repulsed by synthetic pyrethroid compounds delivered as impregnated ear tags (Holbrook 1986), sprays or pour-on treatments that are applied directly to animals. Data concerning the effects of pyrethroid treatments are based on "in vitro" assays with *C. variipennis* (*C. sonorensis*) (Mullens (1993), as well as treatment of cattle. Shemanchuk and Taylor (1984) and Shemanchuk et al.(1991) reported that treatment of cattle with synthetic pyrethroid compounds effectively suppressed blood feeding in black flies and mosquitoes, and a similar effect may occur for *Culicoides*. In a laboratory assay that compared various products to DEET for repellency against *C. imicola* (Braverman 1997), a permethrin product was effective for only 1 hour, but a type II pyrethroid provided repellency for 9 hours. Cattle treated systemically with Ivermectin were lethal to *C. brevitarsis* in Australia (Standfast et al. 1984), but treatment did not prevent blood feeding, and levels of Ivermectin in serum of cattle treated at the recommended 200 mg/kg body weight in the U. S. would have little if any effect on *C. variipennis* (Holbrook & Mullens 1994). The various materials and methods used as adulticides and larvicides for controlling mosquitoes, including insect growth regulators and microbial products, can likely be adapted for suppressiong biting midge populations. Use of *Bacillus thuringiensis israelensis* (H 14) for control of *Culicoides* and *Leptoconops* was evaluated in laboratory studies by Lacey & Kline (1983). The parasitic mermithid nematode, *Heleidomermis magnapapula*, infects wild populations of *C. variipennis* (Mullens and Rutz 1982, Paine and Mullens 1994), can be reared in the laboratory, but infection rates appear to be low (Mullens & Velten 1994). Temephos, an organophophate insecticide, has been demonstrated to be an effective larvicide for area-wide suppression of immature *C. variipennnis* (*sonorensis*) (Holbrook et al. 1993).

Cultural control of immature *C. sonorensis* in dairy waste water lagoons has been demonstrated by manipulation of water and manure loading levels ((Mullens & Rodriguez 1988, Mullens & Rodriguez 1990). Wirth et al. (1977) and Undeen (1984) reviewed viral, bacterial, protozoan and nematode parasites of *Culicoides*, but these organisms have not been developed for control purposes. Confinement of dairy cattle indoors and/or cultivation of extensive acreage surrounding dairy drylots may limit exposure of cattle to *Culicoides* blood feeding, but such practices are not recognized or employed.

Current Support. Research concerning *Culicoides* vectors of Bluetongue in sheep, beef and dairy cattle is conducted by 3 SY, ARS, Laramie, WY., and SAES programs exist in CA, LA, AL and FL.

Summary of Research and Extension Needs

(1) Economic Injury Level. Information concerning any direct effects of *Culicoides* blood feeding from dairy cattle has yet to be developed and probably does not warrant attention. Like other hematophagous arthropods, however, the saliva of *C. sonorensis* contains anti-hemostatic and anti-inflammatory properties that may have systemic immune-compromising effects and facilitate transmission of disease agents (Perez de Leon et al.1997). Key factors involved in the vectorial capacity of *C. sonorensis* populations for transmission of bluetongue viruses, such as population density thresholds, cattle biting rates and infection rates, and female survivorship, need to be established. This information is needed to establish a "vector management level" for use in reducing exposure to bluetongue disease.

(2) Sampling. Existing sampling methods are adequate for qualitative purposes; refinement is required for manipulating or predicting population responses in the context of IPM. Immature *Culicoides* populations are sampled by substrate sieving and flotation procedures (Jamnback 1965, Hribar 1989). Host-seeking females are effectively captured using C02-baited light traps (Nelson 1965), canopy traps (Jones 1961; Humphreys & Turner 1973; Muller & Murray 1977), vacuum devices (Schmidtmann et al. 1980). These methods need to be further standardized with respect to pest or vector species population dynamics and attack rates (pest intensity) on dairy cattle, like the study of Mullens and Gerry (1998) that compared light traps, suction traps and catches from Holstein calves. Vehicle-mounted intercept traps (Barnard & Jones 1980) and D-Vac sampling (Tanner & Turner 1975) represent non animal-biased methods for population sampling. Information concerning various types of light traps and attractants (CO2) and their utility for sampling various age classes of adult *C. sonorensis* has been developed (Holbrook & Bobian 1989).

(3) Control Components. Control methods research should focus upon *C. sonorensis*. In particular, methods are needed to suppress or maintain population densities below levels where late-season virus transmission occurs. Strains or formulations of *Bacillus thuringiensis israelensis* may have potential as an environmentally acceptable larvicidal control technology. Alternatively, methods for decreasing the survivorship, hence vector capability, of adult biting midges that feed from cattle are needed for use during epizootic outbreaks. The synthetic pyrethroid formulations currently registered from use on dairy cattle need to be evaluated in the context of not only killing females, thus interrupting transmission, but also with respect to repellency as a means of protecting genetically valuable animal stocks, such as stud bulls and pure-bred heifers, from blood-feedingfemales.

(4) Biological-Ecological. Emphasis should focus on further defining the geographic distribution of the *C. variipennis* complex, with especial attention to larval habitats in transition zones; GIS habitat-attribute mapping would be useful in defining the distribution of *C. sonorensis* in these areas. Basic ecological data concerning population dynamics, adult phenology and vector competence are needed for species other than the *C. variipennis* complex that consistently blood feed from dairy cattle, such as *C. stellifer, C. obsoletus, C. venustus* and *C. lahillei*. The key factors that lead to transmission bluetongue viruses, including an explanation for late summer and fall periods of virus activity, need clarification and incorporation into epizootiologic models.

(5) Pest-Host Models. Predictive models of vector population dynamics or the components of vector-virus transmission cycles can be developed with existing data, but such models have not been constructed and will need refinement and validation. The role of host animals and periods of viremia in various domestic and wildlife species needs modeling to clarify enzootic virus maintenance and epizootic virus spread conditions for bluetongue and other orbiviruses.

(6) Area vs. Individual Producer Control. Area control may be necessary for *C. sonorensis* populations in arid western regions where breeding substrates are isolated from sources of cattle. The reduction of larval breeding sites and use of larvicidal treatment control methods, along with adult monitoring, have been assimilated into ongoing mosquito abatement programs at a cost-effective level (Holbrook et al. 1994). In areas where development sites for *C. sonorensis* are restricted to immediate dairy premises, producer-level control may be advisable and practical. Components of integrated control programs for *C. sonorensis* at the individual farm level have been outlined by Holbrook (1984) and Mullens (1992). Cost-effective methodology and on-farm demonstration are needed for acceptance by producers.

(7) Research Classification.

(a) Basic Research.

(1) For the *C. variipennis* complex, further characterize genetic basis for vector competence for North American serotypes of bluetongue viruses, including development of gene library for use in genetic control of bluetongue vectors.
(2) Investigate relationship between species of the *C. variipennis* complex and transmission of EHD serotypes and exotic bluetongue viruses.
(3) Derive through intensive epizootiologic studies in endemic areas information about the transmission dynamics of bluetongue virus activity.
(4) Further define and refine understanding of the distribution of non-bluetongue vector, *C. variipennis,* populations.
(5) Refine and improve ecological data for seasonal phenology, breeding sites and sampling methods for *Culicoides* species of potential economic importance other than the *C. variipennis* complex.
(6) Develop colonization and rearing procedures for key species other than the *C. variipennis* complex.

(b) Control Components Research.

(1) For *C. sonorensis*: determine feasibility for genetic replacement of virus susceptible populations with non-susceptible lineages.
(2) Explore use of biological control agents, *Bacillus thuringiensis* in particular, for suppression of larval population density.
(3) Evaluate pyrethroid compounds or novel repellents for use in preventing flies from feeding from cattle, thus interrupting any virus transmission..

(c) IPM Systems Research.

(1) Pursue acceptability of mosquito abatement technology, staff and equipment for suppressing immature *C. variipennis sonorensis* populations in dairy regions where cows risk exposure to bluetongue virus.
(2) Test and develop methods for using whole-body repellent or pyrethroid treatments to protect dairy cattle from *C. v. sonorensis* blood feeding and viral disease agents.

(d) Extension. Develop through sentinel dairy herd field studies the negligible risk for exposure to bluetongue viruses in upper Midwest and Northeast regions.

References Cited

Anderson, R. R. & H. L. Holloway, Jr. 1993. *Culicoides* (Diptera: Ceratopogonidae) associated with White-tailed deer and livestock operations in the northern Great Plains. J. Med. Entomol. 30: 625-627.

Barnard, D. R. & R. H. Jones. 1980. Diel and seasonal patterns of flight activity of Ceratopogonidae in Northeastern Colorado: *Culicoides*. Environ. Entomol. 9:446-451.

Braverman, Y. & A. Chizov-Ginsburg. 1997. Repellency of synthetic and plant-derived prepartations for *Culicoides imicola*. Med. Vet. Entomol. 11: 355-360.

Gibbs, E.P.J. & E. C. Greiner. 1983. Bluetongue infections and *Culicoides* species associated with livestock in Florida and the Caribbean region. Pp. 375-382. *In* R.W. Compans & D.H.L. Bishop [eds.], Double-Stranded RNA Viruses. Elsevier, Amsterdam.

Holbrook, F. R. 1984. Research on the control of Bluetongue in livestock by vector suppression. *pp 617-620. In* Bluetongue and Related Orbiviruses. T. L. Barber & M. M. Jochim, [eds.], Progress in Clinical and Biological Research, Vol. 178. Alan R. Liss, Inc, New York.

Holbrook, F. R. 1986. Exposure of *Culicoides variipennis* (Diptera: Ceratopogonidae) to hair clippings to evaluate insecticide-impregnated ear tags in cattle. J. Econ. Entomol. 79:1127-1129.

Holbrook, F. R. 1988. Bluetongue in the U.S.: Status, transmission and control through vector suppression. Bull. Soc. Vect. Ecol. 13: 350-353.

Holbrook, F. R. & R. J. Bobian. 1989. Comparisons of light traps for monitoring adult *Culicoides variipennis* (Diptera: Ceratopogonidae). J. Amer. Mosq. Cont. Assoc. 5:558-562.

Holbrook, F. R. & B. A. Mullens. 1994. Effects of ivermectin on survival, fecundity and egg fertility in *Culicoides variipennis* (Diptera: Ceratopogonidae). J. Amer. Mosq. Cont. Assoc. 10:70-73.

Holbrook, F. R., S. V. Romney & R. M. Sessions. 1993. Piggybacking bluetongue vector control and mosquito abatement. Amer. Mosq. Cont. Assoc. (Wing Beats) 4:19-23.

Holbrook, F. R., W. J. Tabachnick, E. T. Schmidtmann, C. N. McKinnon, R. J. Bobian and W. L. Grogan. Sympatry in the *Culicoides variipennis* complex (Diptera: Ceratopogonidae): a taxonomic reassessment. J. Med. Entomol. (In press)

Hribar, L. J. 1989. A review of methods for recovering biting midge larvae (Diptera: Ceratopogonidae) from substrate samples. J. Agric. Entomol. 7:71-77.

Humphreys, J. G. & E. C. Turner, Sr. 1973. Blood feeding activity of female *Culicoides* (Diptera: Ceratopogonidae). J. Med. Entomol. 10:79-83.

Jamnback, H. 1965. The *Culicoides* of New York State (Diptera: Ceratopogonidae). N.Y. State Mus. Sci. Serv. Bull. 399, 154p.

Jones. R. H. 1961. Some observations on biting flies attacking sheep. Mosq. News 21:113.

Jones, R. H. & D. H. Akey. 1977. Biting flies attacking Holstein cattle in a bluetongue enzootic area in Colorado, 1976. Mosq. News, 37:372-375.

Kramer, W. L., R. H. Jones, F. R. Holbrook, T. E. Walton & C. H. Calisher. 1990. Isolation of arboviruses from *Culicoides* midges (Diptera: Ceratopogonidae) in Colorado during an epizootic of vesicular stomatitis New Jersey. J. Med. Entomol. 27:487-493.

Lacey, L. A. & D. L. Kline. 1983. Laboratory bioassay of *Bacillus thuringiensis* (H-14) against *Culicoides* spp. and *Leptoconops* spp. (Ceratopogonidae). Mosq. News 43:502-503.

Linley, J. R. 1985. Biting midges (Diptera: Ceratopogonidae) as vectors of nonviral animal pathogens. J. Med. Entomol. 22:589-599.

Metcalf, H. E., J. Lomm & V. C. Beal, Jr. 1980. Estimate of incidence and direct economic losses due to bluetongue in Mississippi cattle during 1979. Proc. Ann. Mtng. U.S. Anim. Hlth. Assoc. 84:186-202.

Metcalf, H. E., J. E. Pearson & A. L. Klingsporn. 1981. Bluetongue in cattle: a serologic survey of slaughter cattle in the United States. Amer. J. Vet. Res. 42:1057-1061.

Mullen, G. R., R. H. Jones, Y. Braverman & K. E. Nusbaum. 1985. Laboratory infections of *Culicoides debilipalpis* and *C. stellifer* (Diptera: Ceratopogonidae) with bluetongue virus. *pp. 239-243. In* Bluetongue and Related Orbiviruses. T. L. Barber & M. M. Jochim, [eds.] , Progress in Clinical and Biological Research, Vol. 178. Alan R. Liss, Inc. New York.

Mullen, G. R. & L. J. Hribar. 1988. Biology and feeding behavior of ceratopogonid larvae (Diptera: Ceratopogonidae) in North America. Bull. Soc. Vector Ecol. 11:60-81.

Mullens, B. A. 1992. Intergrated managment of *Culicoides variipennis*: a problem of applied ecology. *pp, 896-905. In* Bluetongue, African Horse Sickness, and Related Orbiviruses, T. E. Walton & B. I. Osburn, [eds.], Proc. 2nd Internat. Sym., CRC Press, Inc., Boca Raton, FL

Mullens, B. A. 1993. In vitro assay for permethrin persistence and interference with bloodfeeding of *Culicoides* (Diptera: Ceratopogonidae) on animals. J. Amer. Mosq. Contr. Assoc. 9:256-259.

Mullens, B. A. and A. Gerry. 1998. Comparison of bait cattle and carbon dioxide suction traps for collecting *Culicoides variipennissonorensis* (Diptera: Ceratopogonidae) and *Culex quinquefasciatus* (Diptera: Culicidae). J. Med. Entomol. 35: 245-250.

Mullens, B. A. & K. Lii. 1987. Larval population dynamics of *Culicoides variipennis* (Diptera: Ceratopogonidae) in Southern California. J. Med. Entomol. 24:566-574.

Mullens, B. A. & J. L. Rodriguez. 1988. Colonization and response of *Culicoides variipennis* (Diptera: Ceratopogonidae) to pollution levels in experimental dairy wastewater ponds. J. Med. Entomol. 25:441-451.

Mullens, B. A. & J. L. Rodriguez. 1990. Cultural management of bluetongue virus vectors. Calif. Agric. 44:30-32.

Mullens, B. A. & D. A. Rutz. 1982. Development of immature *Culicoides variipennis* (Diptera: Ceratopogonidae) at constant laboratory temperatures. Ann. Entomol. Soc. Amer. 76:747-751.

Mullens, B. A. & R. K. Velten. 1994. Rearing *Culicoides variipennis sonorensis* (Diptera: Ceratopogonidae) on agar and nematodes. J. Med. Entomol. 31:175 177.

Muller, M. J. & N. A. Murray. 1977. Blood sucking flies feeding on sheep in eastern Australia. Aust. J. Zool. 25:75-85.

Nasci, R. S., R. E. McLaughlin, D. Fochs & J. S. Billodeaux. 1990. Effect of topically treating cattle with permethrin on blood feeding of *Psorophora columbiae* (Diptera: Culicidae) in a southwestern Louisiana rice-pasture ecosystem. J. Med. Entomol. 27:1031-1034.

Nelson, R. L. 1965. Carbon dioxide as an attractant for *Culicoides*. J. Med. Entomol. 2:56-57.

Nielsen, B. O. 1971. Some observations on biting midges attacking grazing midges in Denmark. Entomol. Scand. 2:94-98.

O'Rourke, M. J., E. C. Loomis & D. W. Smith. 1983. Observations on some *Culicoides variipennis* (Diptera: Ceratopogonidae) larval habitats in areas with bluetongue virus outbreaks in California. Mosq. News 43:147-152.

Paine, E. O. & B. A. Mullens. 1994. Distribution, seasonal occurrence, and patterns of parasitism of *Heleidomermis magnapapula* (Nematoda: Mermithidae), a parasite of

Culicoides variipennis (Diptera: Ceratopogonidae) in California. Environ. Entomol. 23:154-160.

Pearson, J. E., G. A. Gustafson, A. L. Shafer & A. D. Alstad. 1992. Distribution of bluetongue in the United States. In Bluetongue, African Horse Sickness, and Related Orbiviruses, T. E. Walton and B. I. Osburn, Eds., CRC Press, Boca Raton, FL, pp. 128-139.

Perez de Leon, A. A. & W. J. Tabachnick. 1995. Apyrase activity and ADP-induced platelet aggregation inhibition by salivary gland proteins of *Culicoides variipennis*, the North American vector of bluetongue viruses. Vet. Parasitol. (In press).

Schmidtmann, E. T., C. J. Jones & B. Gollands. 1980. Comparative host-seeking activity of *Culicoides* (Diptera: Ceratopogonidae) attracted to pastured livestock in Central New York State, USA. J. Med. Entomol. 17:221-231.

Schmidtmann, E. T., B. A. Mullens, S. J. Schwager & S. Spear, S. 1983. Distribution, abundance, and a probability model for larval *Culicoides variipennis* (Diptera: Ceratopogonidae) on dairy farms in New York State. Environ. Entomol. 12: 768 773.

Schmidtmann, E. T., R. Bobian & R. Belden. Soil chemistries define aquatic habitats with immature populations of the *Culicoides variipennis* complex. J. Med. Entomol. (In press)

Shemanchuk, J. A. & W. G. Taylor. 1984. Protective action of fenvalerate, deltamethrin and four stereoisomers of permethrin against black flies (*Simulium* spp.) attacking cattle. Pestic. Sci. 15: 557-561.

Shemanchuk. J. A., R. W. Spooner & L. R. Golsteyn. 1991. Evaluation of permethrin for protection of cattle aginst mosquitoes (Diptera: Culicidae), applied as electrostatic and low pressure sprays. Pestic. Sci. 32: 253-258.

Standfast, H. A., M. J. Muller & D. D. Wilson. 1984. Mortality of *Culicoides brevitarsis* (Diptera: Ceratopogonidae) fed on cattle treated with ivermectin. J. Econ. Entomol. 77:419-421.

Tabachnick, W. J. 1992. Genetic differentiation among populations of *Culicoides variipennis* (Diptera: Ceratopogonidae), the North American vector of bluetongue virus. Ann. Entomol. Soc. Am. 85:140-147.

Tabachnick, W. J. & F. R. Holbrook. 1992. The *Culicoides variipennis* complex and the distribution of bluetongue viruses in the United States. Proc. U.S. Anim. Hlth. Assoc. 96:207-212.

Tanner, G. D. & E.C. Turner, Jr. 1975. Seasonal abundance of *Culicoides* spp. as determined by three trapping methods. J. Med. Entomol. 12:87-91.

Tanya, V. N., E. C. Greiner & E.P.J. Gibbs. 1992. Evaluation of *Culicoides insignis* (Diptera: Ceratopogonidae) as a vector of bluetongue virus. Vet. Microbiol. 32:1 14.

Undeen, A. H. 1984. Biological control of the Psychodidae, Ceratopogonidae, and Simuliidae (Diptera), including new data on the effect of *Bacillus thuringiensis*, var. *Israelensis* on Simuliidae. Compar. Patholbiol. 7:143-153.

Walton, T. E., W. J. Tabachnick, L. H. Thompson & F. R. Holbrook. 1992. An entomologic and epidemiologic perspective for bluetongue regulatory changes for livestock movement from the USA and observations on bluetongue in the Caribbean basin. *In* Bluetongue, African Horse Sickness, and Related Orbiviruses, T. E. Walton & B. I. Osburn, [eds.], Proc. 2nd Internat. Sym., CRC Press, Inc., Boca Raton, FL

Wieser-Schimpf, L., W. C. Wilson, D. D. French, A. Baham & L. D. Foil. 1993. Bluetongue virus in sheep and cattle and *Culicoides variipennis* and *C. stellifer* (Diptera: Ceratopogonidae) in Louisiana. J. Med. Entomol. 30:719-724.

Whitsel, R. H. & R. F. Schoeppner. 1966. Summary of the biology and control of the valley black gnat, *Leptoconops torrens* Townsend (Diptera: Ceratopogonidae). Calif. Vect. News 13:17-25.

Wirth, W. W. 1977. Pathogens of Ceratopogonidae (midges). Pathogens of Med. Import. Arthropods. Bull WHO. 54:1-419.

Wirth, W. W. & W. R. Atchley. 1973. A review of the North American *Leptoconops* (Diptera: Ceratopogonidae). Grad. Stud. Texas Tech. Univ. No. 5, 57p.

Wirth, W. W. & R. H. Jones. 1957. The North American subspecies of *Culicoides variipennis* (Diptera: Heleidae). USDA Tech. Bull. 1170:1-35.

Black Flies

Description and biology. Black flies are small, 1 to 5 mm in length, robust, strong flying insects that have a humpbacked appearance, hence the common name buffalo gnat. The females of most species take blood from either mammals or avian hosts to support egg development, whereas males feed only on nectar and plant fluids. The immature stages of black flies are found in flowing waters, where they attach to various aquatic substrates and feed by filtering suspended organic matter. The eggs are either dropped freely into the water or laid on submerged acuatic plants or various other submerged substrates. Most species over-winter or over-summer in the egg stage. The larval period ranges from 12 days to 10 weeks and the pupal period ranges from 2 days to four weeks, depending on the species and water temperature. The entire life history from egg to adult ranges from 60 days to 15 weeks and over. Adult black fly longevity varies with the species and climate, but is usually no more than three weeks. Adult blackflies may disperse for considerable distances (miles) from immature stage development sites. Black flies are active in seeking blood meals during daylight hours and often persist throughout the entire day.

Economic Importance. Black fly feeding affects animal welfare and health in a variety of ways, including nuisance and fly worry, injection of saliva that elicits inflammatory responses, pathogenic toxicoses and allergic reactions (Cupp 1987). Dermatitis, focal hemorrhages and cutaneous and subcutaneous edema characterize localized host responses to black fly feeding (Frese & Thiel 1974). Lesions induced by black fly feeding may also serve as feeding sites for facultative blood feeding flies, such as eye gnats, *Hippelates*, and the face fly, *Musca autumnalis*, or as oviposition sites for myiasis-producing flies whose larvae feed in flesh wounds. Attack by massive populations of black flies are documented to induce toxemia that results in severe morbidity and even mortality. For example, in Canada outbreaks of *Simulium arcticum* have resulted in multiple episodes of death in livestock (Miller & Rempel 1944; Curtis 1954; Fredeen 1956, 1977). Massive outbreaks of *S. venustum* were reported to reduce milk production by 50% and also affected weight gain (Fredeen 1956). Monetary losses due to reduced production and death of over 300 cattle during a one week period in Canada were $500,000 (Hearle 1938). Rempel & Arnason (1947) reported the deaths of 133 domestic livestock that cost $20,000, and that another 600 animals died at a loss of $70,000. In California, Anderson & Voskuil (1967) reported a reduction in milk production during an outbreak of *S. trivittatum*, where milk yield went from a typical pre-outbreak herd average of 446 gallons every two days, to 398 gallons during the outbreak period (ca. 30 days), to 447 gallons every two days after the outbreak; monetary losses were $219 to $300 per dairy. Apart from the irrational and acute effects of black fly feeding, it is important to recognize that dairy cattle are regularly exposed to feeding black flies in major areas of the U.S.; the effects of this stress on health or production are unknown or unappreciated.

Because of their blood feeding activity, black flies also serve as vectors of disease causing agents. Although not well studied, the role of black flies as vectors of disease in dairy cattle is currently limited to the filarial nematode, *Onchocerca lienalis.* In New York state, *O lienalis* is transmitted by *S. jenningsi* (Lok et al. 1983); infections generally go undetected since there is little overt pathology (Scholtens et al. 1977). Black flies (*S. vittatum* and *S. notatum*) may biologically vector vesicular stomatitis virus (Cupp et al. 1992, Mead et al. 1997), but further study is needed using species common to New Mexico and Colorado where epizootics occur periodically.

Current Control Methods. Chemical control of blackflies was reviewed by Jamnback (1973), with most treatments directed at the larval stages that inhabit streams and rivers. Older methods using organophosphate insecticides have environmental limitations and have been replaced by formulations of *Bacillus thuringiensis* (B.t.) added to flowing waters where spores and crystal toxins are ingested by feeding larvae. Lacey & Undeen (1986) and Molloy (1989) have reviewed the development and use of B.t. as a black fly larvicide that is economical, effective and environmentally safe. Shemanchuk and Taylor

(1984) reported that treatment of cattle with synthetic pyrethroid compounds provided 70% or better protection from flakc fly blood feeding for at least one week. The use of insecticides and repellents applied directly to dairy cattle to protect against black fly feeding have not been accepted for practical use with dairy cattle. Area-wide aerial spraying for adults has proven effective, but is of limited utility due to cost concerns. Control Strategies that involve manipulation of the breeding sites by diverting water and removing attachment sites for blackfly egg laying have been reviewed by Jamnback (1967).

Current Support. None

Summary of Research and Extension Needs

(1) Economic Injury Level. Economic injury levels have not been determined for blackfly species that attack dairy cattle, thus the densities and seasonal activity periods that may lead to reduction in milk production, dermatitis or disease agent transmission need to be defined for major dairy regions.

(2) Sampling. Surveillance and collection techniques for immature blackflies in aquatic habitats was reviewed by morris (1960), while Service (1977) reviewed methods for sampling adult blackfly populations. Methods currently available for sampling populations of immature stages (larvae and pupae) in flowing waters are probably adequate for determining when control operations are necessary, but improved methods are needed for determining and predicting emergences of adult populations. Trap systems have been developed that simulate large mammal hosts and effectively attract large-mammal feeding blackfly species (Schmidtmann 19 , Anderson & Yee 1994).

(3) Biological-Ecological. Only limited information is available concerning various parameters of host attack (pest intensity) for black fly species that feed from dairy cattle in primary milk production regions.

(4) Pest-host Modeling. An inadequate data-base exists for developing models of black fly-host interactions.

(5) Research Classification

(a) Basic Research.
(1) Research should be directed to defining hydrologic factors that support production of economically important blackfly populations, including understanding of substrates, flow rates, and organic and chemical conditions, so that predictive capability is available.
(2) Develop better understanding of dispersal of adults from breeding sites to dairy farms where cows and heifers are exposed to black fly blood feeding.
(3) Refine quantitative sampling methods for adult and immature stages, including use of silhouette traps supplemented with host-odor stimuli (adults) and standardized larval recovery methods (larvae and pupae).

(b) Control Components Research.
(1) Determine and define types of economic loss in dairy cattle.
(2) Develop predictive methods for anticipating black fly outbreak situations.
(3) Evaluate formulations of synthetic pyrethroids for protecting cattle from black fly blood feeding.

(c) IPM Systems Research.
(1) Define phenology and population dynamics of large mammal-feeding species by dairy regions.
(2) Characterize environmental conditions affecting adult dispersal.
(3) Determine economic injury levels and correlate with black fly pest intensity based on adult sampling.

References Cited

Anderson, J.R. and G.H. Voskuil. 1963. A reduction in milk production caused by the feeding of black flies (Diptera:Simuliidae) on dairy cattle in California, with notes on the feeding activity on other animals. Mosq. News 23:126-131.

Anderson, J.R. and W.L. Yee. 1994. Trapping black flies (Diptera:Simuliidae) in Northern California. I. Species composition and seasonal abundance on horses, host models, and insect flight traps. J. Vect. Ecol. 20: 7-25.

Cupp, E.W. 1987. The epizootiology of livestock and poultry diseases associated with blackflies. pp. 387-395. In K.C. Kim and R.W. Merritt [eds.], Blackflies: Ecology, Population Management, an Annotated World List. Penn State Press, Univ. Park, PA.

Cupp, E.W., Mare, C.J., Cupp, M.S. & F.B. Ramberg. 1992. Biological transmission of vesicular stomatitis virus (New Jersey) by *Simulium vittatum* (Diptera:Simuliidae). J. Med. Entomol. 29:137-140.

Curtis, L.C. 1954. Observations on a black fly pest of cattle in British Columbia (Diptera:Simuliidae). Proc. Ent. Soc. British Columbia 51:3-6.

Fredeen, F.J .H. 1956. Black flies (Diptera:Simuliidae) of the agricultural areas of Manitoba, Saskatchewan, and Alberta. Proc.10th Int. Congr. Entomol. 10:819-823.

Fredeen, F.J .H. 1977. A reviev of the economic importance of black flies (Simuliidae) in Canada. Quest. Entomol. 13:219-229.

Frese, K. & W. Thiel. 1974. Zur pathologie der Hautveranderunger beim kriebelmucken-befull des Rindes. Zbl. Vet. Med. B, 21: 618-624.

Hearle, E. 1938. Insects and allied parasites injurious to livestock and poultry in Canada. Can. Dept. Agr. Publ, 604, 108p.

Jamnback, H. 1973. Recent developments in control of black flies. Ann. Rev. Entomol. 18:281-304.

Lacey, L. A., and A. H. Undeen. 1986. Microbial control of black flies and mosquitos. Annu. Rev. Entomol. 31: 265-296.

Lok, J. B., E. W. Cupp, M. J. Bernardo, and R. J. Pollack. 1983. Further studies on the development of *Onchocerca* spp. (Nematoda: Filaroidea) in Nearctic black flies (Diptera: Simuliidae). Am. J. Trop. Med. Hyg. 32:1298-1305.

Mead, D. G., C. J. Mare, and E. W. Cupp. 1997. Vector competence of select black fly species for vesicular stomatitis virus (New Jersey serotype). Am. J. Trop. Med. Hyg. 57: 42-48

Millar, J.L., and J.G. Rempel. 1944. Livestock losses in Saskatchewan due to black flies. Can. Jour. Comp. Med. and Vet. Sci. 8(12):334-337.

Molloy, D. P. 1989. Progress in the biological control of black flies with *B.t.i.*, with emphasis on temperate climates. In: H. De Barjac and D. J. Sutherland (eds.), Bacterial control of mosquitos and black flies, Rutgers University Press, New Brunswick, N. J.

Morris, R. F. 1960. Sampling insect populations. Ann. Rev. Entomol. 5:243-264.

Rempel, J., and A. Arnason. 1947. An account of three successive outbreaks of the black fly, *Simlium arcticum*, a serious livestock pest in Saskatchewan. Sci. Agr. 27:428-445.

Schmidtmann, E. T., (1987). A trap/sampling unit for ear-feeding black flies. Canad. Entomol. 119: 747-750.

Shemanchuk, J. A. and W. G. Taylor. (1984). Protective action of fenvalerate, deltamethrin, and four stereoisomers of permethrin against black flies (*Simulium* spp.) Attacking cattle. Pestic. Sci. 15: 557-561.

Scholtens, G., S. R. Adams, and J. R. Broderson. 1977. Evidence of onchocerciasis in Georgia cattle: prevalence at slaughter. Am. J. Vet. Res. 38: 1093-1097.

Service, M.W. 1977. Methods for sampling adult Simuliidae, with special reference to *Simulium damnosum* complex. Trop. Pest Bull. No. 5, Centre for Overseas Pest Research. London. 48p.

Mosquitoes

Description and Biology. Mosquitoes are small, 2-6 mm in length, fragile blood-sucking flies. The females of most species take blood meals to support egg maturation; males, and the females of certain species, do not blood feed. Mosquito eggs are laid singly or in rafts, either on water or on dry substrates that will be flooded by rain or tidal action. Larval and pupal stages develop in water. The length of the mosquito life cycle is quite variable, depending upon the species and the time of year. Under ideal conditions some species complete a generation every 10-16 days; other species have only one generation per year. Females may seek a blood meal every 3-5 days for up to a month. Some species show a narrow range of host preference, feeding only on mammals or birds; other species are variably opportunistic. In many regions of the United States, large populations of flood-water mosquitoes (*Aedes* and *Psorophora spp*.) develop in accumulations of rainfall and flood waters in low areas adjacent to or in pasturelands. These mosquitos readily attack dairy cattle. A considerable number of permanent water species (*Culex, Anopheles* and *Culiseta* spp.) also blood feed from cattle in various areas of the country.

Economic Importance. The annual losses in production and control costs due to mosquito attack are estimated at $5 million annually (Anon. 1976). The effects of mosquito attack on cattle have been largely overlooked because they have been generally considered to be to be human pests. Mosquitoes are efficient vectors of disease-causing agents, especially arboviruses and filarial nematodes, but are not known to transmit agents that cause disease in dairy-breed cattle in the United States. The rickettsial agent, *Anaplasma marginale*, has been mechanically transmitted by the interrupted feeding of *Psorophora columbiae* and *Aedes aegypti* (Howell et al. 1941). In Louisiana, the incidence of anaplasmosis in Holstein dairy cows increased during the mosquito season and followed outbreaks of the *Psorophora* sp. (Steelman et al. 1968), but this relationship is currently not recognized as a dairy problem.

In many areas of the United States mosquitoes occur in massive populations; however, considerable variation exists between regions and within regions due to species differences, season length and other climatic differences, and the effects of environmental use by man (irrigation of crop lands). The economic impact of mosquito blood feeding on dairy cow production or management in dairy regions has received little attention.

Current Control Methods. Area-wide control of adult mosquitoes is achieved by ultra low volume (ULV) applications of several organophosphate insecticides dispersed by specialized application systems contained in aircraft or ground vehicles (Lofgren 1970). Excellent control results have been obtained by area-wide ULV treatments, but they are only economically feasible when applied by an organized management program operating on injury thresholds (Steelman & Schilling 1978). This type of control program has been utilized for human protection by Mosquito Abatement Districts for many years and within the last 3-4 years has been shown to be quite feasible for controlling mosquitoes that attack livestock. The organized area-wide programs also utilize insecticides for the control of mosquito larvae as well as source reduction techniques (ditching, impoundment, etc,) in various types of aquatic habitats.

Treatment of cattle with residual insecticide, such as the synthetic pyrethroid permethrin, results in mortality of female mosquitoes feeding on cattle (McLaughlin et al. 1989), and also may reduce the numbers of mosquitoes in nearby areas (Focks et al. 1991). Further, Shemanchuk et al. (1991) reported that permethin applied as a total whole body spray to cattle is effective in preventing at least 70% suppression of mosquito blood feeding, thus providing a considerable measure of protection from field populations of mosquitoes.

Many biological control agents have been shown in laboratory and small-scale field tests to show promise for controlling mosquitoes (Chapman 1974). Most of the work to date has been directed toward the development of bio-control agents that can be used to control mosquito vectors of human disease. Insectivorous fish, especially *Gambusi*a *affini*s, have been effectively used by mosquito control agencies over the world (Gerberich & Laird 1968). Lacey & Undeen (1986) reviewed methods alternative to chemical insecticides for controlling riceland mosquitoes. Microbial control of mosquitoes with *Bacillus* spp. bacteria is included in this review. The use of microbial agents and insect growth regulator treatments for control of *Culex* mosquitoes in dairy waste-water lagoons has been evaluated by Mulla et al. (1984), Matamini et al. (1990), and Mulligan & Schaeffer (1990). Other approaches to microbial control of mosquitoes include the fungus *Lagenidium* (Jaronski & Axtell 1983, Kerwin & Washino 1987). The potential for controlling container-breeding mosquito species with the microsporidian parasite, *Edhazardia aedis*, also is being developed (Becnel & Johnson 1993).

A considerable amount of research has been conducted on genetic control of mosquitoes primarily regarding vector-control genetics (Pal & LaChance 1974). With sufficient understanding of the ecology and genetics of a mosquito population, genes may be introduced and gene frequencies manipulated as a valuable control method. Nevertheless, the vectorial capacity of genetically altered mosquito populations also may be affected, possibly resulting in enhanced transmission of animal disease agents or other factors that influence vectorial capacity (Gubler 1993).

Many types of source reduction to prevent mosquito larval development have been utilized by mosquito abatement districts to protect humans from adult mosquito attack for some 100 years (Riley & Johannsen 1938) These methods have included ditching and draining, flushing, and impoundment, all of which have been shown to be effective in managing mosquito populations, but these techniques are not being used specifically to manage mosquito populations that attack cattle. Moving cattle to pastures that do not have breeding areas or that are as far as possible from mosquito breeding habitat can reduce the losses caused by mosquito attack, and also may reduce mosquito reproductive potential.

Current Support. No current support is provided for research on the biology and/or control of mosquitoes as pests of dairy cattle.

Summary of Research and Extension Needs

(I) Economic Injury Levels. No data are available on milk production quantity or quality for dairy cattle exposed to mosquito attack in major dairy regions. The documentation of this type of data is necessary to provide information to the producer in order to initiate and maintain area-wide mosquito programs. Documentation of economic damage due to mosquito blood feeding also is needed to support implementation of producer-level control to offset dairy production losses.

(2) Sampling. Sampling materials and methods currently used by mosquito abatement districts can be used satisfactorily in mosquito management systems for livestock. Sampling methods include C02-baited light traps, landing rate counts, truck and boat mounted traps, larval dipping, soil sampling for eggs and resting station counts. Traps baited with cattle have also been shown to be a useful indicator of population trends. Field sampling methods for mosquitoes are reviewed by Service (1988). A reliable and standardized method is needed for accurately determining the pest intensity of mosquitos feeding on an animal.

(3) Biological-Ecological. Basic biological and ecological data currently available for selected areas is probably adequate to support an IPM program, but deficiencies exist. For example, data are not available on the longevity or the number of times that the female feeds for species that attack cattle. Further, little other life-table data are available for species that are known to use cattle as their primary source of blood, and for most species, little data exist on immature- or adult-stage mortality. Some data that indicate general dispersal patterns are available, but great diversity exists between species as well as geographic and weather conditions that influence the risk of dairy cattle to mosquito attack. While in some regions (or states) the species feeding on cattle are known, no information is available for many areas of the country.

4) Pest-Host Models. Models of the life history of pest and vector species, the diseases transmitted, the animal hosts, methods of control, abiotic and biotic factors, and their interactions need to be developed as key parameters that influence the total system. A computer simulation model based on life history parameters was published by Haile & Weidhaas (1977) for *Anopheles albimanus*. The model was used to simulate density trends of a field population and to theoretically compare effectiveness of insecticidal and genetic methods of control. A computer simulation model of management strategies for *Psorophora columbiae* in rice field agroecosystem was developed by Fochs & McLaughlin (1988).

(5) Research Classification.

a) Basic Research.
(1) Characterize quantitatively the attack levels (pest intensity) of primary mosquito species attacking dairy cattle in major dairy regions of the U.S.
(2) Determine economic injury levels for primary pest species in major dairy regions.
(3) Investigate physiologic/allergic/behavioral mechanisms that underlie economic losses attributable to mosquito feeding.

(b) Control Components Research.
(1) Further research microbial control methods.
(2) Further develop use of pyrethroid treatments for cattle for both protecting cattle from mosquito attach and for suppressing mosquito population density.
(3) Develop geographical data bases (GIS) to identify the characteristics of and develop predictability for cattle populations subject to mosquito attack

(c) IPM Systems Research. The mosquito management approaches for use in IPM of dairy cattle need to be tested to determine the conditions of most effective and least-cost use.

(d) Extension. Develop the importance of dairy cattle as a blood meal source for mosquitos as justification for mosquito abatement district control programs.

REFERENCES CITED

Becnel, J. J. & M. A. Johnson. 1993. Mosquito host range and specificity of *Edhazardia aedis* (Microspora: Culicosporidae). J. Amer. Mosq. Control Assoc. 9: 269-274.

Chapman, H. C. 1974. Biological control of mosquito larvae. Ann. Rev. Entomol. 19.33-59.

Focks, D. A. & R. E. McLaughlin. 1988. Computer simulation of management strategies for *Psorophora columbiae* in the rice agroecosystem. J. Amer. Mosq. Control Assoc. 4: 399-

Focks, D. A., R. E. McLaughlin & R. S. Nasci. 1991. Effect of permethrin treatment of cattle on *Psorophora columbiae* populations: Preliminary field test of a host management concept. J. Amer. Mosq. Control Assoc. 7: 1 -

Gerberich, J. B. & M. Laird. 1968. FAO Fisheries Tech. Paper No. 75. FAO United Nations, Rome, 1-70

Haile, D. G. & D. E. Weidhaas. 1977. Computer simulation of mosquito populations (*Anopheles albimanus*) for comparing the effectiveness of control technologies. J. Med. Entomol. 13:553-67.

Howell, D. E., G. W. Stiles, & L. H. Moe. 1941. The transmission of anaplasmosis by mosquitoes (Culicidae). J. Amer. Vet. Med. Assoc. 99:107-10

Kerwin, J. L. & R. K. Washino. 1987. Ground and aerial application of the asexual stage of *Lagenidium giganteum* for control of mosquitoes associated with rice culture in the Central Valley of California. J. Amer. Mosq. Control Assoc. 3: 59- .

Lacey, L. A. & A. H. Undeen. 1986. Microbial control of blackflies and mosquitoes. Annu. Rev. Entomol. 31: 265-296.

Lofgren, C. W. 1970. Ultralow volume applications of concentrated insecticides in medical and veterinary entomology. Ann. Rev. Entomol. 15:321-42.

Matanmi, B. A., F. A. Federici & M. S. Mulla. 1990. Fate and persistence of *Bacillus sphaericus* used as a mosquito larvicide in dairy wastewater lagoons. J. Amer. Mosq. Control Assoc. 6: 384-389.

McLaughlin, R. E., D. A. Focks & D. A. Dame. 1989. Residual activity of permethrin on cattle as determined by mosquito bioassays. J. Amer. Mosq. Control Assoc. 5: 60-63.

Molloy, D. P. 1989. Bacterial control of mosquitoes and blackflies: biochemistry, genetics and applications of *Bacillus thuringiensisisraelensis* and *Bacillus sphaericus*. In: H. de Barjac and Sutherland (eds.), Bacterial Control of Mosquitoes. Rutgers Univ. Press.

Mulligan, F. S., III, & C. H. Schaeffer. 1990. Efficacy of a juvenile hormone mimic, Pyriproxyfen (S-31183), for mosquito control in dairy wastewater lagoons. J. Amer. Mosq. Control Assoc. 6: 89-92.

Mulla, M. S., H. Axelrod, H. A. Darwazeh & B. A. Matanmi. 1988. Efficacy and longevity of *Bacillus sphaericus* 2362 formulations for control of mosquito larvae in dairy wastewater lagoons. J. Amer. Mosq. Control Assoc. 4: 448-452.

Nasci, R. S., R. E. McLaughlin, D. Focks & J. S. Billodeaux. 1990. Effect of topically treating cattle with permethrin on blood feeding of *Psorophora columbiae* (Diptera: Culicidae) in a southwestern Louisiana Rice-pasture ecosystem. J. Med. Entomol. 27: 1031-1034.

Pal, R. and L. E. LaChance. 1974. The operational feasibility of genetic methods for control of insects of medical and veterinary importance. Ann. Rev. Entomol. 19: 269-91.

Riley, W. A. and 0. A. Johannsen. 1932. Culicidae or mosquitoes. IN: Medical Entomology - a survey of insects and allied forms which affect the health of man and animals. McGraw-Hill Book Co., Inc. New York. 476 pp

Steelman, C. D., L. E. Foote, T O. Roby, and B. F. Hollon. 1968. Proc. Nat. Anaplasmosis Conf., Stillwater, OK, 5th, 178-82,

Steelman, C. D. & P. E. Schilling. 1977. Economics of protecting cattle from mosquito attack relative to injury thresholds. J. Econ. Entomol. 70: 15-17.

Cattle Lice

Description and Biology. Five species of lice infest cattle in North America; *Haematopinus eurysternus*, the shortnose cattle louse; *Haematopinusquadripertusus*, the cattle tail louse; *Linognathusvituli*, the longnose cattle louse; *Solenopotescapillatus*, the little blue cattle louse; and *Bovicola bovis*, the cattle biting louse. The cattle tail louse occurs rarely outside of California and Florida. The other four species are found in all cattle producing states. Thorough descriptions of each species can be found in Price and Graham (1997).

Life cycle similarities for *H. eurysternus*, *L. vituli*, *S. capillatus* and *B. bovis* have been well documented (Craufurd-Benson 1941, Lancaster 1957, Dubitsky 1959, Matthysse 1946, Mock 1974). Generally the life cycle from egg to egg approximates 26-29 days. Eggs, attached to the hairs of the host, hatch within 7-12 days. Nymphal lice complete development within 12-21 days. Adult lice may live 30-42 days and female lice produce from 1-3 eggs per day. *B. bovis* have parthenogenic reproduction, with sex ratios of 322 females to every male (Watson et al. 1997). The role of the male and the haplodiploidy determination of males and females have not been determined. Sex ratios for sucking the sucking louse species were slightly over 2 females per male (Watson et al. 1997). Hanlin (1994) discovered sex ratios of 50 females to every male, and 1 female for every male in summer populations of biting and sucking lice, respectively.

Seasonal dynamics for cattle lice in temperate regions of the world demonstrate a population increase in the cooler months and decrease in the warmer months on mature cattle (Watson 1984). Lice infestations may be greater on calves than cows and calf infestations persist until June (Geden et al. 1990). Seasonal changes in host hair coat, hair coat density, shedding, and self-grooming contribute to observed population fluctuation (Imes 1925, Jensen & Roberts 1966, Gojmerac et al. 1959, Lewis & Christensen 1962, Mock 1974). Subsequent studies show little evidence that hair length influences the distribution of cattle lice in either summer or winter (Hanlin 1994, Watson et al. 1997). Other factors contributing to the seasonal population dynamics of cattle lice include; light intensity and ambient temperature (Craufurd-Benson 1941), solar radiation (Matthysse 1946), and relative humidity (Mock 1974).

Economic Importance. Drummond et al. (1981) estimated dairy and beef cattle losses to cattle lice to be 126.3 million dollars annually. Greatest losses occur in young and old stock that experience poor feed conversion, reduced weight gain and limited productivity (Schemenchuck et al. 1960, Drummond et al. 1981, Gibney et al. 1985). DeVaney et al. (1992) found that Holstein calves with a dual infestation of gastrointestinal nematodes and cattle lice had mean weight gains of 11.4 and 10 kg less than uninfested control calves. Calves infested with only nematodes weighed 6.8 and 0.2 kg less than the control group and lice only (*L. vituli* and *H. eurysternus*) infested animals weights were 9.1 kg less than the controls. However, calves infested with *B. bovis* alone, gained 3.8 kg over the un-infested controls. Other damages caused by lice are a result of their feeding and movement. Loss of blood to sucking lice cause an elevated immune response, inflammation, anemia, and a general loss of condition in infested cattle (Peterson et al. 1953, Schemenchuck et al. 1960, Nelson et al. 1970). *B. bovis* does not feed on blood, but use mandibulate mouthparts to scrape and bite the skin and hair of the host. Large numbers of active lice irritate cattle causing severe itching, rubbing, and licking of infested areas. Considerable damage to the skin of the infested host can result from self excoriation (Baker & Oormazdi 1978). Losses to the leather industry from down-graded leather result from either the feeding of insects, or abraded and scratch damaged pelts.

Methods of Control. The effective use of insecticides can relieve suffering animals from severe infestations. Several insecticides are registered for use to control cattle lice. Unfortunately many fall under the organophosposphate classification and are under close scrutiny since passage of the Food Quality Protection Act. Compounds formulated as dusts, whole body sprays and concentrated pour-on can be used to manage cattle lice on lactating dairy stock. Pour-on and injectable endectocides are highly efficacious for cattle louse control (Drummond 1985, Losson and Lonneux 1996, Lloyd et al. 1996), however such systemic compounds are restricted from use on dairy cattle of breeding age. Some spray, pour-on and injectable systemic compounds are restricted from use on dairy cattle of breeding age.

(1) Insect Growth Regulators. Insect growth regulators have not been fully explored for louse control. Juvenile hormone (JH) analogs appear to be effective for the control of shortnose cattle lice experimentally (Meleney & Roberts 1975), however no JH based compounds are currently in use. The chitin synthesis inhibitor diflubenzuron is highly efficacious against cattle lice (J. E. Lloyd, personal communication.)

(2) Biological Control. Few natural enemies of cattle lice are known. The entomopathogenic fungus, *Trenomyces histophtorus* (order Laboulbeniales) has been isolated from infected chicken lice (Meola & DeVaney 1976) and *L. vituli* (J.E. Lloyd, personal communication). DeVaney et al. (1988) observed a similar fungal infection of *H. eurysternus*, *L. vituli*, and *B. bovis* infesting calves in Texas. *T. histophtorus* hyphae invaded the hemocoel of the host, and caused a fatal septicemia. Interestingly, other entomopathogenic fungi have not been explored for louse control, particularly fungi with broad host ranges. For example, *Beauveria bassiana* has been collected from 8 orders of insects (Humber 1992). *B. bassiana* can be cultured on artificial media, and the infective conidia can be harvested and stored frozen for several months without a reduction in viability. Insects may be infected topically or by ingestion. Furthermore, some strains of *B. bassiana* and *Metarhizium anisopliae* fungi produce mycotoxins that kill the insect host within a day to two of infection (Grove & Pople 1980).

Other toxin producing entomopathogens include the bacterium *Bacillusthuringiensis*. New strains of *B. thuringiensis* have been recovered from a variety of habitats (Smith & Couche 1991), including sheep wool (Drummond et al. 1992). Nine *B. thuringiensis* strains isolated from sheep wool were compared with several non-wool strains of *B. thuringiensis* for control of the sheep biting louse, *Damalinia ovis*. All sheep derived strains were toxic to *B. bovis*. Because *B. thuringiensis* endotoxin must be ingested and solubilized in the gut to be effective, *B. bovis*, which feeds on the skin of cattle, may be the only louse species to ingest sufficient endotoxin to induce mortality.

Current Support. Currently, there are no research projects devoted to the biology and ecology of cattle lice. The pesticide industry continues to support the development of chemical controls of cattle lice (< 1 SY).

Summary of Research and Extension Needs

Establishing economic injury levels has been difficult. Although many authors have examined the effect of lice on beef cattle weight, significant losses due to light or moderate infestations in mature cattle have been inconclusive (Collins & Dewhirst 1965, Scharff 1962, Tweedle et al. 1977, Gibney et al. 1985). Heavily infested mature cattle suffer poor weight gains and conditioning (Collins & Dewhirst 1965, Scharff 1962). Furthermore, Holstein calves with multiple infestations of gastrointestinal nematodes and cattle lice had reduced weight gains (DeVaney et al. 1992). The importance of multiple parasitism to the growth and development of cattle has not been examined. Future host-parasite interaction studies must define the impact of cattle lice in terms of host breed, host age, nutrition, and infestation level, with efforts directed toward reducing lice infestations as low as possible using IPM.

Developing an accurate, widely acceptable sampling method for determining the presence or absence of lice is needed. From a production perspective examining known predilection sites on the host will simplify the sampling procedure (Hanlin 1994, Watson et al. 1997). *S. capillatus* are most common on the face and muzzle of the host, *H. eurysternus* on the ears, dewlap and neck, *L. vituli* on the neck and back and *B. bovis* on the poll and withers. Examining these areas of the host for lice will give the best results for the sampling effort. Usually counts are conducted by parting the hair coat of the host and determining the numbers of lice within the area. The most common methods of estimating cattle louse populations involve a rating system of the infestation or an actual count of the individuals (Craufurd-Benson 1941, Matthysse 1946, Watson 1984, Gibney et al. 1985, DeVaney et al. 1988). However these methods require visually detectable louse populations. Immunodiagnostic techniques (Elisa) could be used to determine the presence of lice before louse populations reach detectable levels using the conventional monitoring procedures.

The distribution of *S. capillatus* and *H. quadripertusus* to largely definitive regions of the host (head and tail, respectively) suggests that these regions must favor the growth and development of these species. The mechanisms that influence the distribution on the host are not understood. The role of the host's immune system relative to louse infestations has not been explored. Louse infestations are often higher on calves than on older cattle, suggesting that an acquired immunity may be a factor in the degree of infestation (Mock 1974, Kennedy & Kralka 1986, Geden et al. 1990). Recent studies of the immune response of sheep to louse infestations have demonstrated progress in the understanding of host/parasite interactions (James and Moon 1998, James et al. 1998). Basic studies of the host immune response to louse infestations are needed. Studies of the` immune response to louse antigens could lead to the development of a louse vaccine similar to concealed antigen vaccines recently developed against *Boophilus microplus* (McKenna et al. 1998). Limiting the development of vaccines and novel ectoparasiticides is the inability to rear lice off the host. To date no artificial rearing techniques have been successful.

The discoveries of natural louse enemies by DeVaney et al. (1992) and Drummond et al. (1992) demonstrate the potential of biological control as an alternative to chemical insecticides for louse control. The technology exists for the exploration of non-traditional sources of fungi and bacteria. Once implemented, this technology may lead to the development of innovative IPM programs. Feasibility studies of these potential biological control agents are needed.

Lancaster & Meisch (1986) recommend the prophylactic treatment of all animals being introduced into a louse free herd. Animal housing may afford a convenient opportunity to initiate a whole farm louse management program. Geden et al. (1990) observed that stanchioned cows were more likely to have louse infestations than cows in free stalls. Furthermore, calves held inside barns had significantly more lice than calves held in outdoor hutches. They speculated that animal to animal transmission and exposure to direct sunlight may have contributed to the differences in louse populations. Isolating replacement calves in outdoors hutches allows the producer time to examine the each calf for the presence of lice and the subsequent treatment of infested animals without exposing the entire calf group. The potential of animal housing as a cultural control measure requires further study.

References Cited

Baker, K. P. & H. Oormazdi 1978. The probable cause of the multiple linear scratch defect of cattle hides in Ireland. J. Soc. Leather Tech. Chem. 62: 103-10

Collins, R. C. & L. W. Dewhirst. 1965. Some effects of the sucking louse, *Haematopinus eurysternus*, on cattle on unsupplemented range. J. Amer. Vet. Med. Assoc. 146: 129-131.

Craufurd-Benson, H. J. 1941. The cattle lice of Great Britain. Part I. Biology, with special reference to *Haematopinus eurysternus*. Part II. Lice populations. Parasitol. 33: 331-358

DeVaney, J. A., L. D. Rowe & T. M. Craig. 1988. Density and distribution of three species of lice on calves in central Texas. Southwest. Entomol. 13: 125-130.

DeVaney, J. A., T. M. Craig, L. D. Rowe, C. Wade & D. K. Miller. 1992. Effects of low levels of lice and internal nematodes on weight gain and blood parameters in calves in central Texas. J. Econ. Entomol. 85: 144-149.

Drummond, R. O. 1985. Effectiveness of ivermectin for control of arthropod pests of livestock. Supplement to the Southwestern Entomologist. 7:34-42.

Drummond, J., D. K. Miller & D. E. Pinnock. 1992. Toxicity of *Bacillusthuringiensis* against *Damalinia ovis* (Phthiraptera: Mallophaga). J. Invert. Pathol. 60: 102 103.

Drummond, R. O., G. Lambert, H. E. Smalley Jr. & C. E. Terrill. 1981. Estimated losses of livestock to pests. *In* D. Pimentel (ed.), CRC Handbook of Pest Management in Agriculture, vol. 1. CRC Press, Boca Raton, FL. pp. 111-127.

Dubitsky, A. M. 1959. Seasonal dynamics of cattle tick [sic lice] abundance in southeast Kazakhstan. *In* N. D. Levine (ed.), Natural Nidality of Diseases and Questions of Parasitology. Proceedings IV. Conference. Univ. Illinois Press. Urbana, IL.

Geden, C. J., D. A. Rutz & D. R. Bishop. 1990. Cattle lice (Anoplura: Mallophaga) in New York: Seasonal population changes, effects of housing type on infestations of calves, and sampling efficiency. J. Econ. Entomol. 83: 1435-1438.

Gibney, V. J., J. B. Campbell, D. J. Boxler, D. C. Clanton & G. H. Deutscher. 1985. Effects of various infestation levels of cattle lice (Mallophaga: Trichodectidae & Anoplura: Haematopinidae) on feed efficiency and weight gains of beef heifers. J. Econ. Entomol. 78: 1304-1307.

Gojmerac, W. L., R. J. Dicke & N. N. Allen. 1959. Factors affecting the biology of cattle lice. J. Econ. Entomol. 52: 79-82.

Grove, J. F. & M. Pople. 1980. The insecticidal activity of beauveracin and enniatin complex. Mycopathologia. 70: 103-105.

Hanlin, S. J. 1994. The summer distribution of cattle lice in Wyoming. M. S. thesis, University of Wyoming, Laramie.

Humber, R. A. 1992. Collection of entomopathogenic fungal cultures. Catalog of strains. ARS-110. USDA-APHIS, National Biolog. Control Inst. Ithaca, NY.

Imes, M. 1925. Cattle lice and how to eradicate them. Farmers Bull. 909. USDA.

James, P. J., R. D. Moon, and D. D. Ragsdale. 1998. Serum and skin surface antibodies and their associations with sheep biting lice, *Bovicola ovis* on experimentally infested sheep. Med. Vet. Entomol. 12:276-283.

Jensen, R. E. & J. E. Roberts. 1966. A model relating microhabitat temperatures to seasonal changes in the little blue louse (*Solenopotescapillatus*) populations. GA. Agric. Exp. Stn., Athens, GA.

Kennedy, M. J. & R. A. Kralka. 1986. A survey of ectoparasites on cattle in central Alberta, November 1984-July 1985. Can. Vet. J. 27: 459-460.

Lancaster, J. L. 1957. Cattle lice. Agric. Exp. Sta. Bull. 591. Univ. of AK.

Lancaster, J. L. & M. V. Meisch. 1986. Lice. *In* Arthropods in Livestock and Poultry Production. Wiley & Sons. New York, NY.

Lewis, L. F. & D. M. Christensen. 1962. Induced build-up of populations of *Bovicolabovis* on cattle in Oregon. J. Econ. Entomol. 55: 947-949.

Lloyd, J. E., R. Kumar, J. W. Waggoner, and F. E. Phillips. 1996. Doramectin systemic activity against cattle grubs, *Hypoderma lineatum* and *H. bovis* (Diptera: Oestridae), and cattle lice, *Bovicola bovis* (Mallophaga: Trichodectidae), *Linognathus vituli* and *Solenopotes capillatus* (Anoplura: Linognathidae), and *Haematopinus eurysternus* (Anoplura: Haematopinidae), in Wyoming. Vet. Parasitol. 63:307-317.

Matthysse, J. G. 1946. Cattle lice: their biology and control. Cornell University Agricultural Experiment Station Bulletin no. 832.

McKenna, R. V., G. A. Riding, J. M. Jarmey, R. D. Pearson, and P. Willadsen. 1998. Vaccination of cattle against the tick *Boophilus microplus* using a mucin-like membrane glycoprotein. Parasite Immunology. 20: 325-336.

Meleney, W. P. & I. H. Roberts. 1975. Insect juvenile hormone mimics against the short-nosed cattle louse, *Heamatopinus eurysternus* Denny (Anoplura), and their effect on warbles of *Hypoderma* spp. (Diptera: Oestridae). J. Parasitol 61: 956 959.

Meola, S. & J. DeVaney. 1976. Parasitism of Mallophaga by *Trenomyceshistophtorus*. J. Invert. Pathol. 28: 195-201.

Mock, D. E. 1974. The cattle biting louse, *Bovicola bovis* Linn. I. *In-vitro* culturing, seasonal populations, and role of the male. II. Immune response of cattle. Ph.D. dissertation, Cornell University, Ithaca, NY.

Nelson, W. A., J. A. Schemenchuck & W. D. Haufe. 1970. *Haematopinuseurysternus*: Blood of cattle infested with the short-nosed cattle louse. Exp. Parasitol. 28: 263-271.

Peterson, H. O., I. H. Roberts, W. W. Becklund & H. E. Kemper. 1953. Anemia in cattle caused by heavy infestations of the blood-sucking louse, *Haematopinuseurysternus*. J. Amer. Vet. Med. Assoc. 122: 373-377.

Price, M. A. and O. H. Graham. 1997. Chewing and Sucking Lice and Parasites of Mammals and Birds. USDA/ARS, Technical Bull. 1849. 309 pp.

Scharff, D. K. 1962. An investigation of the cattle louse problem. J. Econ. Entomol. 55: 684-688.

Schemenchuck, J. A., W. O. Haufe & C. O. M. Thompson. 1960. Anemia in range cattle heavily infested with the short-nosed sucking louse, *Haematopinuseurysternus* (Anoplura: Haematominidae). Can. J. Comp. Vet. Sci. 24: 158-161.

Smith, R. A. & G. A. Couche. 1991. The phylloplane as a source of *Bacillusthuringiensis* varients. Appl. Environ. Microbiol. 57: 311-315.

Tweedle, N. E., L. J. Cummins & J. F. Grahm. 1977. The prevalence of cattle lice and their effect on live weight changes of beef cattle. Proc. 54th Ann. Conf. Aust. Vet. Assoc. p. 89-91.

Watson, D. W. 1984. The dynamics and distribution of cattle lice populations on naturally infested steers. M. S. thesis, University of Wyoming, Laramie.

Watson, D. W., J. E. Lloyd and r. Kumar. 1997. Density and distribution of cattle lice (Phthiraptera: Haematopinidae, Linognathidae, Trichodectidae) on six steers. Veterinary Parasitology 69: 283-296.

Mange Mites

Description and Biology. Of five species of mites associated with cattle, *Sarcoptesscabiei bovis* and *Chorioptesbovis* occurs most frequently on dairy cattle. *Psoroptes ovis* seems to be restricted to beef cattle (Hourrigan 1979). Infestation by mites can result in a condition known as mange, which is caused by a combination of mite feeding, host immune response and injury due to scratching and rubbing. Mange is primarily a winter condition characterized by hair loss, thickened skin, raw lesions and scabbing. Summaries of the mites' life cycles, impact, and management are provided by Hall (1985) and Schmidtmann (1985).

Sarcoptes mites occur on cattle in the brisket and around the base of tail. Males, larvae and nymphs occur on hair follicles and the surface of the skin. Females burrow into the skin. Development from egg to adult requires 10-13 days. Transfer is mainly by direct contact.

Chorioptes infestations are widespread in temperate areas. The entire life cycle is also passed on the host, requiring 18 - 28 days for completion (Sweatman 1957). These do not burrow into the skin, but scabbing may occur. Mange occurs in the neck, tail and lower legs.

Economic Importance. *Sarcoptes scabei* infestations, resultant mange and induced behavioral changes in cattle can cause decreases in feed-conversion efficiency, reduced milk production of 10-15%, mechanical injuries, emaciation and even death (Baker & Howe 1950, Nusbaum et al. 1975). Sarcoptic mange has been the subject of a Federal-State eradication program. The parasite has become reestablished (Nusbaum et al. 1975) but its present status is unclear.

The economic impact of chorioptic mange has not been evaluated.

Methods of Control. Mange is currently controlled using sprays and systemic parasiticides. Most compounds cannot be used for treatment of lactating animals. Recently, eprinomectin has been approved for use on lactating dairy cattle, and provides mange control for at least 56 days (Barth, et al. 1997).

Current Support. Research support is non-existent.

Summary of Research and Extension Needs

(1) Economic Injury Level. Since sarcoptic mange is a reportable and quarantinable disease, a single mite represents the economic injury level. Economic injury levels for chorioptic mange need to be established.

(2) Sampling. Sampling *Sarcoptes* requires taking deep skin scrapings of infected areas and examining the scrapings for active mites, or dissolving scraping in 10 % KOH and examining the residue for mites (Schwardt 1949). Diagnostic methods must be specific due to regulatory needs. Use of immunodiagnostic methods has not been explored, but may be worthwhile.

(3) Biological and Ecological. Role of wildlife and fomites as sources of transmission requires investigation. The influence of management factors such as housing on the predisposition to outbreaks requires investigation. Immunological studies on host susceptibility and resistance to mange also requires evaluation.

(4) Legal Barriers. Mange is a reportable disease. Eradication programs must be coordinated through State and Federal regulatory agencies. Coordinated efforts will be more effective than reliance on producer-level control. Also, currently registered systemic parasiticides cannot be used on lactating animals.

References Cited

Baker, D. W. & G. G. Howe. 1950. Cattle mange in New York State. J. Am. Vet. Med. Assoc. 116: 280-281.

Barth, D., J. A. Hair, B. N. Kunkle, W. L. Langhoff, M. Lowenstein, S. Rehbein, L. L. Smith, J. S. Eagleson and E. Kutzer. 1997. Efficacy of eprinomectin against mange mite in cattle. Am. J. Vet. Res. 58: 1257-1259.

Hall, R. D. 1985. Mites of veterinary importance, pp. 151-181. *In* R. E. Williams, R. D. Hall, A. B. Broce & P. J. Scholl (eds.), Livestock Entomology. Wiley, NY.

Hourrigan, J. L. 1979. Spread and detection of psoroptic scabies of cattle in the United States. J. Am. Vet. Med. Assoc. 175: 1278-1280

Nusbaum, S. R., F. J. Drazek, H. Holden, T. J. Love, J. Marvin, D. Rowe & L. L. Tyler. 1975. *Sarcoptes scabei bovis* - a potential danger. J. Am. Vet. Med. Assoc. 166: 252-256.

Schmidtmann, E. T. 1985. Arthropod pests of dairy cattle, pp. 223-238. *In* R. E. Williams, R. D. Hall, A. B. Broce & P. J. Scholl (eds.), Livestock Entomology. Wiley, NY.

Schwardt, H. H. 1949. Diagnosis and control of mange in dairy cattle. J. Econ. Entomol. 42: 444-446.

Sweatman, G. K. 1957. Life history, non-specificity, and revision of the genus *Chorioptes*, a parasitic mite of herbivores. Can. J. Zool. 35: 641-689.

Cattle Grubs

Description and Biology. Cattle grubs are the parasitic immature stages of the well known "heel fly." Female heel flies, which are non-feeding and bee-like in appearance, attach their eggs only to hairs on the legs or lower body regions of pastured cattle. After hatching from the egg, the larval cattle grub penetrates the host's skin and undergoes an extended period of migration in tissues of the host. Ultimately, after 6 to 8 months, the grub positions itself just under the skin of the back. A respiratory opening is cut soon after a larva migrates to the skin. Here, in a cyst-like "warble" of host origin, the grub undergoes further maturation for 30-90 days. When mature, the grub exits the warble via the respiratory opening, falls to the ground, and undergoes metamorphosis to the adult fly. The adult emerges about 4 to 5 weeks after pupation.

Adult heel flies are strong, robust flyers and are active only for a limited period each year. Two species, the common cattle grub, *Hypoderma lineatum,* and the Northern cattle grub, *H. bovis,* are widespread in the United States. The common cattle grub is found throughout the U.S. and up into Canada. The distribution of the northern cattle grub is restricted to Canada and states in the northern two thirds of the U. S.

Economic Importance. The running or "gadding" of pastured cattle to avoid the egg laying of heel flies may result in physical injuries, interrupt grazing patterns, reduce milk flow, and rate of weight gain. Current data on economic losses attributable to the effects of cattle grubs on milk production or dairy operations in general are unavailable. A review of literature on the economic impact of cattle grubs on beef production (Drummond 1987) reports significant differences related to cattle grub infestations in the average daily gain of treated and untreated confined cattle. Losses at slaughter from excess trimming and from hide damage were also reported, but profits from cattle grub control benefit abattoirs, not the applicator of the treatment. Reliable data to support arguments that the control of cattle grubs in dairy herds is economically beneficial are unavailable.

Methods of Control. EprinexTM, a pour-on formulation of the macrocyclic lactone eprinomectin, is labeled for use against cattle grubs in lactating dairy cows with no milk withholding period and no milk discard. A variety of parasiticides are available for treating heifers and dry cows.

Current Support. No SYs are currently devoted to research on the control of cattle grubs in dairy cattle.

Summary of Research and Extension Needs

(1) Economic Injury Levels. Economic injury levels for cattle grubs in beef cattle have little relevance to dairy cattle, which are either marketed for veal prior to exposure, or reared to maturity as milkers. Any effect of grub infestation on weight gain or meat loss would thus be minimized. Carry-over hide damage from grub infestations acquired as young stock may affect hide quality in older cull cows, but information concerning this relationship is unavailable.

(2) Sampling. As developing cattle grubs are readily observed in the skin of host cattle for a relatively fixed time interval each year, sampling populations is a straightforward procedure. Grub populations may also be readily mapped and sampled periodically to develop specific information concerning rates of development and emergence under normal and manipulated conditions. Immunodiagnostic sampling methods are available if ever needed for area-wide monitoring.

(3) Control Components. At this point there is no need for IPM to control cattle grubs in dairy cattle.

(4) Biological-Ecological. Information is needed concerning the relationship between adult population density and subsequent grub populations. The role that acquired or enhanced immunity plays in regulating cattle grub populations should be investigated relative to future area-wide grub control programs. Lastly, an appreciation of the degree to which lactating cows, which are commonly exposed to grubs as heifers, contribute to populations of adult heel flies is needed in lieu of the lack of control procedures for treating milking cows.

(5) Pest-Host Models. Certain elements of a model for cattle grubs, notably ecological and physiological data concerning natural mortality mechanisms and population frequency distributions (Breyev 1976), have been developed. Additional information is needed on a regional basis to develop model predictive capability for estimating the impact of manipulative strategies.

(6) Legal Barriers. Fundamental barriers will continue to exist regarding use of systemically active compounds for controlling cattle grubs in dairy cows.

References Cited

Breyev, K. A. 1976. Solia Parasitol. 23: 343-356.

Drummond, R. O. 1987. *In* Proceedings of the MSD AGVET Symposium, The Economic Impact of Parasitism in Cattle, p. 9-24.

****
****

End of Dairy section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****
****

RANGE CATTLE SUMMARY

Statistics show that cattle and calves continued to be the leading cash commodity with the combined livestock and poultry income exceeding that from crops. Livestock producers continued to have generally good animal health products available resulting from research conducted in the public and private sector. A problem that has developed since the 1979 IPM workshop is that of resistance in the horn fly populations throughout most of the cattle producing areas. Another is that of the lack of new chemistries being produced and developed in the animal health industry. Not only are fewer products being developed, some product loss is resulting from re registration requirements by Environmental Protection Agency.

There is continued concern for the development of alternative pest controls to make products available to the producer which are safer to the applicator, the host animal, and the environment. To do this will require the integration of knowledge and technology into management systems to be used by the animal health industry. The 1979 workshop identified requirements necessary before IPM strategies could be implemented for the pests for range cattle. The following is a list of accomplishments that were completed to help implement these strategies.

- The screwworm has been eradicated from the United States and Mexico and much of Central America by integrating chemicals and sterile male releases. This is probably the greatest entomological feat ever in pest control efforts. Interest is being expressed in South America for a screwworm program.
- A joint Canadian-USA project successfully demonstrated the effect of integrating systemic insecticide treatments with the use of sterile insect releases. Populations of *Hypoderma bovis* and *Hypoderma lineatum* were eradicated in field trials.
- A vaccine has been developed that would be effective in reducing *Hypoderma* populations. The vaccine is not damage-protecting but significantly reduces the production of offspring thus reducing the biotic potential.
- Significant biological information on the cattle grub has been developed.
- The resistance mechanism in the horn fly making them resistant to pyrethroids has been identified.
- Strategies for management of resistant populations of horn fly to pyrethroids have been developed.
- Biological-ecological studies have provided information on the behavior, flight range, etc., of horn fly and face fly resulting in population models.
- The economic impact of both horn fly and face flies have been developed.
- Control of all range cattle pests have benefitted from the development of avermectins, pyrethroids, IGR's and microbes. Formulation chemistry has provided a list of delivery systems for fly and tick control.
- The role of habitat and wildlife management has been identified for tick control. Wildlife plays a much larger role in the maintenance of *Boophilus* ticks than previously thought.

With these accomplishments duly noted, the present workshop identified additional requirements for the continued development of IPM systems.

1. Enhancement of Livestock Insect Extension and Research Programs.

- The last decade has witnessed the decline in the number of extension personnel working with producers. This coupled with a decline in research personnel will erode the technology base available to the producer.
- Information delivery systems utilizing technology developments need to be enhanced and made available to producers. With the decline in extension personnel, this role will unfortunately become a greater part of research responsibilities, if new technologies are to be available to producers.

2. Development and Incorporation of IPM Strategies into Computer-aided Decision Management Systems for Animal Production.

- In order for these computer (SMART) systems to be most helpful, injury thresholds for range cattle pests need to be refined or developed.

- The relationships between the endo- and ecto-parasite burden of livestock need to be developed to provide the producer a "total" animal health package.

3. Development of Environmentally Compatible Control Strategies/Tactics.

- Research will need to seek new chemistries to replace those not being re registered and to encourage the animal health industry to further develop new products.
- Development of alternative tactics or alternative use patterns for established tactics to make them safer.
- Identification and selection of resistant animals and determine the genetic basics for this resistance for use in breeding programs to develop pest resistant/tolerant animals.
- Direct the development of strategies and tactics to the smaller producer who will have greater control of herd operations.

4. Biology-Ecology Studies to Support Decision Management Systems.

- Field studies such as host-parasite and host landscape interactions, diapause and dispersal need to be continued.
- Resistance monitoring systems need to be refined and made more sensitive to more quickly identify changes in resistance levels.

5. Surveillance/Quarantine.

- Biological databases for potential exotic pests need to be developed. Literature surveys of exotic potential pests in the Caribbean, Central America, South America, and Africa need to be conducted and compiled for ready reference.
- Database of possible chemical control(s) for exotic pests would be helpful to quarantine personnel
- Eradication efforts of pests already underway (screwworm) should be maintained and expanded into the Caribbean to completely protect North America.

6. Interdisciplinary Interactions.

- Increase cooperative interaction between biological sciences, production animal scientists, forage and range management specialists and economists is encouraged to develop a more complete insect pest management system.

RANGE CATTLE

Committee Members

Dr. Sidney E. Kunz, Chair
Dr. Alberto B. Broce
Dr. Jerry F. Butler
Dr. Ronnie L. Byford
Mr. William F. Fisher
Dr. Lane D. Foil
Dr. John E. George
Dr. Reid R. Gerhardt
Dr. Fred R. Holbrook
Dr. H. Grant Kinzer
Dr. John E. Lloyd
Dr. Lamar Meek
Dr. Rick J. Meyer
Dr. John A. Miller
Dr. Gary R. Mullen
Dr. C. Dayton Steelman
Dr. Pete D. Teel

Economic Importance of Range Cattle and Industry Trends

Livestock continues to be the leading cash crop of all farm production. Cattle/calves was the leading cash commodity for 1994 with an income of $37,882 million; combined livestock and poultry income, $86,358 million, compared to $84,810 million income for all crops.

Livestock as rich sources of essential amino acids, fats, vitamins, and minerals for human needs, are the only organisms that can convert cellulose in plants into food for human consumption. Much of the land of the United States is not capable of producing cultivated crops but does produce abundant forage for livestock production. Most of these forages are consumed on or near the source of production and do not enter the market channels directly. Consequently, the important economic values of forages are largely unrecognized as they are marketed in the form of an animal product.

In the United States, range cattle operations will make best use of this renewable resource. The cow/calf unit is the basic production unit providing the animals to convert this resource to a marketable product. Cattle are produced in every state in the nation and in each state there are vast acreages which will more efficiently produce forage than crops. The only way to convert cellulose plants into food for human consumption is through livestock. If, in the future, confined feeding operations decline because of environmental problems, these grazing lands will be even more important.

The most important external pests of rangeland cattle are the biting flies (horn flies, mosquitoes, culicoides, horse flies and black flies), face flies, cattle grubs, lice, mites and ticks. Generally, the American producer has not been faced with serious pest control problems, except for horn fly control due to resistance. With the exception of the tabanid group (horse and deer flies), the producer has had an effective arsenal of insecticides and application techniques available that, if necessary or if he chooses to do so, could effect an adequate pest control program.

Horn Fly, *Haematobia irritans* (L.)

Description and Biology. The horn fly is an obligate blood-sucking parasite of cattle. The adults spend most of their time on cattle either feeding or resting with the female leaving its host only for brief periods of time to oviposit on fresh manure. The fly feeds 24-38 times per day, thus causing continued annoyance to its host (Harris et al. 1974). Their dependence on bovine blood is not complete, but Butler et al. (1995) demonstrated that a component of bovine erythrocytes is required for horn flies to complete oogenesis in the laboratory. Anticoagulant activity in the horn fly is not accomplished through the use of apyrase inhibition of platelet aggregation, as is true in many ectoparasites, and their saliva appears to contain no vasodilators (Cupp et al. 1998). Obviously a great deal of coevolution went into this system. Depending on the location, several thousand flies may build up on cattle during the peak season of activity. Flies are active 3-4 months in the northern part of the country with some fly activity being present during most of the year in the southernmost area.

Development rates of the immature stages are of course temperature dependent. The immatures can develop over the range of approximately 10-35oC with an optimum of approximately 25-27oC. Within this range, eggs can hatch in as little as 11-12 hr or require as long as 4-5 days. The lst instar larvae burrow into the manure pat where development continues through 3 larval instars. The larval period can range from 3-15 days. Pupation takes place either in the pat or in the soil immediately beneath or around the pat with the pupal period lasting from 4-30 days. In the southern climate, adult emergence will occur within 7-9 days in peak season; in the north this time is considerably longer with up to 4 weeks required in Canada.

Economic Importance. Insects, ticks, and mites cost the U.S. livestock producer in excess of $3 billion annually; loss of over $2 billion annually is suffered by the beef cattle industry. The horn fly is a major pest of the industry and inflicts an estimated loss in excess of $800 million annually (Kunz et al. 1992). These losses are manifested in livestock in the form of reduced efficiency of feed conversion, reduced weight gains and decreased milk production. They are the results of blood loss, annoyance, irritation, and behavioral responses to avoid the discomforts caused by the pest. The major costs of control of livestock pests are insecticides, labor, and equipment. Morrison and Foil, in Louisiana, found that 12 weeks of horn fly treatment of fall calving cows did not result in significant changes in calf weaning weight or body condition scores. However, research in Louisiana by Derouen et al. (1995), in which mean horn fly control was moderate (@68%), found that weight gains in treated cattle was 17% greater for treated cattle and that cross breeding with Brahman cattle led to improved animal performance as well. Although estimates of the quantity of pesticide applied solely for horn fly control are unavailable, an estimated 10-12 million pounds of insecticides are applied annually to livestock at a cost of $60 million for the pesticides alone. No figures are available on the cost of application (labor and debilitation to animals); however, these probably exceed the cost of the pesticide. Riley et al. (1994) developed a serum profile for infested and uninfested cattle. Under laboratory conditions, using crossbred beef heifers, populations of as high as 1000 horn flies did not statistically alter the primary blood parameters measured. This leads to the surprising conclusion that physiological changes in the blood constituents due to horn fly feeding may have little effect on host weight gain. A similar study (Presley et al. 1995) presented somewhat conflicting results, with differences in temperature, water consumption, feed intake, packed cell volume, serum cortisol and blood urea nitrogen all significantly different in cattle exposed to levels of horn fly infestation up to 225. Conflicts between designs of these studies include fly numbers and breeding, and it is probable that neither represents the real picture of what is happening on pasture.

Methods of Control.

(1) Insecticides. Current control of horn flies is dependent on the use of conventional insecticides from the organophorous, pyrethroids, carbamates and avermectin groups. These are applied primarily as sprays, dust, pourons, or in insecticidal ear tags. When organophosphate insecticides are used in various formulations as sprays and dusts, they are effective for short periods and require retreatment at 2-3 week intervals, if satisfactory continuous control is to be achieved. Effective control is defined as <50 flies/head in some parts of the country while in others it is considered to be below 250 flies/head. Haufe (1973) indicates that this number may be considerably less than 250/head in the more northern areas. Dust formulations are used in dust bag stations providing a more or less continuous treatment regimen depending on cattle usage. The application of pour-ons for cattle grubs provides short-term indirect control of horn flies. The application of the ivermectin pouron for control of internal parasites also provides up to 35 days control of horn flies. Eprinomectin delivered topically to flies on Holstein calves controlled 94-99% of horn fly adults at four weeks after application of doses of .32 and 0.5 mg/kg. Lysyk and Colwell (1996) found that use of Diazinon and Ivermectin on pastured cattle increased the length of time that horn flies were effectively controlled, but did not increase the level of control beyond 90% when herds under study were not isolated from other cattle. Delivery of Ivermectin via microspheres (Miller et al. 1998) provided up to 10 weeks of „ 98% control of horn fly larvae in manure after a single injection. Moxidectin given in daily doses to cattle gradually increases in the serum over time, at least up to 21 days (Miller et al. 1994). Survival of larval and adult populations of horn flies was adversely affected by manure and blood, respectively, from treated cattle.

Horn flies are relatively easy to control with any of the several registered insecticides. Producers have been applying effective insecticides for 40 years. Fortunately, relatively little insecticide resistance developed (Burns & Wilson 1963) until the introduction of the pyrethroid ear tags. The introduction of the pyrethroid ear tags enabled convenient, inexpensive, long-lasting control of horn flies. The technology was rapidly accepted and widely used. Consequently, in many areas, large percentages of the total horn fly population were exposed to heavy selection pressures over multiple generations. As a result, horn fly resistance to the pyrethroids began to appear, first in isolated populations in the south followed by larger areas and then in the northern states (Sheppard 1983, 1984, Quisenberry et al. 1984, Harvey et al. 1985, Schmidt et al. 1985). Other Diptera have shown cross-resistance (Chadwick et al. 1984) and multiple resistance (Harris et al. 1982), and horn flies are cross-resistant to selected pyrethroids (Sheppard 1984, Schmidt et al. 1985, Weinzierl et al. 1990). Several management strategies have been suggested for other resistant insect pests (Plapp 1979, Georghiou 1980, 1983, Rawlins et at. 1982, Taylor & Georghiou 1982). Roush & Plapp (1982) showed that biotic potential of resistant house flies may be less than susceptible strains; McKenzie & Whitten (1984) found the same characteristic in sheep blow flies. Recently, progress has been made in characterizing the genetics of resistance (Roush et at. 1986, McDonald & Schmidt 1987, McDonald et at. 1987a, 1987b, McDonald & Schmidt l99Oa, l99Ob).

At least 3 bioassay techniques are in use for study of horn fly susceptibility (Lewis & Eddy 1961, Sheppard 1984, Schmidt et al. 1985, Schmidt & Robertson 1986). No single technique has been accepted as a standard. Kunz & Schmidt (1985) updated the occurrence of resistance and identified the areas of the United States in which resistance has been established.

(2) Insect Growth Regulators. Insect growth regulators (IGR's) have been used against the horn fly. Methoprene, a juvenile hormone mimic, is registered for use in mineral mixes. Diflubenzuron, a chitin inhibitor, is also effective in disrupting molting of the immature stages in the manure. Both materials have been successfully used in a bolus formulation. A sustained-release bolus containing diflubenzuron is marketed under the trade name Vigilante, and one containing methoprene is sold as the Inhibitor bolus. Little damage to other arthropods within the manure pat has been demonstrated when animals were treated with methoprene (Blume et al. 1974, Roth 1989), but Loomis et al. (unpubl.) found that for the dung beetle, *Onthophagus gazella*, there was a 32 and 100% mortality of developing larvae in dung treated at 4.0 and 40.0 ppm, respectively. Cook & Gerhardt (1977) found a significant reduction of only 1 nontarget species and pats decomposed after 8 weeks following diflubenzuron treatment. Fincher (1991) demonstrated that adult emergence of *O. gazella* and *S. rubrus* from brood balls made from dung from steers treated with a diflubenzuron bolus was reduced for 7 weeks but not different from untreated controls during week 9-27.

These materials are less useful to individual producers because adults are not killed. Flies can migrate from outside premises in sufficient numbers to maintain damaging adult populations on herds (Kunz et at. 1972). These compounds could, however, be of significant value if used in areawide control programs in which the area was large enough to significantly reduce the effects of migration. In addition, they could provide a valuable tool in the management of resistance (Cilek & Knapp 1991).

(3) Biological Control. Predation and competition by other endemic arthropods for food and space within the manure pat has been shown to be responsible for a 9O% reduction in horn fly production in Texas (Kunz et at. 1972). Hu and Frank (1996) assessed the field mortality of flies in pats exposed to the north-central Florida arthropod community and found that mid-summer mortality in exposed immature flies averaged 71.3%. Some of the competing organisms have been identified (Depner 1968, Thomas & Morgan 1972, Legner 1978, Harris 1981, Butler et al. 1981, Thomas 1981), but the effect of the intraspecific competition of these species has not been determined. Fungicides compatible with horn fly larval development in the laboratory are discussed in Temeyer (1998). Effective and safe doses of ethanol, methanol, amphotericin B, clotimazole, haloprogin, miconazole, nystatin and tolfonate were found. Dimethylsulfoxide and dimethylformamide were incompatible with horn fly larval development. How fungi affect larval development in field situations is unknown.

O. gazella imported from Australia has been successfully established in Texas, California and Georgia; *O. taurus* and *Euniticellus intermedins* are also established in California. Continued foreign exploration may find species better-suited for the more northern area of the country. Dung beetles have been released into Australia with limited effect against the buffalo fly, but numbers of bush flies produced have been significantly reduced. The effect of *O. gazella* in a large part of south Texas has not been fully assessed, but no significant reduction in production of horn flies has been noted. These beetles do a thorough job of destroying the manure, thus greatly reducing the chance of larval horn fly survival.

Bacillus thuringiensis (Bt) has been demonstrated to be effective against horn fly larvae (Gingrich 1984). Various strains have been shown to produce at least 3 different larvicidal toxins. Larval toxins have been related to the presence of beta exotoxin (Gingrich 1984) and to a component of the subsp. *israelensis* and another sporalation-specific protein of subsp. *israelensis* (Temeyer 1984). Use of beta exotoxin, a heat-stable analog of ATP, has been reported for control of sheep bots and other flies in the republics of the former Soviet Union, but its use in western countries is prohibited due to concerns about mammalian toxicity. Crystal proteins, sometimes called delta-endotoxins, are the principle active moiety in most commercial preparations of Bt. These have been cloned into plants and other microbes. They generally exhibit specificity for target insects based on the alkaline or proteolytic conditions for the protoxin activation and the presence of specific membrane-bound receptors in the gut of the susceptible insect. Very little is known about the third type of larvicidal toxin produced by some strains of Bt except that it is different from the beta exotoxin and the crystal protein (Temeyer 1990). Various potential applications can be considered in order to present the larvicidal toxin in ruminant dung, providing the possibility of environmentally friendly fly control. Development of pest resistance to Bt technology could be controlled or delayed through control of pest exposure to the toxins.

(4) Cultural Control. Sanitation and environmental modifications are of little help in controlling horn flies, since horn flies are a range problem. Host resistance to horn flies have long been suspected, but it is only recently that solid experimental work to ferret out the appropriate factors has been begun. Tarn et al. (1994) identified serological markers that differentiated between horn fly resistant and horn fly susceptible cattle. The resistant factors marked by these proteins are not characterized except by molecular weight, but other work by members of the same group (Brown et al. 1994) has clearly identified breed differences in susceptibility to both horn flies and face flies. In fact, Brown et al. (1994) were able to determine that resistance to horn flies in cattle increased with the proportion of Brahman heritage in Angus crossbred cattle. The levels of humoral antibody in horn fly infested cattle could not be correlated to fly density (Baron and Lysyk 1995), possibly because of postulated immunomodulatory compounds in salivary secretions. Steelman et al. (1997) reported that horn fly density is inversely correlated to the number of hairs per cm2 and that the amount of sebum was directly correlated to the number of hairs. In their studies, Brahman and Chianina steers had higher mean hairs per cm2 than Angus, Brahman x Angus, Charolais and Red Poll steers.

(5) Genetic. Limited research has been done with respect to genetic control of horn flies. Adults have been sterilized with CO60 and CS37. The sterile insect technique (SIT) was shown to impact reproduction in wild populations (Eschle et al. 1973, Kunz et al. 1974). Eschle et al (1977) demonstrated that the combination of methoprene in the drinking water of cattle and the simultaneous release of sterile flies was capable of eliminating the native population from a large ranch on the east end of Molokai, Hawaii. The methoprene treatment eliminated reproduction of native flies in the manure without impacting the released sterile flies. Matings of the released sterile flies with the native population resulted in infertile eggs. This is perhaps the most significant demonstration of the potential of an integrated pest management approach against the horn fly. The potential of cross-mating of diapausing (overwinter as pupae) strains with nondiapausing strains should be investigated as a control tool for areas where diapause is necessary for overwintering survival. If diapausing and nondiapausing strains have become sufficiently isolated, then some genetic manipulation may be useful.

(6) Area Control. Horn fly control has been conducted by individual producers with no organized effort aimed at area control. As a result of this uncoordinated effort, large amounts of insecticide have been used over the years with no effects on areawide populations of horn flies. Although insecticides will remain the backbone of horn fly control for the foreseeable future, technology exists that will permit significant reduction in the volume of pesticides used if this pest is attacked with the areawide IPM concept.

Research is needed to determine the size of the area which will need to be treated to provide economic control of the horn fly (i.e., 100 sq.mi. area will provide 80% control to 65% of the area treated). Following this determination and the establishment of economic infestation thresholds, areawide horn fly control programs need to be considered. New techniques available to assist in areawide control include modifications of fly removal traps (Tozer and Sutherst 1996).

Areawide horn fly control should not be undertaken with an eradication objective, but IPM programs should be aimed at managing horn fly populations below established economic levels. An organization will be required to operate these programs, such as a

Co-op or Pest Management district. Possibly, referendums will be required to obtain the necessary participation by the producers within the geographical area to be managed.

Needs for Insect Pest Management.

(1) Economic Injury Level. The datbase on economic thresholds for horn flies, although limited, has improved in recent years. Loomis (1969) reported 0.55 lbs/head per day weight gains for calves protected from horn flies. Also, the horn fly transmits a filarial-worm parasite that causes a dermatitis along the midventral surface of the body (Hibler 1966). This parasite is common in beef cattle from western and southwestern range areas (Lucker 1956, Maddy 1955).

The economics of horn fly infestations have been studied under field conditions (Campbell 1976, Harvey et al. 1979, Haufe 1982, Kunz et al. 1984, Cocke et al. 1989, Hogsette et al. 1991). Calves weaned from cows on which continuous control was provided showed a 12-14 lb per head weight advantage over calves from cows with horn flies (Campbell 1976). More detailed studies under caged conditions have been conducted by Kinzer et al. (1984). In summary, these data show that range cattle incur a 14% weight loss due to horn fly infestations. The average weaning weights of calves on cattle protected from horn flies are approximately 6.3 kg/head greater than that of calves weaned from infested cows. The increased milk production in protected cows is credited with the greater weaning weights in their calves.

(2) Sampling. Currently used sampling methods allow for the estimation of horn flies per head or on a per side basis. This method has been accepted by researchers if the estimated numbers are below 500 per head; infestations above these numbers become more difficult to estimate, and the method has never been tested and evaluated to determine its accuracy. An experienced technician can, however, provide a consistent relative estimate, and his estimates have been accepted as being sufficient when the same person is making the counts throughout the test. An animal estimate is more accurate than making a side estimate X2. Light conditions, the degree of animal irritation, etc., are too variable to make this a reliable estimate. At least 10 animals or 10% of a herd should be examined to establish an average estimate per head for the herd.

(3) Biological-Ecological. Considerable data have been collected in the past 20 years on the biology-ecology of the horn fly. Basic biology-ecology data available are probably adequate to support an IPM program, but some deficiencies exist. We now have a good database on development times for each immature stage, but we do not have data on immature mortality as a function of environment and parasites, predators and competitors. Some data on the diapause of overwintering populations are available for Texas, Mississippi, Missouri, and Arkansas (Hoelscher & Combs 1970, Wright 1970, Blume et al. 1970, Kunz et al. 1970, 1972, 1973, 1976, Thomas et al. 1974, Kunz & Cunningham 1977, Kunz 1980, Kunz & Miller 1985, Thomas & Kunz 1985, 1986, Thomas et al. 1987, Klein & Lancaster 1992). Diapause, induction, development, and emergence of the horn fly are dependent upon temperature and photoperiod. Consequently, the precise time for diapause varies with location (Thomas & Kunz 1986, Thomas et al. 1987). The horn fly passes the winter in diapause as an intrapuparial, pharate adult (Thomas 1985). In Texas, horn flies enter diapause in October and November with peak production of diapausing individuals in November. Spring emergence begins in late February and continues through early May. Only limited data are available on mortalities occurring during diapause.

Factors influencing the dispersal of horn flies have been investigated. Eddy et al. (1962), Hoelscher et al. (1968) and Kinzer & Reeves (1974) defined flight distance and habits of the horn fly. Generally, data show that dispersal occurs prior to the first blood meal. Once the fly has found its host, very little migration from herd to herd occurs (Kunz et al. 1977). Migration from animal to animal within a herd is significant. Extensive research on dispersal at distances up to 400 meters was conducted by Chamberlain (1981, 1982, 1984). Kunz et al. (1983) noted that fly-free zones could not be established within 4-6 miles of existing populations. Byford et al. (1987) concluded that horn flies dispersed at equal rates to cattle within a 1.7 km range. Guillot et al. (1988) defined the physiological age of those flies naturally dispersing in a native population. Marley et al. (1991) studied the temporal, climatic and physiological mediation of dispersal. Research to elucidate migration (lst host) in relation to wind direction and speed, light, temperature, humidity, distance, and direction continues. A small percentage of released flies are known to disperse up to 8-10 miles. We need to know what happens to the 98% that probably move only relatively short distances. The role of alternate hosts (horse, sheep, rodents, etc.) in terms of primary host finding requires definition. Other blood-feeding Diptera are thought to sustain themselves on alternate food sources while in search of a satisfactory host (Magnarelli & Anderson 1981). This is not known for the horn fly.

Not all animals within a herd of cattle will support the same number of flies. Population variations within herds need to be defined. Steelman et al. (1991) and Brown et al. (1992) have shown a relationship between horn fly population and cattle breed and the heritability of horn fly resistance within breeds.

(4) Immunology. The development of immunological technology against biting flies would reduce the reliance on conventional insecticides. No information on the possible immune response of cattle to the bite of horn flies and stable flies exist. Unlike the acquired immune response of cattle to such parasites as ticks and cattle grubs, cattle do not develop a similar response to biting flies. However, it is commonly observed that infestations in a herd are concentrated on certain individuals. The possible mechanisms that result in this concentration may be a behavioral response of the parasites, or it may be mechanisms associated with the host such as immunity or factors that render certain individuals less attractive or less suitable for parasite development. Sorting out the possible mechanisms that result in the concentration of these ectoparasites on certain individuals will require studies on host attractancy and suitability as well as immune response to the bites of these ectoparasites.

(5) Pest-Host Models. Computer simulation can be a valuable aid in the development of more effective and efficient pest management systems. The ability to simulate the responses of a pest population to various control strategies can provide a basis for decisions as to the types of controls to impose and the timing of these for optimal results.

Computer models of varying complexity have been developed to describe the population dynamics of crop and forest pests; however, increasing attention has been devoted to the development of such models for livestock pests. The work by Berry et al. (1973), Wilkerson (1974), Kunz & Cunningham (1977), Miller (1977) and Palmer & Bay (1984) is aimed directly at modeling populations of the horn fly.

These modeling efforts (Miller 1977, Palmer & Bay 1984, Miller 1986) point to the need for quantitating many important relationships within the system for which data is currently unavailable. There is a need for a better understanding of horn fly biology and ecology during the cooler times of the year when the pests are not a severe problem. Entrance into diapause in the fall and emergence from diapause in the spring needs to be described more precisely in terms of environmental factors. There is need for better description of the survival of individual life stages in response to their environment as well as the effect of interspecific competition and predation. Age specific survival and fecundity of adult flies should be more precisely defined over a variety of field conditions. Emigration and immigration patterns as well as success of host location by newly emerged flies should also be investigated.

Despite these gaps in our knowledge, the computer simulation in its current stage of development can be used to evaluate control strategies for long-term effect against the horn fly in an area. The horn fly model developed at the Knipling-Bushland U.S. Livestock Insects Research Laboratory has been extended to enable imposing one or a combination of control techniques on the system. The results of the simulated strategies can then be evaluated in terms of effectiveness and efficiency. Those strategies appearing most promising from the results of the simulation can then be investigated in the field. For example, the model was used very effectively by an advisor group assembled by Merck & Co. for developing horn fly control strategies using their Ivomec® Pour-on formulation.

(6) Natural Products/Biopesticides. There is a national sentiment encouraging a move away from the synthetic pesticides to "natural products or biopesticides." The history of livestock pest control includes a variety of natural products for control ranging from plant extracts such as rotenone, pyrethrins and nicotine to sulfur, arsenic, kerosene, and various oils. However, because of their limited effectiveness, control agents followed a natural progression from the organochlorines, organophosphates, and carbamates to the pyrethroids and avermectins. As various natural products are developed for control of crop pests, they will become available for research against the horn fly.

Research Needs.

(1) Basic Research.

(a) The current method of estimating horn fly numbers both on cattle and in manure droppings should be evaluated for its accuracy.
(b) Migration and dispersal studies need to be continued to elucidate the horn fly

movements within larger control areas and, also, within herds to determine the treatment coverage necessary to achieve economic control levels.
(c) Investigate the diapause phenomenon and determine its role in late season control which affects the overwintering success and the ensuing spring buildup.
(d) Investigate possible adverse effects of orally administered insecticides on beneficial nontarget insects in dung pats, and the breakdown and recycling of dung in different ecosystems. Quantitate the impact of such treatments on the environment/ecosystem.
(e) Step up foreign exploration for natural enemies that also inhabit dung pats.
(f) Further investigate the genetics of resistance and develop a genetic marker to provide early detection of resistance development.
(g) Determine the potential for using immunological techniques for horn fly control.

(2) Control Components Research.
(a) Determine economic injury levels for the various climatic zones representative of the cattle-producing areas. The upper permissible levels of horn fly infestations allowable on cattle need to be defined for long-term IPM programs.
(b) Develop delivery systems for chemical control including sustained-release technology. Effective formulations and devices will greatly reduce the amount of insecticide used and related management production costs and prolong the useful life of available control agents.
(c) Supporting research to develop efficacy, residue, etc., data necessary for the registration of sustained-release formulations and devices.
(d) Develop population management models that will provide theoretical descriptions of the effects of certain control strategies when applied to horn fly populations.
(e) Continue to seek natural products/biopesticides/biorationals (including Bt, plant extracts, and fungi) as control agents for the horn fly.

REFERENCES CITED

Ahrens, E. H. 1977. Horn fly control with an insecticide impregnated ear tag. Southwest. Entomol. 2: 8-10.

Ahrens, E. H. & J. Cocke. 1979. Season long horn fly control with an insecticide impregnated ear tag. J. Econ. Entomol. 72: 215.

Anonymous. 1976. Control of Insects Affecting Livestock. ARS-USDA National Research Program, NRP No. 20480. 99 p.

Anonymous. 1977. Chap. VII, Statistics of Cattle, Hogs, and Sheep, pp. 305-362. *In*: Agricultural Statistics, U.S. Dept. Agric.

Beadles, M. L., A. R. Gingrich & J. A. Miller. 1977. Slow-release devices for livestock insect control: cattle body surfaces contacted by five types of devices. J. Econ. Entomol. 70: 72-75.

Beadles, M. L., J. A. Miller, B. K. Shelley & D. P. Ingenuett. 1979. Comparison of the efficacy of ear tags, leg bands, and tail tags for control of the horn fly *Haematobia irritans* on range cattle. Southwest. Entomol. 4: 70-72.

Berry, I. L., J. A. Miller, C. D. Schmidt & J. L. Eschle. 1973. Simulation of horn fly populations. Trans. ASAE 16: 482-485.

Blume, R. R., A. A. Ago, D. D. Oehler & R. L. Younger. 1974. *Onthophagus gazella*: a non-target arthropod for the evaluation of bovine feces containing methoprene. Environ. Entomol. 3: 947 949.

Blume, R. R., S. E. Kunz, B. F. Hogan & J. J. Matter. 1970.Biological and ecological investigations of horn flies in central Texas: influence of other insects in cattle manure. J. Econ. Entomol. 63: 1121-1123.

Brown, A. H., C. D. Steelman, Z. B. Johnson, C. F. Rosenkrans & T. M. Brasuell. 1992. Estimates of repeatability and heritability of horn fly resistance in beef cattle. J. Anim. Sci. 70: 1375-1381.

Burns, E. C. & B. H. Wilson. 1963. Field resistance of horn flies to the organic phosphate insecticide ronnel. J. Econ. Entomol. 56: 718.

Butler, J. F. & R. L. Escher. 1981.Life cycle: development times of various pupal parasites of house flies and horn flies in Florida. pp. 80-89. *In:* R. S. Patterson, P. G. Koehler, P. B. Morgan, & R. L. Harris (eds.) Status of Biological Control of Filth Flies, Proceedings of a Workshop. ARS, SEA, USDA, US Gov't. Printing Office, New Orleans, LA.

Byford, R. L., A. B. Broce, J. A. Lockwood, S. M. Smith, D. C. Morris & C. P. Bagley. 1987. Horn fly (Diptera: Muscidae) dispersal among cattle herds. J. Econ. Entomol. 80: 421-426.

Campbell, J. B. 1976. Effect of horn fly control on cows as expressed by weaning weights of calves. J. Econ. Entomol. 69: 711-712.

Chadwick, P. R., R. Slatter & M. J. Bowron. 1984. Cross-resistance to pyrethroids and other insecticides in *Aedes aegypti*. Pestic. Sci. 15: 112-120.

Chamberlain, W. F. 1981. Dispersal of horn flies: effect of host proximity. Southwest. Entomol. 6: 316-325.

Chamberlain, W. F. 1982. Dispersal of horn flies II. Wild flies. Southwest. Entomol. 7: 230-234.

Chamberlain, W. F. 1984. Dispersal of horn flies: III. Effect of environmental factors. Southwest. Entomol. 9: 73-78.

Cilek, J. E. & F. W. Knapp. 1991. Diflubenzuron as a possible tool for managing insecticide-resistant horn flies (Diptera: Muscidae). J. Agric. Entomol. 8: 7-16.

Cook, C. W. & R. R. Gerhardt. 1977. Selective mortality of insects in manure from cattle fed Rabon® and Dimilin®. Environ. Entomol. 6: 589-590.

Eddy, G. W., A. R. Roth & F. W. Plapp. 1962. Studies on the flight habits of some marked insects. J. Econ. Entomol. 55: 603-607.

Eschle, J. L., S. E. Kunz, C. D. Schmidt, B. F. Hogan & R. O. Drummond. 1973. Suppression of a population of horn flies with the sterile-male technique. Environ. Entomol. 2: 976-980.

Eschle, J. L., J. A. Miller & C. D. Schmidt. 1977. Insect growth regulator and sterile males for suppression of horn flies (*Haematobia irritans*). Nature 265: 325-326.

Fincher, G. T. 1991. Sustained-release bolus for horn fly (Diptera: Muscidae) control: effects of methoprene and diflubenzuron on some nontarget species. Environ. Entomol. 20: 77-82.

Georghiou, G. P. 1980. Implications of the development of resistance to pesticides: basic principles and consideration of countermeasures to insecticides. *In:* Pest and pesticide management in the Caribbean: proceedings of seminar and workshop, CICP/USAID, 3-7 November 1980/Consort. Internat. Crop Protection. Bridgetown, Barbados, W. I. 2: 116-129.

Georghiou, G. P. 1983. Management of resistance. pp. 769-722. *In* G. P. Georghiou and T. Saito (eds.) Pest Resistance to Pesticides. Plenum Press, NY.

Gingrich, R. E. 1984. Control of the horn fly, *Haematobia irritans,* with *Bacillus thuringiensis*. Comparat. Pathobiol. 7: 47-57.

Guillot, F. S., J. A. Miller & S. E. Kunz. 1988. The physiological age of female horn flies (Diptera: Muscidae) emigrating from a natural population. J. Econ. Entomol. 81: 555-561.

Harris, C. R., S. A. Turnbull & J. W. Whistlecraft. 1982. Multiple resistance shown by field strains of house fly, *Musca domestica* (Diptera: Muscidae), to organochlorine, organophosphorous, carbamate and pyrethroid insecticides. Can. Entomol. 114: 447-454.

Harris, R. L. 1981. The influence of parasites, predators, and competitors on horn fly populations. pp. 125-127. *In:* R. S. Patterson, P. G. Koehler, P. B. Morgan, & R. L. Harris (eds.) Status of Biological Control of Filth Flies, Proceedings of a Workshop. ARS, SEA, USDA, US Gov't. Printing Office, New Orleans, LA.

Harris, R. L., J. A. Miller & E. D. Frazar. 1974. Horn flies and stable flies: feeding activity. Ann. Entomol. Soc. Am. 67: 891-894.

Harvey, T. L., J. R. Brethour & A. B. Broce. 1984. Loss of effectiveness of insecticide ear tags for horn fly (Diptera: Muscidae) control. J. Kansas Entomol. Soc. 57: 715 717.

Haufe, W. O. 1973. Interaction of pesticidal toxicity, parasites, and reversible anticholinesterase activity as stresses on growth in cattle infested with horn flies (*Haematobia irritans*). Toxicol. Appl. Pharmacol. 25: 130-144.

Haufe, W. O. 1982. Growth of range cattle protected from horn flies (*Haematobia irritans*) by ear tags impregnated with fenvalerate. Can. J. Anim. Sci. 62: 567-575.

Hibler, C. P. 1966. Development of "*Stephanofilaria stilesi*" in the horn fly. J. Parasitol. 52: 890-898.

Hoelscher, C. E. & R. L. Combs. 1970. The horn fly II. Comparative physiological studies of reproductive and diapaused pupae. Physiol. Zool. 43: 241-248.

Hoelscher, C. E., R. L. Combs & J. R. Brazzel. 1968. Horn fly dispersal. J. Econ. Entomol. 61: 370-373.

Hogsette, J. A., D. L. Prichard & J. P. Ruff. 1991. Economic effects of horn fly (Diptera: Muscidae) populations on beef cattle exposed to three pesticide treatment regimes. J. Econ. Entomol. 84: 1270-1274.

Kinzer, H. G., W. E. Houghton, J. M. Reeves, S. E. Kunz, J. D. Wallace & N. S. Urquhart. 1984. Influence of horn flies on weight loss in cattle with notes on prevention of loss by insecticide treatment. Southwest. Entomol. 9: 212-217.

Kinzer, H. G. & J. M. Reeves. 1974. Dispersal and host location of the horn fly. Environ. Entomol. 3: 107-111.

Klein, C.D., Jr. & J. L. Lancaster. 1992. Diapause of the horn fly in Arkansas. Southwest. Entomol. 17: 109-113.

Kunz, S. E. 1980. Horn fly, *Haematobia irritans,* production as affected by seasonal changes in rangeland forage conditions. Southwest. Entomol. 5: 80-83.

Kunz, S. E., R. R. Blume, B. F. Hogan & J. J. Matter. 1970. Biological and ecological investigations of horn flies in central Texas: influence of time of manure deposition on oviposition. J. Econ. Entomol. 63: 930-933.

Kunz, S. E. & J. R. Cunningham. 1977. A population prediction equation with notes on the biology of the horn fly, *Haematobia irritans,* in Texas. Southwest. Entomol. 2: 79-87.

Kunz, S. E., J. R. Cunningham & J. L. Eschle. 1973. Horn fly: use of insecticides to disrupt life cycle. J. Econ. Entomol. 66 :1239-1240.

Kunz, S. E., B. F. Hogan & J. L. Eschle. 1976. Some bionomical aspects of horn fly populations in west Texas. Southwest. Entomol. 1: 46-48.

Kunz, S. E., M. R. Graham, B. F. Hogan & J. L. Eschle. 1974. Effect of releases of sterile horn flies into a native population of horn flies. Environ. Entomol. 3: 159 162.

Kunz, S. E., R. L. Harris, B. F. Hogan & J. E. Wright. 1977. Inhibition of development in a field population of horn flies treated with diflubenzuron. J. Econ. Entomol. 70: 298-300.

Kunz, S. E., B. F. Hogan, R. R. Blume & J. L. Eschle. 1972. Some bionomical aspects of horn fly populations in central Texas. Environ. Entomol. 1: 565-568.

Kunz, S. E., H. G. Kinzer & J. A. Miller. 1983. Areawide cattle treatments on populations of horn flies (Diptera: Muscidae). J. Econ. Entomol. 76: 525-528.

Kunz, S. E. & J. A. Miller. 1985. Temperature threshold for the development of diapausing horn flies. Southwest. Entomol. 10: 152-155.

Kunz, S. E., J. A. Miller, P. L. Sims & D. C. Meyerhoeffer. 1984. Economics of controlling horn flies (Diptera: Muscidae) in range cattle management. J. Econ. Entomol. 77: 657-660.

Kunz, S. E. & C. D. Schmidt. 1985. The pyrethroid resistance problem in the horn fly. J. Agric. Entomol. 2: 358-363.

Lewis, L. F. & G. W. Eddy. 1961. Laboratory evaluation of insecticides against the adult horn fly. J. Econ. Entomol. 54: 392-393.

Loomis, E. C., D. C. Cannon, C. W. Timbey & L. L. Dunning. 1969. Dust bags for horn fly control on beef cattle. California Agric. 23: 8-11.

Loomis, E. C., J. A. Anderson & R. Moon. 1979. Unpublished.

Lucker, J. G. 1956. U.S. Dept. Agric. Yr. Bk. pp. 298-300.

Maddy, K. G. 1955. N. AMA Vet. 36: 275-278.

Magnarelli, L. A. & J. F. Anderson. 1981. Sugar feeding by female tabanids (Diptera: Tabanidae) and its relation to gonotrophic activity. J. Med. Entomol. 18: 429-433.

McDonald, P. T., L. R. Hilburn, and S. E. Kunz. 1987a. Genetic similarities among natural populations of the horn fly (Diptera: Muscidae). Ann. Entomol. Soc. Am. 80: 288-292.

McDonald, P. T. & C. D. Schmidt. 1987. Genetics of permethrin resistance in the horn fly (Diptera: Muscidae). J. Econ. Entomol. 80: 433-437.

McDonald, P. T. & C. D. Schmidt. l990a. Differential survival of male and female partially resistant horn flies (Diptera: Muscidae) on steers treated with permethrin. J. Econ. Entomol. 83: 1715-1717.

McDonald P. T. & C. D. Schmidt. l990b. Linkage, expression, and distribution of the pyrethroid resistance gene in the horn fly (Diptera: Muscidae). J. Econ. Entomol. 83: 1723-1732.

McDonald, P. T., C. D. Schmidt, W. F. Fisher & S. E. Kunz. 1987. Survival of permethrin susceptible, resistant, and F1 hybrid strains of *Haematobia irritans* (Diptera: Muscidae) on ear-tagged steers. J. Econ. Entomol. 80: 1218-1222.

McKenzie, J. A. & M. J. Whitten. Estimation of the relative viabilities of insecticide resistance genotypes of the Australian sheep blowfly, *Lucilia cuprina*. 1984. Aust. J. Blot. Sci. 37: 45-52.

Miller, J. A. 1977. A computer simulation of populations of the horn fly, *Haematobia irritans* (L.). Dissertation, Texas A&M Univ. 118 p.

Miller, J. A. 1986. Computer modeling of populations of the horn fly. pp. 33-40. *In:* J. A. Miller (ed.) Modeling and Simulation: Tools for Management of Veterinary Pests. ARS-U.S. Depart. Agric., Res. Serv. Beltsville, MD. ARS-46.

Miller, J. A., M. L. Beadles & R. O. Drummond. 1979. U.S. Pat. No. 4,166,107, Aug. 28, 1979. 4 p.

Miller, J. A. & W. F. Chamberlain. 1989. Azadirachtin as a larvicide against the horn fly, stable fly, and house fly (Diptera: Muscidae). J. Econ. Entomol. 82: 1375-1378.

Miller, J. A., R. O. Drummond & D. D. Oehler. 1983a. Sustained-release implant for delivery of ivermectin for control of livestock pests. pp. 223-236. *In:* Roseman, T. J. & S. Z. Mansdorf (eds.) Control Release Delivery Systems. Marcel-Dekker, New York.

Miller, J. A., F. W. Knapp, R. W. Miller, C. W. Pitts & J. Weintraub. 1986. Diflubenzuron bolus for control of fly larvae. J. Agric. Entomol. 3: 48-55.

Miller, J. A. & D. D. Oehler. 1988. A reservoir neckband system for delivery of organophosphorus insecticides to cattle. J. Contr. Release 8: 73-78.

Miller, J. A., D. D. Oehler & S. E. Kunz. 1983b. The release of pyrethroids from insecticidal ear tags. J. Econ. Entomol. 76: 1335-1340.

Miller, J. A., D. D. Oehler, A. J. Siebenaler & S. E. Kunz. 1986. Effect of ivermectin on survival and fecundity of horn flies and stable flies (Diptera: Muscidae). J. Econ. Entomol. 79: 1564-1569.

Oehler, D. D. & J. A. Miller. 1989. Liquid chromatographic determination of ivermectin in bovine serum. J. Assoc. Off. Anal. Chem. 75: 59-61.

Palmer, W. A. & D. E. Bay. 1984. A computer simulation model for describing the relative abundance of the horn fly, *Haematobia irritans irritans* (L.), under various ecological and pest management regimes. Protect. Ecol. 7: 27-35.

Plapp, F. W., Jr. 1979. T. Kommedahl [ed.] Proc. Symp. IX Internal. Congr. Plant Protect., Washington, DC. 1: 244-249.

Quisenberry, S. S., J. A. Lockwood, R. L. Byford, H. K. Wilson & T. C. Sparks. 1984. Pyrethroid resistance in the horn fly, *Haematobia irritans* (L.) (Diptera: Muscidae). J. Econ. Entomol. 77: 1095-1098.

Rawlins, S. C., D. B. Woodard, J. R. Coppedge & L. Jurd. 1982. Management of an insecticide-resistant house fly (Diptera: Muscidae) population by the strategic use of benzylphenol chemosterilant. J. Econ. Entomol. 75: 728-732.

Roland, N. C. & C. W. Wingo. 1973. Cost and effectiveness of horn fly and face fly control programs in semi-isolated range herds in central Missouri. J. Econ. Entomol. 66: 693-696.

Roth, J. P. 1989. Some effects of methoprene on *Spalangia cameroni*, a parasitoid of horn fly pupae. Southwest. Entomol. 14: 91-96.

Roush, R. T., R. L. Combs, T. C. Randolph. J. McDonald & J. A. Hawkins. 1986. Inheritance and effective dominance of pyrethroid resistance in the horn fly (Diptera: Muscidae). J. Econ. Entomol. 79: 1178-1182.

Roush, R. T. & F. W. Plapp, Jr. 1982. Effects of insecticide resistance on biotic potential of the house fly (Diptera: Muscidae). J. Econ. Entomol. 75: 708-713.

Schmidt, C. D. & S. E. Kunz. 1980a. Testing immature laboratory-reared stable flies and horn flies for susceptibility to insecticides. J. Econ. Entomol. 73: 702-703.

Schmidt, C. D. & S. E. Kunz. 1980b. Fenvalerate and stirofos ear tags for control of horn flies on range cattle. Southwest. Entomol. 4: 202-206.

Schmidt, C. D., S. E. Kunz, H. D. Petersen & J. L. Robertson. 1985. Resistance of horn flies (Diptera: Muscidae) to permethrin and fenvalerate. J. Econ. Entomol. 78: 402 406.

Schmidt, C. D., J. J. Matter & J. H. Meurer. 1977. Efficacy of ronnel and chlorpyrifos in controlling biting flies on horses. Southwest. Entomol. 2: 144-148.

Schmidt, C. D., J. J. Matter, J. H. Meurer, R. E. Reeves & B.K. Shelley. 1976. Evaluation of a synthetic pyrethroid for control of stable flies and horn flies on cattle. J. Econ. Entomol. 69: 484-486.

Schmidt, C. D. & J. L. Robertson. 1986. Effects of treatment technique on response to horn flies (Diptera: Muscidae) to permethrin at different temperatures. J. Econ. Entomol. l3: 684-687.

Sheppard, C. 1983. Stirofos resistance in a population of horn flies. J. Ga. Entomol. Soc. 18: 370-376.

Sheppard, C. 1984. Fenvalerate and flucythrinate resistance in a horn fly population. J. Agric. Entomol. 1: 305-310.

Steelman, C. D., A. H. Brown, E. E. Gbur & G. Tolley. 1991. Interactive response of the horn fly (Diptera: Muscidae) and selected breeds of beef cattle. J. Econ. Entomol. 84: 1275 1282.

Tabashnik, B. E. & B. A. Croft. 1986. Managing resistance: insights through simulation. pp. 7-13. *In:* J. A. Miller (ed.) Modeling and Simulation: Tools for Management of Veterinary Pests. ARS-U.S. Depart. Agric., Res. Serv. Beltsville, MD. ARS-46.

Taylor, C. E. & G. P. Georghiou. 1982. Influence of pesticide persistence in evolution of resistance. Environ. Entomol. 11: 746-750.

Temeyer, K. B. 1984. Larvicidal activity of *Bacillus thuringiensis* subsp. *israelensis* in the dipteran *Haematobia irritans*. Applied and Environmental Microbiology 47: 952-955.

Temeyer, K. B. 1990. pp. 352-356. In Pinnock, D. E. (ed.) 5th Int. Colloq. Invert. Path. Microbial Cont. Proc./Abst. Adelaid, Aust.

Thomas, D. B. 1985. Phenology of intra-pupal metamorphosis in horn fly and stable fly; a note on the diapause stage of the horn fly. Southwest. Entomol. 10: 139-149.

Thomas, D. B. & S. E. Kunz. 1985. Effects of season and density on the fecundity and survival of caged populations of adult horn flies (Diptera: Muscidae). J. Econ. Entomol. 78: 106-109.

Thomas, D. B. & S. E. Kunz. 1986. Diapause survival of overwintering populations of the horn fly, *Haematobia irritans* (Diptera: Muscidae) in south-central Texas. Environ. Entomol. 15: 44-48.

Thomas, G. D. 1981. Insect parasites of the horn fly and face fly in Missouri. pp. 118 124. *In:* R. S. Patterson, P. G. Koehler, P. B. Morgan, & R. L. Harris (eds.) Status of Biological Control of Filth Flies, Proceedings of a Workshop. ARS, SEA, USDA, US Gov't. Printing Office, New Orleans, LA.

Thomas, G. D., I. L. Berry & C. E. Morgan. 1974. Field developmental time of nondiapausing horn flies in Missouri. Environ. Entomol. 3: 151-155.

Thomas, G. D., R. D. Hall & I. L. Berry. 1987. Diapause of the horn fly (Diptera: Muscidae) in the field. Environ. Entomol. 16: 1092-1097.

Weinzierl, R. A., C. D. Schmidt, D. B. Faulkner, G. F. Cmarik, & G. D. Zinn. 1990. Chronology of permethrin resistance in a southern Illinois population of the horn fly (Diptera: Muscidae) during and after selection by pyrethroid use. J. Econ. Entomol. 83: 690-697.

Wilkerson, G. G. 1974. A population model for the horn fly, *Haematobia irritans* (Linnaeus) (Diptera: Muscidae), in north central Florida. M.S. Thesis. Univ. Florida, Gainesville. 111 p.

Williams, R. E., E. J. Westby. K. A. Hendrix & R. P. Lemenager. 1981. Use of insecticide-impregnated ear tags for the control of face flies and horn flies on pastured cattle. J. Anim. Sci. 53: 1159-1165.

Wright, J. E. 1970. Hormones for control of livestock arthropods: development of an assay to select candidate compounds with juvenile hormone activity in the stable fly. Ann. Entomol. Soc. Am. 63: 878-883.

Face Fly, *Musca autumnalis* De Geer

Description and Biology. The face fly, a recent introduction into the North American continent, probably entered the United States from Nova Scotia in 1952 (Vockeroth 1953). Since then its spread has been rapid across the northern tier of states and then southward with the only states free of the face fly today being Texas, Florida, New Mexico and Arizona (Drummond et al. 1988).

Female face flies do not have mouthparts capable of piercing the skin of their hosts, so they are not normally blood feeders, but cause annoyance while feeding on wounds or the moist mucus secretions of the face. Structure of the prestomal teeth of *Musca autumnalis* aid the feeding fly, and these structures allow penetration of the corneal epithelium

(Broce & Elzinga 1984, Annunziata & Broce 1993). Annoying populations of flies can build up under range conditions causing the animals to group together head first in an attempt to keep the flies away. Face fly populations have been associated with increased incidence of pinkeye and other eye disorders. Since the last IPM workshop, Hall (1984) has reviewed literature on the relationship of the face fly to pinkeye. Shugart (1978), in Nebraska, demonstrated that only 1-2 flies can cause the transmission of pinkeye and Shugart et al. (1979) demonstrated that the face fly can damage the eyes of cattle. Glass & Gerhardt (1983, 1984) recovered *Moraxella bovis* from the crops of face flies and Arends et al. (1984) demonstrated transmission of *M. bovis* from blood agar cultures to Hereford cattle; Glass & Gerhardt (1984) showed transmission of *M. bovis* by regurgitation of the pathogen from the crop. Serious, untreated cases can lead to blindness of the animal. Gunn (1993) investigated the contamination of both biting and non-biting flies with bovine viral diarrhoea virus (BVD) and concluded that *M. autumnalis* could carry the virus; the potential role of this fly in disease transmission requires further investigation. Eyeworms in the genus *Thelazia* are also transmitted by face flies (Geden & Stoffolano 1980, 1981, 1982, 1984). Incidence and intensity of *Thelazia* sp. has been investigated both by Geden & Stoffolano (1981) and others for the United States and in Alberta (O'Hara & Kennedy 1989, Kennedy et al. 1990, O'Hara & Kennedy 1991). The short term effects of the eyeworms on cattle require further investigation. Ivermectin has been demonstrated to be effective in controlling *T. skrjabini* in experimentally infected cattle (Kennedy 1992).

Females oviposit on fresh cattle manure; larval development occurs within the pat and pupation occurs in the soil around the pat. We now know that the mortality of face flies in the larval stages is related to pat composition and to competition for the resource. Moisture content in the pat (Bay et al. 1969) and percent of grain in the cattle diet (Meyer et al. 1978) which result in changes in pH, elevated osmolality and production of volatile fatty acids and lactic acid (Grodowitz et al. 1987) affect face fly mortality. How these changes affect mineralization in the fly are not understood. The face fly is one of a small group of flies with a mineralized puparium. The mineralization and composition of this puparium has been studied (Fraenkel & Hsiao 1967, Darlington et al. 1983). The morphology, composition, dissolution and transport of mineral from the Malpighian tubules has also been elucidated (Grodowitz et al. 1987, Krueger et al. 1987, 1988). An enzyme, carbonic anhydrase, known to be important in reversible hydration of CO2 and which may be involved in mineralization has been isolated, purified and characterized from the face fly (Darlington et al. 1984, 1985, Burt et al. 1992). Mortality of immature face flies in the dung pat from biotic factors has been the subject of some research effort both before and since the 1979 IPM Workshop. Parasitism in the field by hymenopterous parasites known to attack face flies apparently plays a minor role in field mortality of immature stages of the fly (Blickle 1961, Thomas & Wingo 1968, Turner et al. 1968, Thomas et al. 1983); however, predation is a major factor (Valiela 1969, Burton & Turner 1970, Thomas et al. 1983). Studies by Moon (1980) to deduce effects of competition in the pat indicated that competition results in stunting of flies, and when competition is heavy, survival to adulthood is low. The relationship of the face fly to the host-specific nematode, *Heterotylenchus autumnalis* has been investigated (Stoffolano 1970, Stoffolano & Nickle 1966, Thomas et al. 1972, Kaya & Moon 1978, Kaya et al.

1979, Krafsur et al. 1983, Chirico 1990) and may have some potential for biological control of the face fly. Peitzmeier et al. (1992) have concluded that a combination of biotic, climatic and environmental factors may reduce face fly populations. Female feeding on faces of cattle affects animal behavior (Dougherty et al. 1993) by increasing the rate of dry matter intake linearly as the number of face flies increases. Continuous overlapping generations occur, and the face flies overwinter as adults in barns, farm houses, under tree bark and other shelters. The behavior of male face flies differs significantly from that of the female. Males spend little time on cattle or feeding on fecal fluids but are anthophilic feeders on a number of flower species and frequent pasture margins, wooded areas at pasture edges and fence rows. Fly populations appear to fluctuate and damaging infestations vary from year to year. It is potentially a national pest with only the southwest escaping its effects up to now.

Economic Importance. The annual losses in control costs and production losses due to face flies have been estimated to be in excess of $52 million (Drummond 1987) and is considered to be a major pest of range cattle in the United States.

Methods of Control.

(1) Insecticides. Chemical control methods are inadequate. Currently utilized control technologies include a number of self-treating devices such as dust bags and oilers of various designs. Feed-through insecticides, sprays and ear tags have had limited, but not effective, use. None of the presently available control technologies are completely effective for face fly control. Some of the registered animal insecticides provide some control if insecticide residue can be maintained on the animal. Research has continued on evaluation of pesticides for face fly control at a number of localities throughout the range of the face fly.

(2) Biological Control. The face fly is infested by the nematode, *H. autumnalis*, which effectively renders the female incapable of oviposition, thus effectively removing her from the reproductive population. Research on the incidence, distribution and effect of the nematode on fly behavior (cited earlier) has been done since the last IPM workshop. Some attempts at rearing and release of the nematode have been made, but much work remains to be done in this area. A predatory parasitic Staphylinid, *Aleochara tristis*, has also been reared for release. Legner (1978) introduced a number of species of Coleoptera into California for the control of the face and horn flies. Among these were 3 *Hister* species, one *Peranus* species and one *Sandalus* species. Campbell & Hermanussen (1974) found that *Philonthus theventi* was easily reared, long-lived and easily established and could function as a biological control if released repeatedly. Kessler & Balsbaugh (1972) in South Dakota and Wingo et al. (1974) in Missouri found that at times *P. cruentatus* was an effective predator. Wingo et al. (1974) also found several species of Carabidae and Hydrophilidae that preyed on face fly larvae and pupae. In spite of these observations and limited attempts at biological control, much work is needed in this area. Similar observations have been reported for a number of hymenopterous parasites but have not been pursued further.

Hower & Cheng (1968) experimented with *Bacillus thuringiensis* as a feed additive and found it effectively reduced face fly development. No recent pathogenic studies area being conducted, but possibly studies being conducted with the horn fly will be useful in face fly control. The introduction of dung beetles which could establish in face fly areas could be beneficial; however, none are presently available.

Peterson & Meyer (1978) have studied male behavior, and these studies could have implications for areawide off-animal control utilizing trapping or insecticide-treated panels at areas frequented by male flies. Additional studies need to be conducted on plant feeding, and attractants from plants to lure male flies.

Face fly pheromones have been isolated and identified (Sonnet et al. l975, Uebel et al. 1975), but further research is needed to implement these into surveillance and/or control programs. Trapping to supplant the current-used face or eye counts as population sampling techniques would be desirable. Both screen and pyrimidal sampling methods have been published (Peterson & Meyer 1978, Pickens et al. 1977), and the pyrimidal traps have been utilized in the field (Easton 1979). Development of an attractive target for face flies would have value, either as a population estimating technique or in some instances as a control method. Spectral responses of face flies have been investigated since the last workshop and have been determined by behavioral (Pickens 1983) and electrophysiological studies (Agee & Patterson 1983). Pickens (1990) used a trichromatic visual system to determine which contrasting colors would be attractive to flies under field conditions. Genetic variability of face fly populations has been examined (Bryant et al. 1983) in relation to a population bottleneck which would have occurred if the face fly entered the United States from Nova Scotia and spread across the country over the ensuing years. Krafsur & Black (1992) used polyacrylamide electrophoresis to resolve enzymes at 50 loci in Iowa flies and surveyed gene diversity at 12 loci among 6 geographically independent lab colonies. These studies demonstrated statistically significant differentiation, probably due to drift after colonization. Polytene chromnosomes of the face fly proved unsuitable for detailed investigations of face fly genetics (Dev & Meyer 1988). Genetic control of face flies may be possible and should be explored further. Mansour & Krafsur (1991) induced dominant lethal mutations in face flies using 1600 rads to produce 97% dominant lethality of sperm in treated males. Irradiation did not affect insemination rates. Eclosion was unaffected, but fecundity and fertility were determined to be inversely proportional to the radiation dose. Breed differences of cattle with respect to face fly density have been investigated by Steelman et al. (1993), and these studies should be pursued.

(3) Insect Growth/Development Regulators (IGR). Dimilin,® an IGR, has been effective when the manure of animals treated with the IGR was bioassayed with face fly larvae. This treatment would need to be continuous and could only be provided at the present time with bolus formulations (Miller et al. 1977). This provided control for up to 16-20 weeks and could play an important role in a large area IPM program. Treatment of individual herds would not be effective as immigrating adults will provide continuous infestation pressures. These compounds could, however, be of significant value if used in areawide control programs in which the area was large enough to significantly reduce the effects of migration. Cook & Gerhardt (1977) found a significant reduction of only one nontarget species and pats decomposed after 8 weeks following Dimilin treatment. In California, however, Loomis et al. (unpublished) found that for the dung beetle *Onthophagous gazella*, there was a 32 and 100% mortality of developing larvae in dung treated at 4.0 and 40.0 ppm, respectively. Methoprene and other growth regulating compounds have some potential for face fly control, and methoprene is known to to be effective in killing larvae of the fly in the pat (Miller & Uebel, 1974). Both ivermectin and Moxidectin® show some potential for face fly control (Webb et al. 1991, Williams & Towell 1992) and would provide an additional group of compounds with a different mode of action from materials currently in use.

Research at Beltsville shows that attractant panels with specific characteristics can be useful in trapping and reducing face fly populations from around dairy or range cattle (Pickens et al. 1977). Also the traps can be treated with insecticide to kill the attracted flies. The number of traps, spacing, timing, etc., require definition, but this could prove to be an important part of an areawide IPM program. A pilot program currently being conducted will provide information regarding the effectiveness and possible use of these panels in IPM programs.

Host resistance could be important, but environmental modifications and husbandry practices will probably not play an important role in controlling face flies. Genetic manipulation of face flies has not been done but should be investigated to determine control possibilities.

Needs for Insect Pest Management.

(1) Economic Injury Level. Recent studies in Nebraska showed that as little as only one fly per eye could set up the condition necessary for the invasion of the causative agent of cattle pinkeye or bovine infectious keratitis (Shugart 1978). Production by cattle affected by pinkeye can be reduced by 25% or more. No definitive data is available on losses due directly to the annoyance of face flies which could result in loss of production.

(2) Sampling. Sampling to determine infestation on cattle is made by visual counts of flies occurring per face on the animals. This only provides an estimate of the number of flies occurring on the host but gives no indication of the total populations in the environment. Studies to correlate face counts to total area population counts (i.e., 5/head = 10% of total population in area) would be useful. Sampling by sticky trap surfaces has been utilized and correlated with age classes of flies caught at various habitat types (Moon et al. 1986) based on the physiological age classes established by Van Geem et al. (1983). Age distributions of flies were biased by source and were most abundant (concentrated) at dung.

(3) Biological - Ecological. Biology-ecology studies specific for many of the areas in which the face fly occurs are sparse. Since the face fly overwinters as an adult, studies to determine how this life stage could be manipulated to reduce the number producing offspring the following season needs to be investigated. We now have significant information on the diapausing behavior of the face fly based on studies both in the laboratory and the field. The rate of fat body hypertrophy is temperature dependent (Read 1984). Stoffolano & Matthysse (1967), Valder et al. (1969), Caldwell & Wright (1978) and Read (1984) have all demonstrated that temperature and day lenght are important in inducing diapause. Evans & Krafsur (1990) showed a relationship between light intensity, photoperiod and diapause induction. The average daily survival rates of parous flies in September was determined by Krafsur et al. (1986) to be >0.89 which is consistent with that of summer flies. There is further evidence that flies destined for diapause do not visit host cattle, since previttellenogenic flies did not show developing *Thelazia* infections (Krafsur & Church 1985). Population trends as governed by environmental and or intrinsic factors need to be further elucidated. Control strategies based on managing overwintering populations at hibernaculia need to be developed and evaluated.

(4) Post-Host Models. The Moon model for face fly development should receive continued support and refinement. The development of an initial model could well define the biological-ecological deficiencies that exist in this area.

The outcome of a pilot test currently being conducted in Maryland by the U.S. Department of Agriculture will help determine the role of IPM in future programs. The use of attractant panels and insecticides is being used in an areawide program to determine their effectiveness. For a successful IPM program, areawide (size of which needs to be determined) programs need to be developed. Control of flies on farms and small premises will not affect the overall population unless producers in an area all participate.

REFERENCES CITED

Agee, H. R. & R. S. Patterson. 1984. Spectral sensitivity of stable, face, and horn flies and behavioral responses of stable flies to visual traps (Diptera: Musicadae). Environ. Entomol. 12: 1823-1828.

Arends, J. J., R. E. Wright, P. B. Barto & K. S. Lusby. 1984. Transmission of *Moraxella bovis* from blood agar cultures to Hereford cattle by face flies (Diptera: Muscidae). J. Econ. Entomol. 77: 394-398.

Bay, D. E. & C. W. Pitts. 1976. Olfactory responses of face flies, *Musca autumnalis,* to bovine feces. Ann. Ent. Soc. Amer. 69: 933-936.

Bay, D. E., C. W. Pitts & G. Ward. 1969. Influence of moisture content of bovine feces on oviposition and development of the face fly. J. Econ. Entomol. 62: 41-44.

Blickle, R. L. 1961. Parasites of the face fly, *Musca autumnalis*, in New Hampshire. J. Econ. Entomol. 54: 802

Broce, A. B. & R. J. Elzinga. 1984. Comparison of prestomal teeth in the face fly (*Musca autumnalis*) and the house fly (*Musca Domestica*) (Diptera: Muscidae). J. Med. Entomol. 21: 82-85.

Bryant, E. H., H. Van Dijk & W. Van Deldon. 1981. Genetic variability of the face fly, *Musca autumnalis de geer*, in relation to a population bottleneck. Evolution. 35: 872-881.

Burt, E., M. V. Darlington, G. Graf & H. J. Meyer. 1992. Isolation, purification and characterization of an insect carbonic anhydrase. Insect Biochem. Molec. Biol. 22: 285-291.

Caldwell, E. T. N. & R. E. Wright. 1978. Induction and termination of diapause in the face fly, *Musca autumnalis*, (Diptera: Muscidae), in the laboratory. Can. Entomol. 110: 617-622.

Campbell, J. B. & J. F. Hermanussen. 1974. *Philonthus heveneti*: Life history and predatory habits against stable fly, house fly, and face fly under laboratory conditions. Environ. Entomol. 3: 356-358.

Chirico, J. 1990. Sex ratio, emergence and infection rates of Heterotylenchus-exposed *Musca autumnalis* (Diptera: Muscidae). J. Parasitol. 76: 124-127.

Cook, C. W. & R. R. Gerhardt. 1978. Selective mortality of insects in manure from cattle fed rabon and dimilin. Environ. Entomol. 6: 589-590.

Darlington, M. V., H. J. Meyer, G. Graf & T. P. Freeman. 1993. The calcified puparium of the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Insect. Physiol. 29: 157 162.

Darlington, M. V., H. J. Meyer & G. Graf. 1984. The localization, purification and partial characterization of carbonic anhydrase in the face fly, *Musca autumnalis*. Ann. N.Y. Acad. Sci. 429: 219-221.

Darlington, M. V., H. J. Meyer & G. Graf. 1985. Carbonic anhydrase in the face fly, *Musca autumnalis*. 1985. Insect Biochem. 15: 411-418.

Dougherty, C. T., F. W. Knapp, P. B. Burrus, D. C. Willis & N. W. Bradley. 1993. Face flies (*Musca autumnalis de geer*) and the behavior of grazing beef cattle. Appl. Anim. Behav. Sci. 35: 313-326.

Drummond, R.O. 1987. Economic Aspects of Ectoparasites of Cattle in North America. pp. 9-24. In Leaning, W. H. D. & J. Guerrero [eds] The economic impact of parasitism in cattle. Merck, Sharp & Dohme Agric. Vet. Dic. Symp. Proc., Vet. Learning Systems.

Drummond, R. O., J. E. George & S. E. Kunz. 1988. Control of screwworms on cattle and other livestock in: "Control of arthropod pests of livestock: a review of technology." CRC Press, Boca Ratom. pp. 63-70.

Easton, E. E. 1979. The value of the pyramid sticky trap for sampling face flies, *Musca autumnalis,* under field conditions in South Dakota. Environ. Entomol. 8: 1161 1164.

Evans, L. D. & E. S. Krafsur. 1990. Light intensity, photoperiod, and diapause induction in face flies (Diptera: Muscidae). Environ. Entomol. 19: 1217-1222.

Fraenkel, G. & C. Hsiao. 1967. Calcification, tanning, and the role of ecdysone in the formation of the puparium of the face fly, *Musca autumnalis*. J. Insect Physiol. 13: 1387-1394.

Geden, C. J. & J. G. Stoffolano. 1980. Bovine thelaziasis in Massachusetts. Cornell Veterinarian. 70: 344-359.

Geden, C. J. & J. G. Stoffolano. 1981. Geographic range and temporal patterns of parasitization of *Musca autumnalis* (Diptera: Muscidae) by *Thelazia sp* (Nematoda: Spirurata) in Massachusetts, with observations on *Musca domestica* (Diptera: Muscidae) as an unsuitable intermediate host. 1981. J. Med. Entomol. 18: 449 456.

Geden, C. J. & J. G. Stoffolano. 1982. Development of the bovine eyeworm, *Thelazia gulosa* (Railliet and Henry), in experimentally infected, female *Musca autumnalis* de Geer. J. Parasitol. 68: 287-292.

Geden, C. J. & J. G. Stoffolano. 1984. Nematode Parasites of Other Dipterans. pp. 849-898. *In* Plant and insect nematodes. W. R. Nickle [ed]. Marcel Dekker, New York.

Gianaspero, A. & A. B. Broce. 1993. Micromorphology of the prestomal teeth and feeding behaviour of *Musca autumnalis, M. larvipara* and *M. osiris* (Diptera: Muscidae). Med. Vet. Entomol. 7: 398-400.

Glass, H. W. & R. R. Gerhardt. 1983. Recovery of *Moraxella bovis* (Handuroy) from the crops of face flies (Diptera: Muscidae) fed on the eyes of cattle with infectious Bovine Keratoconjunctivitis. J. Econ. Entomol. 76: 532-534.

Glass, H. W. & R. R. Gerhardt. 1984. Transmission of *Moraxella bovis* by regurgitation from the crop of the face fly (Diptera: Muscidae). J. Econ. Entomol. 77: 399-401.

Grodowitz, M. J., A. B. Broce & L. H. Harbers. 1987. Characteristics of dung that affect face flies, *Musca autumnalis* (Diptera: Muscidae), larva survival and puparial mineralization. Environ. Entomol. 16: 722-730.

Grodowitz, M. J., A. B. Broce & K. J. Kramer. 1987. Morphology and biochemical composition of mineralized granules from the Malpighian tubules of *Musca autumnalis* de Geer larvae (Diptera: Muscidae). Insect Biochem. 17: 335-345

Gunn, H. M. 1993. Role of fomites and flies in the transmission of bovine viral diarrhoea virus. Vet. Record 132: 584-585.

Hall, R. D. 1984. Relationship of the face fly (Diptera:Muscidae) to pinkeye in cattle: a review and synthesis of the relevant literature. J.Med.Entomol. 21: 361-365.

Hower, A. A. & Tien-Hsei Cheng. 1968. Inhibitive effect of *Bacillus thuringiensis* on the development of the face fly in cow manure. J. Econ. Entomol.61: 26-31.

Johnson, G.D., J. B. Campbell, H. C. Minocha & A. B. Broce. 1991. Ability of Musca autumnalis (Diptera: Muscidae) to acquire and transmit bovine herpesvirus-1. J. Med. Entomol.28: 841- 846.

Kaya, H. K. & R. D. Moon. 1978. The nematode *Heterotylenchus autumnalis* and face fly *Musca autumnalis*: A field study in Northern California. J. Nematol. 10: 333-341.

Kaya, H. K. R. D. Moon & P. L. Witt, 1979. Influence of the nematode, *Heterotylenchus autumnalis*, on the behavior of face fly, *Musca autumnalis*. Environ. Entomol. 8: 537-54.

Kennedy, M. J. 1992. The efficacy of ivermectin against the eyeworm, *Thelazia skrjabini*, in expermintally infected cattle. Vet. Parasitol. 45: 127-131

Kennedy, M.J., D. T. Moraiko & L. Goonewardene. 1990. A study of the prevalence and intensity of occurrence of *Thelazia skrjabini* (Nematoda: Thelazioidea) in cattle in central Alberta, Canada. J. Parasitol.76:196-200.

Kessler, H. & E. U. Balsbaugh, Jr. 1972. Parasites and predators of the face fly in east-central South Dakota (*Musca autumnalis*). J. Econ. Entomol. 65: 1636-1638.

Knapp, F. W. & J. D. Webb. 1992. Use of dichlorvos in a portable automatic sprayer mineral station to control face flies (Diptera: Muscidae) and pyrethroid-resistant horn flies (Diptera: Muscidae) on beef cattle. J. Agric. Entomol. 9: 273-281.

Krafsur, E. S. & W. C. Black, IV. 1992. Analysis of isozyme loci in the face fly, *Musca autumnalis* DeGeer. Biochem. Genet. 30: 625-634.

Krafsur, E. S. C. J. Church, M. K. Elvin & C. M. Ernst. 1983. Epizootiology of *Heterotylenchus autumnalis* (Nematoda) among face flies (Diptera: Muscidae) in central Iowa, USA. J. Med. Entomol. entomology. 20: 318-
324.
Krafsur, E. S. & C. J. Church. 1985. Bovine thelaziasis in Iowa.J. Parasitol. 71: 279 286.

Krafsur, E. S. L. D. Evans & W. C. Black.1986. Autumn phenology of face fly (Diptera: Muscidae) populations in Iowa, USA. J. Med. Entomol. 23: 389-395.

Krueger, R. A., A. B. Broce & T. L. Hopkins.1987. Dissolution of granules in the Malpighian tubules of *Musca autumnalis* DeGeer, during mineralization of the puparum. J. Insect Physiol. 33: 255-263.

Krueger, R.A., A. B. Broce, T. L. Hopkins & K. J. Kramer. 1988. Calcium transport from Malpighian tubules to puparial cuticle of *Musca autumnalis*. J. Comp. Physiol. B 158: 413-419.

Legner, E. F. 1978. Natural enemies Histeridae, Staphylinidae, *Heterotylenchusautumnalis* imported in California for the biological control of face fly, *Musca autumnalis* De Geer, and horn fly, *Haematobia irritans* (L.). *In* Proc. Forty-sixth Annu. Conf. Calif. Mosquito and Vector Control. 197: 77-79.

Loomis, E. C., J. R. Anderson and R. Moon. Unpublished Data

Lysyk, T. J. and E. R. Easton. 1986. Relationships between body size and fecundity in the face fly *Musca autumnalis*. Proc. S. D. Acad. Sci. 64: 171-176.

Mansour, Y. M. & E. S. Krafsur.Induction of dominant lethal mutations for control of the face fly, *Musca autumnalis* DeGeer. Med. Vet. Entomol. 5: 175-181.

Meyer, J. A. C. M. Christensen & F. W. Knapp.1978. The influence of various levels of ground ear corn and alfalfa hay in the bovine diet on the development of the face fly, *Musca autumnalis*. Environ. Entomol.7:829-830.

Miller, J. A., M. L. Beadles, J. S. Palmer & M. O. Pickens. 1977. Methoprene for control of the horn fly (*Haematobia irritans)*: A sustained-release bolus fromulation for cattle. J. Econ. Entomol. 70: 589-591.

Miller, R. W. & E. C. Uebel. 1974. Juvenile hormone mimics as feed additives for control of the face fly and house fly (*Musca autumnalis, Musca domestica)*. J. Econ. Entomol. 67: 69-70.

Moon, R. D. 1980. Effects of larval competition on face fly *Musca autumnalis*. Environ. Entomol. 9: 325-330.

Moon, R. D., K. Schwinghammer, F. W. Knapp, H. J. Meyer, E. T.

Morgan, C. E, G. D. Thomas & R. D. Hall. 1983. Annotated bibliography of the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. Suppl. 4: 1-25.

O'Hara, J. E. & M. J. Kennedy. 1989. Prevalence and intensity of *Thelazia* spp. (Nematoda: Thelazioidea) in a *Musca autumnalis* (Diptera: Muscidae population from central Alberta. J. Parasitol. 75: 803-806.

O'Hara, J. E. & M. J. Kennedy. 1991. Development of the nematode eyeworm, *Thelazia skrjabini* (Namatoda: Thelazioidea), in experimentally infected face flies, *Musca autumnalis* (Diptera: Muscidae. J. Parasitol. 77: 417-425.

Peterson, R. D., II, and H. J. Meyer. 1982. Seasonal activity of male face flies (Diptera: Muscidae) in North Dakota. Environ. Entomol. 11: 884-888.

Peterson, R. D. II, and H. J. Meyer. 1978. Trapping technique for male face flies (*Musca autumnalis*). J. Econ. Entomol. 71: 40-42.

Peitzmeier, B. A., J. B. Campbell, J.B. & G. D. Thomas, 1992. Insect fauna of bovine dung in northeastern Nebraska and their possible effect on the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Kansas Entomol. Soc. 65: 267-274.

Pickens, L. G. 1983. Color responses of the face fly, *Musca autumnalis*. Entomol. Exp.Appl.33: 229-234.

Pickens, L. G. 1990. Colorimetric versus behavioral studies of face fly (Diptera:Muscidae) vision. Environ. Entomol.19: 1242-1252.

Pickens, L G. , R. W. Miller, & J. J. Grasela. 1977. Sticky panels as traps for *Musca autumnalis*. J. Econ. Entomol. 70: 549-552.

Pickens, L. G. & R. W. Miller. 1980. Biology and control of the face fly, *Musca autumnalis* (Diptera: Muscidae) [Pest of cattle]. J. Med. Entomol.17: 195-210.

Read, N. R. 1984. Diapause induction and development in face fly (*Musca autumnalis* De Geer). M.S. Thesis. Univ. Minn., St. Paul 117 p.

Schmidtmann,E. T. & D. R. Berkebile. 1985. Relationship between numbers of face flies (Diptera: Muscuidae) and nearest-neighbor distance among Holstein cows. J. Econ. Entomol. 78:102-105.

Schmidtmann, E.T. & R. D. Hall. 1986. Sampling bias and habitat variation in gonotrophic age of female *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol.23: 269-273.

Shugart,J. 1978. The face fly, *Musca autumnalis* De Geer: Ability to cause mechanical damage and transmit pinkeye pathogens. Ph.D. Dissertation, Univ. Nebr. Lincoln 54 pp.

Shugart,J. I., J. B. Campbell, D. B. Hudson, C. M. Hibbs, R. G. White & D. C. Clanton. 1979. Ability of the face fly to cause damage to eyes of cattle. J. Econ. Entomol. 72:633-635.

Sonnet,P. E., E. C. Uebel & R. W. Miller. 1975. Sex pheromone of the face fly and compounds influencing pheromone activity. Environ. Entomol. 4:761-764.

Steelman,C. D., E. E. Gbur, G. Tolley & A. H. Brown, Jr. 1993. Variation in population density of the face fly, *Musca autumnalis* De Geer, among selected breeds of beef cattle. J. Agric. Entomol. 10:97-106.

Stoffolano,J. G. & W. R. Nickle. 1966. Nematode parasite (*Heterotylenchus* sp.) of face fly in New York state. J. Econ. Entomol. 59:221-222.

Stoffolano,J. G. & J. G. Matthysse. 1967. Influence of photoperiod and temperature on diapause in the face fly, *Musca autumnalis* (Diptera: Muscidae). Ann. Entomol. Soc. Amer. 60:1242-1246.

Stoffalono,J. G. 1970. Parasitism of *Heterotylenchus autumnalis* Nickle (Nematoda: Sphaerulariidae) to the face fly, *Musca autumnalis* De Geer (Diptera: Muscidae). J. Nematol. 2:324-329.

Thomas,G. D., Hall, R. D., Wingo, C. W., Smith, D. B. & C. E. Morgan. 1983. Field mortality studies of the immature stages of the face fly (Diptera: Muscidae) in Missouri. Environ. Entomol. 12:823-830.

Thomas,G. D. & C. W. Wingo. 1968. Parasites of the face fly and two other species of dung-inhabiting flies in Missouri. J. Econ. Entomol. 61:147-152.

Ubel,E. C., Sonnet, P. E., Miller, R. W. & M. Beroza. 1975. Sex pheromone of the face fly, *Musca autumnalis* De Geer (Diptera: Muscidae). J. Chem. Ecol. 1:195-202.

Turner,E. C., Burton, R. P. & R. R. Gerhardt. 1968. Natural parasitism of dung breeding Diptera: A comparison between native host and an introduced host, the face fly. J. Econ. Ent. 61:1012-1015.

Valder,S. M., T. L. Hopkins & S. A. Valder. 1969. Diapause induction and changes in lipid composition in diapausing and reproducing face flies (*Musca autumnalis*). J. Insect Physiol. 15:1199-1214.
Valiela, I. 1969. An experimental study of the mortality factors of larva *Musca autumnalis* De Geer. Ecol. Monograph 39:199-255.

Van Geem, T. A. B. Broce & R. D. Moon. 1983. A system for physiological age grading of female face flies, *Musca autumnalis* De Geer. Ks. St. Univ. Agric. Ept. Stn. Bull. 643. 23 pp.

Vockeroth,J. R. 1953. *Musca autumnalis* De Geer in North America (Diptera: Muscidae). Can. Entomol. 85:422-423.

Webb,J. D., J. G. Burg & F. W. Knapp. 1991. Moxidectin evaluation against *Solenoptes capillatus* (Anoplura: Linognathidae), *Bovicola bovis* (Mallophaga: Trichodectidae), and *Musca autumnalis* (Diptera; Muscidae) on cattle. J. Econ. Entomol. 84:1266-1269.

Williams,R. E. & C. A. Towell. 1992. Comparison of insecticidal ear tags and ivermectin in a topical formulation for controlling horn flies and face flies (Diptera: Muscidae) on pastured cattle. J. Agric. Entomol. 9:283-288.

Wingo, C. W., G. D. Thomas, G. N. Clark & C. E. Morgan. 1974. Succession and abundance of insects in pasture manure: relationship to face fly survival. Ann. Entomol. Soc. Amer. 67:386-390.

Common Cattle Grub, *Hypoderma lineatum* (Villers), and Northern Cattle Grub, *Hypoderma bovis* (L.)

Description and Biology. The common cattle grub, *Hypoderma lineatum* (Villers), and the northern cattle grub, *Hypoderma bovis* (L.) are univoltine dipteran species whose larval forms parasitize cattle. *H. lineatum* is found throughout the United States and Canada and *H. bovis* is found in Canada and the northern 2/3 tier of the U.S. states. The newly-hatched larvae penetrate the skin and presumably migrate via connective tissue fascial planes to the connective tissues of the esophagus and diaphragm where they reside for ca. 8 months. Then they digest a breathing hole in the host's back, molt twice, and undergo further development in the subcutaneous tissues over a period of ca. 2 months in cyst-like furuncles. Warble stage cattle grub larvae can be observed seasonally in the subcutaneous tissues of the backs of cattle for a 3 to 5 month period for any given locality. Third-stage larvae drop from the warbles and pupate on the ground. Adult flies emerge from the puparium and during their brief (1 wk), non-feeding life, mate and oviposit onto the hair of cattle. The characteristic running ("gadding") of cattle in an effort to avoid ovipositing female flies, causes injuries, interrupted grazing, milk loss, and weight loss. Occasionally, edema and inflammation occur at the site of entry of first-stage larvae (Gingrich 1982), and eosinophilic mediastinitis, myositis, pleuritis, and pneumonia associated with migrating first-stage larvae have been reported (Panciera et al. 1993). Larvae in the subcutaneous tissues of the back (warble phase) can reduce weight gains and result in losses at slaughter incurred as a result of meat and carcass devaluation. Devaluation of the hide as a result of breathing-holes is also a significant economic factor.

Economic Importance. The financial impact of cattle grub infestation on the cattle industry was originally estimated at $600 million dollars by Drummond et al (1981). Upon re-evaluation, the estimated losses were revised down to $61 million dollars, largely the result of the intensive use of systemic insecticides and a further evaluation of the effect of cattle grubs on weight gain (Drummond, 1987). Research on cattle grub has continued to receive only limited support by both USDA and SAES since the development of the systemically active organic phosphate insecticides in the late 1950's-early 1960's. Distribution of the 2 species of cattle grubs and the resultant economic damage has remained nearly constant since the early part of the century. Cattle grub infestations are heaviest in the northern Great Plains and mountain areas, moderate in the southern Great Plains, the far West and the Midwest, and light in the eastern seaboard regions. Shipment of cattle to feedlots redistributes the problem, as younger stock bear the heaviest infestations. Both biology and control of cattle grubs have been reviewed by Scholl (1993).

Methods of Control. Organophosphate systemic insecticides have been and still are a mainstay of cattle grub control programs. They are applied to cattle as sprays, pour-ons or spot treatment as single treatment in the early summer to early fall, depending on location, and are still a mainstay of cattle grub control programs. The activity of the avermectins, a group of macrocyclic lactones, against cattle grubs has now been well documented since the last national IPM conference in 1979 (Preston 1984, Benz 1985, Drummond 1985, Scholl 1993). Treatments with both organophosphate systemics and avermectins kill most of the larvae migrating within cattle before appreciable damage occurs. However, organophosphate treatment may cause undesirable side-effects in cattle (when larvae die within sensitive tissues such as the esophagus and spinal canal tissues), unless applied very early in the development of the larvae. Avermectins are also effective against second- and third-stage larvae in warbles and may have an advantage in late season treatment when organophosphates cannot be used (Scholl et al. 1985). Activity of several avermectins has now been confirmed, and other avermectins like Moxidectin (Scholl et al. 1992) and Doramectin can be expected to see use. Unfortunately, the use of effective chemical control on one farm does not protect that farm's cattle from subsequent exposure from flies invading from neighboring untreated farms. Elimination of the cattle grub problem can only occur as a result of area wide control. Legislated programs of area wide control for cattle grubs have existed in Canada, Ireland, England, and Europe. These programs have effectively reduced the number of grub infested cattle to very low numbers (Khan 1977). Since the last national livestock IPM workshop, a joint 5-year United States-Canada pilot sterile cattle grub test, initiated in 1982 (Kunz et al. 1984, Weintraub & Scholl 1984), has been conducted. The biological results of the test were published in 1986 (Scholl 1986) and in 1990 (Kunz et al. 1990). In spite of difficulties in rearing large numbers of insects for production of sterile males, an economic evaluation of the project (Klein et al. 1990) concluded that the program was cost effective. Other chemical control materials (mostly systemic insecticides) which would provide alternative modes of action or would improve the application, safety and environmental acceptability of treatments would still be desirable. Although the insect growth regulator, Methoprene,® has demonstrated some control of cattle grubs when applied in a sustained-release formulation (Barrett et al. 1978), current research on growth regulating materials has been discontinued or terminated.

The acquisition of immunological resistance to cattle grub infestation by cattle repeatedly exposed to the parasite has been clearly documented (Evstafjev, 1980; Gingrich, 1980, 1982). These studies and earlier observations elicited interest in utilizing host resistance to control this parasite. Early attempts at vaccination used crude or partially purified antigen preparations (Magat and Boulard, 1970; Baron and Weintraub, 1981). At approximately the same time considerable interest was developing in the digestive enzymes of Hypoderma spp. (Lecroisey et al, 1979; Tong et al, 1981; Lecroisey et al, 1983; Lecroisey and Keil, 1985). These studies lead to investigations of the immunogenicity and antigenicity of these proteins in the bovine host (Pruett & Barrett, 1984; Pruett et al, 1988; Schwinghammer et al, 1988; Pruett et al, 1990). Also at this time were a number of immunobiological studies (Boulard & Bencharif, 1984; Baron & Weintraub, 1987; Boulard, 1989; Pruett & Temeyer, 1989; Baron, 1990; Fisher et al, 1991; Pruett, 1993; Nicolas-Gaulard et al, 1995; Moire, et al, 1997) and vaccination attempts, with partially purified and purified natural proteins, whose purpose was to search for good vaccine candidates (Pruett & Barrett, 1985; Baron & Weintraub, 1986;

Pruett, et al, 1987; Pruett, et al, 1989; Baron & Colwell, 1991; Chabaudie, et al, 1991; Temeyer, et al, 1993). Development of the methodology and the basic studies necessary for control of cattle grubs by host resistance elicited by vaccination have been the subject of intensive research efforts since the last national livestock IPM workshop (Magat & Boulard 1970, Baron & Weintraub 1987, Pruett et al. 1987, Pruett et al. 1989, Pruett et al. 1990, Baron 1990, Losson & Lonneux 1990, Chabaudie et al. 1991, Fisher et al. 1991, Martinez-Gomez et al. 1991). This intensive research effort has produced a prototype recombinant vaccine (Temeyer, et al, 1993) and the technology is currently licensed by the Alberta/Canada Livestock Trust Inc. for product development and commercialization (Pruett 1999). However, unlike the activity of chemicals, this vaccine does not induce a rapid knockdown of the parasite population and does not protect the individual from parasitism. Development of herd resistance to cattle grubs as a result of repeated experimental exposure, results in a reduction of the cattle grub population within a herd (Pruett and Kunz 1996a). Anti-*Hypoderma* spp. vaccines that are currently available, if used in an area-wide IPM program, should elicit protective immunity in naive cattle and therefore reduce the cattle grub population by eliciting host killing of *H. lineatum* larvae, primarily in the back tissues. Repeated use of the vaccine in an area-wide control program would be expected to reduce the parasite population to subeconomic levels (Pruett 1999).

Biological control methods have not been investigated to any extent since the 1979 IPM workshop. Most of the natural enemies, such as predators (rodents, birds, ants) that attack pupae or newly emerged adults are not likely candidates for systematic exploitation. Scholl (1990) has summarized the state of the knowledge of parasites, pathogens and predators of cattle grubs. Investigations on the mortality factors for both *H. bovis* and *H. lineatum* would appear to be warranted. Physiological approaches to control include nutrition of the host (vitamin A-deficient animals were more susceptible than normal to infestation by invading newly hatched larvae) (Gingrich & Barrett 1975).

Serological testing procedures that allow the monitoring of cattle grub populations have been developed (Sinclair & Wassal 1983, Colwell & Baron 1990). (Sinclair et al (1990) used an ELISA test to determine the incidence of H. bovis in England and Wales). Use of a serological test for incidence of *H. bovis* has been reported for England and Wales (Sinclair et al. 1990). Lysyk et al. (1991) used an ELISA method outlined by Colwell & Baron (1990) for determining the proportion of uninfested cattle (that allowed the construction of a model) to estimate cattle grub abundance in 30 herds in Canada and the United States.

Sustained, area wide control procedures directed toward *Hypoderma* spp. population control utilizing either chemical, immunological, or biological integrated control technologies are needed. Perhaps, producersknowledgeable on the economic and animal welfare advantages to successful parasite control, could be motivated into voluntary cooperation in regional programs of area-wide management. An area wide program would require good organization, knowledge of cattle grub ecology and systems to monitor the grub populations in the designated area. However, before embarking on such programs, the issue of whether the goal is to eradicate or to control (at a subeconomic level) must be defined.

The goal of most area wide control efforts would have to be defined to be either eradication or control (at a subeconomic level). Eradication of the pest populations by treating cattle with systemic insecticides (Khan 1977), has not been attained in North America and elsewhere (Ireland), despite population reductions to extremely low levels (Drummond et al. 1978) and may be unattainable (Rich 1965). The initial research in Canada, which demonstrated that releases of sterile male *H. lineatum* integrated with a chemical control regimen could effectively eliminate the remaining population of cattle grubs (Weintraub 1977), was reinforced by the studies conducted jointly between the U.S. and Canada (Scholl et al. 1986, Klein et al. 1990, Kunz et al. 1990).Control technologies to include chemical, immunological, and biological are available for use in programs of integrated control. However, the integration of these methodologies remains as a need. These control technologies are available for use in the area-wide control of cattle grubs, if their introduction has the favor of producer and societal-sponsored programs of sustained development.Producer education and acceptance is an obtainable goal.

Needs for Insect Pest Management.

(1) Economic Injury Level. Perhaps the economic data thus far developed (Drummond et al. 1978) is sufficient to support an IPM program. The most convincing data show that cattle grub control prevents significant economic losses at the packing plant (Rich 1970, Klein 1977). Rich (1970) concluded that organized continuous control is inherently profitable even at definable low level infestations. Klein (1977) concluded that the individual producer's economic requirements were enough to stimulate grub control by individual producers, and Klein et al. (1990) demonstrated that an IPM program should be economically justifiable. Further research is needed on the effect of cattle grubs on host physiology to provide an adequate incentive to instigate and control cattle grubs in an IPM program. For an adequate incentive to control cattle grubs in an IPM program, further research is needed to determine losses by the producer, e.g., effect on weight gains, milk flow. This information in turn may require basic research into the effects of cattle grubs on the nutrition, health and physiology of the host. Economic grub population thresholds have not been defined, and grub populations rebound to heavy infestations if controls are relaxed (Graham & Hourrigan 1977). Therefore, the only IPM program for cattle grubs may be one that integrates all existing control strategies and does not rely mainly on the sterile insect technique (SIT). Over emphasis of the sterile male technique as a solution to the control of *Hypoderma* spp. might at this point be unsound, considering the difficulty in rearing this insect. The dogmatic reliance and prolonged anticipation of the SIT to solve this problem and the continued acceptance of eradication, rather than population management, as the only satisfactory goal of control of the cattle grub has perhaps only prolonged the development and implementation of IPM strategies with technologies that we currently possess and know to be efficacious. In fact, according to Scholl (1993) cattle grub populations are depressed in the U.S.; si,ilarly, Drummond (1987) states that their economic damage has declined. Thus, while the debate over IPM techniques continues without directed IPM intervention, both authors agreed that the decline in population and damage is due to increased use of systemic insecticides (perhaps primarily as a result of the increased use of avermectins as an anthelminthic, thus fortuitously killing cattle grubs as a secondary parasite). If this level of effectiveness can be achieved passively without directed intervention, one must consider that the results of an area-wide intensive IPM approach in a specific problem area (Scholl et al, 1986) would be quite feasible. An economic comparison of the IPM approach was published based on the joint U.S. and Canadian study (Klein et al. 1990). The results of this study would indicate that the IPM approach is economically feasible. Highest net benefits would be achieved with chemical control alone; however, net benefits would also be very high where sterile insect releases were used in conjunction with insecticidal control.

(2) Sampling. Sampling to determine infestation levels is well established. The entire grub population is in bovine hosts for extensive periods of time and available for sampling by palpation or squeezing out the grubs. Periodic charting of grub positions in the backs will determine total grub populations or counting the grubs at times of peak numbers will be sufficient for comparative assessment. However, refined statistical treatment of sampling data is needed for IPM purposes. Recently, a model for estimating the abundance of cattle grubs based on proportion of uninfested cattle as determined by serology has been published (Lysyk et al. 1991). This research should be pursued.

(3) Biological - Ecological. Biological-ecological research has been mainly on the phenological development of grub infestations to determine timing of pesticide application (Pfadt 1952, Scharff 1950, Simco & Lancaster 1964, Weintraub & Howell 1964, Collins et al. 1969, Pfadt et al. 1975). The reproductive behavior and flight range of heel flies are important to an IPM program and have received some research attention (Gregson 1958, Weintraub 1961, Weintraub et al. 1968). Mating aggregation sites have been defined (Catts et al. 1965); radiation dosages for sexual sterility have been determined (Drummond 1963). Procedures for enhancing survival of larvae of cattle grubs in rearing media have been investigated with the goal of increasing survival for research (Chamberlain 1964, Chamberlain 1989, Chamberlain & Scholl 1991). Studies of cattle grub biology have been conducted to enhance the reliability of vaccine bioassay assessment (Pruett & Kunz, 1996b) and to provide rearing techniques for reliable supplies of biological material for challenge experiments (Pruett & Kunz, 1996c; Pruett & Kunz, 1997). These studies are needed to continue effective assessment of control strategies. Other such data (Weintraub, unpublished) have been generated for colonizing cattle grubs in cattle hosts and managing them for IPM requirements. Further basic and developmental research is required in these areas as well as in biochemical-physiological areas to develop more elements of an IPM program. The research into effective use of vaccines should continue as a priority.

(4) Pest-Host Models. Models of grub populations, or their elements, exist. The data in mortality table and other ecological-behavioral studies constitute a model highlighting natural mechanisms that regulate grub populations (Weintraub 1977). The incorporation of host resistance/immunity as a variable in these models should enhance their utilization.

Models of natural infestations based on population frequency distribution have also been elaborated (Breev 1968, 1976). The model for estimating abundance based on the serological determination of uninfested cattle (Lysyk et al. 1991) should be tested in various geographic localities to refine the model and test it against control strategies, or in order to produce alternative models.

REFERENCES CITED

Ashizawa, H., Y. Taguchi, K. Nagao, D. Nosaka, S. Tateyama. 1976. Pathological findings of warble swellings caused by *Hypoderma* larvae in the bovine skin. Bull. Fac. Agric. Univ. Miyazaki 23: 87-95.

Baron, R. W. 1990. Cleavage of purified bovine complement component C3 in larval *Hypoderma lineatum* (Diptera: Oestridae) hypodermins. J. Med. Entomol. 27: 899-904.

Baron, R. W. & J. Weintraub. 1981. Immunization of cattle against warbles. Research Highlights 1981, Research Station, Lethbridge, Alberta, Canada, pp. 24 25.

Baron, R. W. & J. Weintraub 1986. Immunization of cattle against hypodermatosis *Hypoderma lineatum*, (DeVill) and *H. bovis* (L.) using H. lineatum antigens. Vet. Parasitol. 21: 43-50.

Baron, R. W., & J. Weintraub. 1987. Lymphocyte responsiveness in cattle previously infested and uninfested with *Hypoderma lineatum* (de Vill.) And *H. bovis* (L.) (Diptera: Oestridae). Vet. Parasitol. 24: 285-296.

Baron, R. W. & D. D. Colwell. 1991. Enhanced resistance to cattle grub infestation (*Hypoderma lineatum* de Vill.) In calves immunized with purified hypodermin A, B and C plus monophosphoryl lipid A (MPL). Vet. Parasitol. 38: 185-197.

Barrett, C. C., J. A. Miller, R. O. Drummond, & M. O. Pickens. 1978. Effect of methoprene on eclosion of the common cattle grub and the northern cattle grub. Southwest. Entomol. 3: 323-326 (232-236).

Benz, G. W. 1985. Animal health applications of ivermectin. Southwest. Entomol. Suppl 7: 43-50. 7: 2-10 (Suppl.)

Boulard, C., & R. Garronne. 1978. Characterization of a collagenolytic enzyme from larvae of *Hypoderma lineatum* (Insecta: Diptera, Oestriform). Comp. Biochem. Physiol. 59B: 251-255.

Boulard, C., & J. Weintraub. 1973. Immunological responses of rabbits artificially infested with the cattle grubs *Hypoderma bovis* (L.) and *H. lineatum* (De Vill.) (Diptera: Oestridae). Int. J. Parasitol. 3: 379-386.

Boulard, C. & F, Bencharif. 1984. Changes in the haemolytic activity of bovine serum complement by *Hypoderma lineatum* (insect oestridae) larval proteinases naive and immune cattle. Parasite Immunol. 6: 459-467.

Boulard, C. 1989. Degradation of bovine C3 by serine proteases from parasites *Hypoderma lineatum* (Diptera, Oestridae). Vet. Immunol. and Immunopath. 20: 387-398.

Breev, K. A. 1968. On the distribution of warble-fly larvae (*Hypoderma bovis* [L.] and *H. lineatum* [Vill.]) in herds of cattle. I. Negative binomial distribution as a model of the distribution of warble-fly larvae. Parazitologiya 2: 322-333.

Breev, K. A. & J. Minar. 1976. On the regularities of distribution of *Hypoderma bovis* De Geer larvae parasitizing cattle herds in different parts of the range of this warble fly. Folia Parasitol. 23: 343-356.

Catts, E. P., R. Garcia, & J. H. Poorbaugh. 1965. Aggregation sites of males of the common cattle grub, *Hypoderma lineatum* (De Villers) (Diptera: Oestridae). J. Med. Entomol. 1: 357-358.

Chabaudie, N., C. Villejoubert, & C. Boulard. 1991. The response of cattle vaccinated with hypodermin A to a natural infestation of *Hypoderma bovis* and *Hypoderma lineatum*. Intern. J. Parasitol. 21: 859-862.

Chamberlain, W. F. 1964. Survival of first-instar common cattle grubs under anaerobic conditions *in vitro*. Ann. Ent. Soc. Amer. 57: 799-800.

Chamberlain, W. F. 1989. Survival of second and third-instars of the common cattle grub, *Hypoderma lineatum* (Villers) and *Hypoderma bovis* (L.) In artificial media. Southwest. Entomol. 14: 233-239.

Chamberlain, W. F., & P. J. Scholl. 1991. New procedure to enhance survival of third instar *Hypoderma lineatum* (Villers) (Diptera: Oestridae) in artificial media. J. Med. Entomol. 28: 266-269.

Collins, R. C., L. W. Dewhirst, & L. A. Carruth. 1969. The cattle grub problem in Arizona. I. Timing of adult activity of the common cattle grub. J. Econ. Entomol. 62: 652-656.

Colwell, D. D., & R. W. Baron. 1990. Early detection of cattle grubs (*Hypoderma lineatum* and *H. bovis*) (Diptera: Oestridae) using ELISA. Med. Vet. Entomol. 4: 35-42.

Drummond, R. O. 1963. Int. J. Radiation Biol. 7: 491-495.

Drummond, R. O. 1985. Effectiveness of ivermectin for control of arthropod pests of livestock. Southwest. Entomol. 7: 34-42 (Suppl.)

Drummond, R. O., J. E. George, & S. E. Kunz. 1987. Control of cattle grubs in cattle. pp. 77-91. In Control of Arthropod Pests of Livestock: A Review of Technology. CRC Press, Boca Raton. 245 pp.

Drummond, R. O., G. Lambert, H. E. Smalley Jr., & C. E. Terrill. 1981. Estimated losses of livestock to pests. pp. 111-127. In CRC Handbook of Pest Management in Agriculture Vol. 1 [ed.] Pimentel, D. CRC Press, Boca Raton. 597 pp.

Drummond, R. O., R. A. Bram, & N. Konnerup. 1978. Animal Pests and World Food Production. Chap. 5, pp. 63-93. In Pimentel, D. World Food, Pest Losses, and the Environment. AAAS Selected Symposium 13, Boulder CO, Westview Press.

Evstafjev, M. N. 1980. The role of immunity during hypodermatosis of cattle (English summary). Parazitologiya 14: 197-205.

Fisher, W. F., J. H. Pruett, V. M. Howard, & P. J. Scholl. 1991. Antigen-specific lymphocyte proliferative responses in vaccinated and *Hypoderma lineatum* infested calves. Vet. Parasitol. 40: 135 145.

Gingrich, R. E., & C. C. Barrett. 1975. Effect of dietary vitamin A on the innate resistance of cattle to infestation by larvae of *Hypoderma lineatum* (Diptera: Oestridae). J. Med. Ent. 12: 13-15.

Gingrich, R. E. 1980. Differentiation of resistance in cattle to larval *Hypoderma lineatum*. Vet. Parasitol. 7: 243-254.

Gingrich, R. E. 1982. Acquired resistance to *Hypoderma lineatum*: comparative immune response of resistant and susceptible cattle. Vet. Parasitol. 9: 233-242.

Graham, O. H., & J. L. Hourrigan. 1977. Eradication programs for the arthropod parasites of livestock. J. Med. Ent. 13: 629-658.

Gregson, J. D. 1958. Recent cattle grub life-history studies a Kamloops, British Columbia, and Lethbridge, Alberta. Proc. Xth Int. Congress Entomol. 3: 725-734.

Khan, M. A. 1977. Vet. Parasitol. 3: 203-276. Khan, M. A. 1977. The feasibility of exterminating warble flies (*Hypoderma spp*.) On a regional basis. Vet. Parasitol. 3:217-223.

Khan, M. A., J. Weintraub, & G. C. R. Croome. 1977. Progress in extermination of cattle grubs (*Hypoderma* spp.): Summary of the symposium. Vet. Parasitol. 3: 271-274.

Klein, K. K. 1977. A Report to the Western Comm. on Livestock Pests, Winnipeg, Manitoba.

Klein, K. K., C. S. Fleming, D. D. Colwell, & P. J. Scholl. 1990. Economic analysis of an integrated approach to cattle grub (*Hypoderma spp.*) Control. Can. J. Agric Econ. 38: 159-173.

Kunz, S. E., R. O. Drummond, & J. Weintraub. 1984. A pilot test to study the use of the sterile insect technique for eradication of cattle grubs. Prev. Vet. Med. 2: 523-527.

Kunz, S. E., P. J. Scholl, D. D. Colwell, & J. Weintraub. 1990. Use of the sterile insect technique for control of cattle grubs (*Hypoderma lineatum* and *H. bovis*) (Diptera: Oestridae). J. Med. Entomol. 27: 523 529.

Lecroisey, A., C. Boulard, and B. Keil. 1979. Chemical and enzymatic characterization of the collagenase from the insect *Hypoderma lineatum*. Eur. J. Biochem. 101: 385-393.

Lecroisey, A., N. T. Tong, and B. Keil. 1983. Hypodermin B, trypsin related enzyme from the insect *Hypoderma lineatum*. Eur. J. Biochem. 134: 261-267.

Lecroisey, A. & B. Keil. 1985. Specificity of the collagenase from the insect *Hypoderma lineatum*. Eur. J. Biochem. 152: 123-130.

Losson, B., & J. F. Lonneux. 1991. Ann. Med. Vet. 135: 373-375.

Lysyk, T. J., D. D. Colwell, & R. W. Baron. 1991. A model for estimating abundance of cattle grub (Diptera: Oestridae) from the proportion of uninfested cattle as determined by serology. Med. Vet. Entomol. 5: 253-258.

Magat, A., & C. Boulard. 1970. Essais de vaccination contre l'hypodermose bovine avec un vaccin contenant une collagenase brute extraite des larves de ler stade d'*Hypoderma lineatum*. Comptes Rend. de l'Acad. Sci., Paris 270: 728-730.

Martinez-Gomez, F., A. Gasca-Arroyo, F. De Juan, & S. Hernandez-Rodriguez. 1991. Maternal anti-*Hypoderma* antibodies in calves. Med. Vet. Entomol. 5: 381-383.

Moire, N., I. Nicolas-Gaulard, Y. Le Vern, & C. Boulard. 1997. Enzymatic effect of hypodermin A, a parasite protease, on bovine lymphocyte membrane antigens. Parasite Immunol. 19: 21-27.

Mrozik, H., & 10 authors. 1979. Amer. Chem. Soc./Chem. Soc. Japan, Chem. Congr., Honolulu, HI, Med. Chem. Abst. #50.

Nicolas-Gaulard, I., N. Moire, & C. Boulard. 1995. Effect of the parasite enzyme, hypodermin A, on bovine lymphocyte proliferation and interleukin-2 production via the prostaglandin pathway. Immunol. 84: 160-165.

Nelson, W. A. N., & J. Weintraub. 1972. *Hypoderma lineatum* (De Vill.) (Diptera: Oestridae): Invasion of the bovine skin by newly hatched larvae. J. Parasitol. 58: 614-624.

Panciera, R. J., S. A. Ewing, E. M. Johnson, B. J. Johnson, & D. L. Whitenack. 1993. Eosinophilic mediastinitis, myositis, pleuritis, and pneumonia of cattle associated with migration of first-instar larvae of *Hypoderma lineatum*. J. Vet. Diagn. Invest. 5: 226-231.

Pfadt, R. E. 1952. Seasonal development of cattle grubs in Wyoming. J. Kans. Entomol. Soc. 25: 137-143.

Pfadt, R. E., J. E. Lloyd, & G. Sharafi. 1975. Pupal development of cattle grubs at constant and alternating temperatures. J. Econ. Entomol. 68: 325-328.

Preston, J. M. 1984. The avermectins: new molecules for use in warble fly control. pp. 17-20 In Boulard, C. & H. Thornberry [eds.] Warble Fly Control in Europe. Rotterdam/Boston: Balkema. 156 pp.

Prieto, M., A. Espi, J. A. Garcia Paloma, & F. Parra. 1990. Effect of ivermectin treatment on anti-hypodermin C titers of Asturiana cattle naturally infested with *Hypoderma lineatum*. Vet. Parasitol. 35: 211-218.

Pruett, J. H. & C. C. Barrett. 1984. Induction of intradermal skin reactions in the bovine by fractionated proteins of *Hypodermin lineatum*. Vet. Parasitol. 16: 137 146.

Pruett, J. H. & C. C. Barrett. 1985. Kinetic development of humoral anti-Hypoderma lineatum antibody activity in the serum of vaccinated and infested cattle. Southwest. Entomol. 10(1): 39-48.

Pruett, J. H., C. C. Barrett, & W. F. Fisher. 1987. Kinetic development of serum antibody to purified *Hypoderma lineatum* proteins in vaccinated and nonvaccinated cattle. Southwest. Entomol. 12: 79-88.

Pruett, J. H., K. B. Temeyer, & B. K. Burkett. 1988. Antigenicity and immunogenicity of *Hypoderma lineatum* soluble proteins in the bovine host. Vet. Parasitol. 29: 53 63.

Pruett, J. H., W. F. Fisher, & K. B. Temeyer. 1989. Evaluation of purified proteins of *Hypoderma lineatum* as candidate immunogens for a vaccine against bovine hypodermiasis. Southwest. Entomol. 14: 363-373.

Pruett, J. H. & K. B. Temeyer. 1989. Colostral transfer of antibodies specific for *Hypoderma lineatum* proteins. Vet. Res. Commun. 13: 213-223.

Pruett, J. H., P. J. Scholl, & K. B. Temeyer. 1990. Shared epitopes between the soluble proteins of *Hypoderma lineatum* and *H. bovis* first instars. J. Parasitol. 76: 881-888.

Pruett, J. H. 1993. Proteolytic cleavage of bovine IgG by hypodermin A, a serine protease of *Hypoderma lineatum* (Diptera: Oestridae). J. Parasitol. 79(6): 829 833.

Pruett, J. H. & S. E. Kunz. 1996a. Development of resistance to *Hypoderma lineatum* (Diptera: Oestridae) within a cattle herd. J. Med. Entomol. 33(1): 49-52.

Pruett, J. H. & S. E. Kunz. 1996b. Warble stage development of third instars of *Hypoderma lineatum* (Diptera: Oestridae). J. Med. Entomol. 33(2): 220-223.

Pruett, J. H. & S. E. Kunz. 1996c. Thermal requirements for *Hypoderma lineatum* (Diptera: Oestridae) egg development. J. Med. Entomol. 33(6): 976-978.

Pruett, J. H. & S. E. Kunz. 1997. A technique for the collection and rearing late-stage 3rd-instars of *Hypoderma lineatum* (Diptera: Oestridae). Southwest. Entomol. 22(2): 201-206.

Pruett, J. H. 1999. Immunological control of arthropod ectoparasites - a review. Int. J. Parasitol. 29: 25-32.

Rich, G. B. 1965. Systemic treatments for control of cattle grubs *Hypoderma spp*. In an isolated range herd. Can. J. Anim. Sci. 45: 165-172.

Rich, G. B. 1970. The economics of systemic insecticide treatment for reduction of slaughter trim loss caused by cattle grubs, *Hypoderma spp*. Can. J. Anim. Sci. 50: 301-310.

Robertson, R. H. 1966. Thesis, Queen Univ., Kingston, Ont., Canada.

Scharff, D. K. 1950. Cattle grubs: their biologies, their distribution, and experiments in their control. Montana Agr. Exp. Sta. Bull. 471.

Scholl, P. J., J. Weintraub, & M. A. Khan. 1985. Late-season treatment of cattle for control of warble grubs. pp. 29-30. In Research Highlights-1984 [ed.] L. J. Sears & E. E. Swierstra.

Scholl, P. J., D. D. Colwell, J. Weintraub, & S. E. Kunz. 1986. Area-wide systemic insecticide treatment for control of cattle grubs, *Hypoderma spp*. (Diptera: Oestridae) two approaches. J. Econ. Entomol. 79: 1558-1563.

Scholl, P. J. 1990. A review of parasites, pathogens and predators of cattle grubs. Southwest. Entomol. 15: 360-365.

Scholl, P. J., F. S. Guillot, & G. T. Wang. 1992. Moxidectin: systemic activity against common cattle grubs (*Hypoderma lineatum*) (Diptera: Oestridae) and trichostrongyle nematodes in cattle. Vet. Parasitol. 41: 203-209.

Scholl, P. J. 1993. Biology and control of cattle grubs. Annu. Rev. Entomol. 39: 53-70.

Schwinghammer, K. A., J. H. Pruett, & K. B. Temeyer. 1988. Biochemical and immunochemical properties of HPLC peak 2, an ion-exchange fraction of common cattle grub (Diptera: Oestridae). J. Econ. Entomol. 81(2): 549-554.

Simco, J. S., & J. L. Lancaster, Jr. 1964. Effects of soil type, moisture level, and temperature on larval and pupal stages of the common cattle grub, *Hypoderma lineatum*. J. Kansas Entomol. Soc. 37: 11-20.

Sinclair, I. J., & D. A. Wassal. 1983. Enzyme-linked immunosorbent assay for the detection of antibodies to *Hypoderma bovis* in cattle. Research in Vet. Sci. 34: 251-252.

Sinclair, I. J., D. W. Tarry, D. A. Wassal, & A. D. Hayward. 1990. Vet. Record 126: 327 329.

Temeyer, K. B., J. H. Pruett, I. Kuhn, & J. Files. 1993. Recombinant vaccine development: A novel approach to ectoparasite control. - A Review of development of a recombinant vaccine for hypodermosis based on hypodermin A. SAAS Bull. Biochem. & Biotech. 6: 31-35.

Tong, N. T., J. M. Imhoff, A. Lecroisey, and B. Keil. 1981. Hypodermin A, a trypsin-like neutral proteinase from the insect *Hypoderma lineatum*. Biochem. Biophys. Acta. 658: 209-219.

Weintraub, J. 1961. Inducing mating and oviposition of the warble flies *Hypoderma bovis* (L.) and *H. lineatum* (De Vill.) (Diptera: Oestridae) in captivity. Can. Entomol. 93: 149-156.

Weintraub, J. 1977. Pilot test of sterile insect releases for warble fly control. Research Highlights, Lethbridge Research Station, 1977: 48-50.

Weintraub, J., & D. E. Howell. 1964. Development of cattle grubs in Oklahoma cattle imported into Canada. J. Econ. Entomol. 57: 494-500.

Weintraub, J., W. S. McGregor, & H. M. Brundrett. 1961. Artificial infestations of the northern cattle grub, *Hypoderma bovis*, in Texas. J. Econ. Entomol. 54: 84-87.

Weintraub, J., R. H. Robertson, & R. H. Gooding. 1968. Experiments in hybridizing the warble flies *Hypoderma lineatum* (de Villers) and *H. bovis* (Linnaeus) (Diptera: Oestridae). Can. J. Zool. 46: 129-133.

Weintraub, J., & P. J. Scholl. 1984. The joint Canada-U.S.A. pilot project on warble grub control. In Research Highlights-1983, pp. 30-32. Lethbridge, Alta.: Agric. Can. Res. Stn.

STABLE FLY

Description and Biology. Stable flies have only recently been considered pests of range cattle, except where rainfall is 30 inches or more, or where small herds are fed during the winter in farmstead corrals and pastured in the summer close to the farmstead where the cattle return for water, salt, or shelter. Stable flies develop in decaying matter mixed with manure, soil, and moisture. If winter feeding areas are not cleaned thoroughly in the spring, these areas may turn into excellent stable fly larval habitats. Stable flies feed on cattle as they come to water and may follow or seek out cattle in pastures. Under these conditions, flies must return to the larval habitats for oviposition. The possibility that the presence of stable fly adults in pastures is due to their dispersal from distant sources is also being considered.

For additional discussion on the biology of this fly, please refer to the Dairy Cattle and Confined Cattle Section of this document.

Economic Importance: Until recently, the effects of stable flies on range cattle have been unknown. Studies by Cutkomp and Harvey (1958) and Cheng (1958) did not differentiate between species of biting flies, whereas Campbell et al. (1977, 1987) dealt only with feedlot cattle. Observations by numerous workers are that range or pastured cattle bothered by stable flies will bunch, expend energy fighting the flies, and fail to graze normally.

Stable flies have long been known as a serious pest of confined cattle at dairies (Bruce & Decker 1958) and feedlots (Campbell et al. 1987). However, in the past two decades, the stable fly has also become a notable pest of range cattle, at least in the Midwest USA. Hall et al. (1982) noted high numbers of stable flies attacking pasture cattle in Missouri, something they had not observed previously. Entomologists have also reported stable fly problems on grazing cattle in North and South Dakota, Wyoming, Colorado, Nebraska, and Kansas.

Cattle react to stable fly attack by bunching, with each animal attempting to protect its front legs (the favorite feeding site of stable flies). Foot stomping, tail switching, heads thrown down toward the front legs, and standing in any available water are all behavioral changes exhibited by cattle under stable fly attack in confined as well as pasture conditions. In addition, pasture cattle attacked by stable flies are seen lying with their legs tucked beneath their bodies. Nebraska ranchers indicate that bunching behavior results in tramping out forage, which on fragile soils may create blowouts. They have also indicated that bunching of the cows causes injury to the calves when they get stepped on and that there is a higher incidence of footrot. In addition, heat stress increases because cattle that are bunched can't dissipate the heat. Feedlot cattle under heat stress reduce their feed intake. This may also be true for range and pasture cattle, in which case they would fail to graze properly. Nebraska studies with grazing steers, where some of the animals were protected from stable flies by sprayed insecticides, indicated these flies reduced average daily gains by a mean of _ lb/day in 84-day trials (Campbell, unpublished). Nebraska ranchers have estimated that yearling weight gain losses are 40-50 lb/animal with a reduction in weaning weights of 20-25 lbs per calf. This is close to the losses observed in the research trials.

Methods of Control

Control of stable flies on pasture or range cattle is difficult, if not impossible. Research is needed to find the source of these flies. Hall et al. (1982) found considerable stable fly larvae development at the edge of the big round hay bales if the hay was wet and had remained in the field for any length of time. However, there is no evidence that these round bales are a significant source of stable flies in western Nebraska (Campbell, unpublished) and central Kansas (Broce, unpublished), probably because of drier conditions. In addition, it doesn't seem likely that all of the stable flies in pastures are coming from feedlots and dairies because these units generally practice good sanitation and manure management practices, and apply insecticides if stable flies are numerous and affect the confined cattle.

There may be numerous stable fly larval habitats that heretofore have gone unnoticed. Some possibilities include winter feeding grounds where the forage (hay) is fed on the ground and considerable residue remains (cattle wastes as much as 40% of the hay fed in tubs or on the ground). Some forage may be spread on trails when it is difficult to get vehicles through. Some cattle are fed from portable racks and hay gets trampled and remains uneaten. All of these may develop into ideal larval habitats. Hay is often stored in stack yards located near the winter quarters for the cattle. Spilled hay and parts of stacks and bales may not be fed in the winter and remain during the spring and summer where, if wet, the residue may be ideal for stable fly development. Wet vegetative areas around ponds, streams or areas where tanks have overflowed may also become stable fly larval habitats.

During a study in Central Kansas in 1991-92, Marquez (1992) monitored stable fly populations in confined livestock operations and in pastures some of which were relatively close by and some of which were >3.5 km away. In addition to monitoring stable fly numbers, female flies were physiologically and chronologically age-graded. The numbers of stable flies in both types of operations fluctuated dependently through time. Likewise, the physiological and chronological age profiles of flies in both types of operations fluctuated dependently on each other through time, showing they had common bionomical features. These results suggest that dispersal from major larval habitats in neighboring (in the tens of km) confined operations could account for the observed dependency. An alternative and plausible explanation is that populations in pastures are produced locally and temporarily-synchronized with those in confined operations. However, the failure to identify larval habitats in pastures makes the latter possibility doubtful. Another possibility might be distant sources from which flies disperse in connection with synoptic weather fronts. Broce & Hogsette (unpublished) have gathered evidence for the dispersal of stable flies into Kansas with the masses of warm air preceding cold fronts in the spring.

All of the possible sources of stable flies in range environments discussed above are only speculative. Intense research efforts are needed to identify these sources of stable flies infesting range and pasture cattle as currently there is no effective means of controlling them. Wet sprays on the legs of the cattle can provide some temporary relief, but these are washed off by wet vegetation; thus a spray is seldom effective for longer than a week (Campbell & Hermanussen 1971). Dust bags, oilers and ear tags are ineffective because they fail to treat the legs of cattle. Feed additives are also ineffective because stable flies do not appear to use fresh manure as larval habitat (Broce & Haas In Press). The lack of control of stable flies with insecticides doesn't appear to be because of the development of resistant stable fly populations, as is the case with horn flies. Although Cilek & Greene (1994) found a few stable fly populations in Western Kansas to have some levels of resistance, the magnitude and prevalence of this resistance was not alarmingly high. In addition, Marcon et al. (1997) bioassayed stable flies from southeastern Nebraska against three chemical groups used for stable fly control and found that flies are still susceptible to all three types of insecticides.

References Cited

Berry, I.L., K.W. Foerster & J.B. Campbell. 1978. Overwintering behavior of stable flies in manure mounds. Environ. Entomol. 7:67-72.

Broce, A.B. & M.S. Haas. Relation of cattle manure age to colonization by stable fly and house fly (Diptera: Muscidae). J. Kans. Entomol. (In Press).

Bruce, N.W. & G.C. Decker. 1958. The relationship of stable fly abundance to milk production in dairy cattle. J. Econ. Entomol. 51:269-274

Campbell, J.B., R.G. White, R. Croosshank & D.C. Clanton. 1977. Effects of stable flies on weight gains and feed efficiency of calves on growing or finishing rations. J. Econ. Entomol. 70:59-594.

Campbell, J.B., I.L. Berry, D.J. Boxler, R.L. Davis, D.C. Clanton & G.H. Deutscher. 1987. Effects ofstable flies (Siptera: Muscidae) on weight gain and feed efficiency of feedlot cattle. J. Econ. Entomol. 80:117-119.

Campbell, J.B. & J.F. Hermanussen. 1971. Efficacy of insecticides and methods od insecticidal application for control of stable flies in Nebraska. J. Econ. Entomol. 54:1189-1190.

Cheng, T.H. 1958. The effect of biting fly control on weight gain of beef cattle. J. Econ. Entomol. 51:278-281.

Cutkomp, L.K. & T.L. Harvey. 1958. The weight responses of beef cattle in relationto control of horn and stable flies. J. Econ. Entomol. 51:72-75.

Hall, R.D., G.D. Thomas & C.E. Morgan. 1982. Stable flies, *Stomoxys calcitrans* (L.), breeding in large round hay bales: Initial associations (Diptera: Muscidae). J. Kans. Entomol. 55:617-620.

Marcon, P., G.D. Thomas, B.D. Siegfried & J.B. Campbell. 1997. Susceptibility of stable flies (Diptera: Muscidae) from southeastern Nebraska beef cattle feedlots to selected insecticides and comparison of 3 bioassay techniques. J. Econ. Entomol. 90:293-298.

Marquez, J.G. 1992. Dynamics, and chronological and physiological age profiles, of stable fly, *Stomoxys calcitrans* (L.), populations in confined and pastured livestock operations. M.S. thesis. Kans. Sta. Univ. 91p.

Mosquitoes

Description and Biology. The 3,500 mosquito species distributed worldwide are characterized as small-bodied flies (3-9 mm in length) with assorted patterns of scales on the body, legs and wings. Each adult possesses an elongate proboscis, and, unlike other common dipterans, the wing have scales which are located only along the wing veins. Mosquito eggs are dark colored and elongate. Permanent water mosquito species deposit eggs singularly on the water surface (i.e., *Anopheles*) or in raft-like formations (i.e., *Culex*). The eggs often hatch within 1-3 days depending on temperatures.

The other general group, known as the floodwater mosquitoes, contains the largest genus, *Aedes*, and other genera such as *Psorophora*. The females of this group select moist substrates for oviposition that are susceptible to periodic inundation (e.g., marshes, rice fields). Egg incubation may last a few days; however, the egg will not hatch until inundated, that may not occur for several days, weeks, months, or longer.

The other immature forms (i.e., larvae and pupae) of the mosquito life cycle are exclusively associated with water and are morphologically distinguishable among the species, with a few exceptions. In this life stage, immatures pose no threat to the health and well being of humans, livestock, wildlife or companion animals. The immatures feed mostly on microorganisms in the water and also serve as a food source for fish and many aquatic insects (including some predacious mosquito larvae).

It is the pestiferous nature and the pathogen-carrying capability of the adult female that serve as threats to humans and animals. These females further impact human and animal populations by their flight capabilities, aggressive blood-seeking instincts and high reproductivity rates. Adult dispersal distances of up to 30-40 miles from their larval habitats are not uncommon for some of the stronger flying species (e.g., *Aedes sollicitans*, the tan salt marsh mosquito).

With a few exceptions, the females are haematophagous on a wide range of animals with most *Culex* species preferring birds, and *Aedes*, *Anopheles* and *Psorophora* species generally preferring large mammals. Female mosquitoes seek a blood host every 3-5 days to support subsequent egg development. The female mosquito is somewhat glutinous in her blood feeding activity, i.e., consuming up to 120% of unfed body weight in blood. Once replete, females usually require 60-70 hrs to digest a bloodmeal (Lehane 1991).

Mosquitoes that use livestock as a blood source generally begin their daily feeding pattern about sunset and continue throughout the night until dawn with the more intensive activity occurring about 2-3 hrs following sunset. However, there are some common species that will feed aggressively during the day also if disturbed (e.g., *Psorophora columbiae* known as the dark rice field mosquito and *Ae. sollicitans*).

Economic Importance. Mosquitoes represent more of an aggressive, blood-seeking pest to U.S. cattle than a primary vector of bovine pathogens. When considering equines, however, mosquitoes can dramatically impact the health of horses. Mosquitoes are directly associated with the transmission of several equine encephalitis viral types. In any case, blood-feeding activities of mosquitoes should keep animal health officials alert to the fact that mosquitoes may be marginally important vectors of insect-borne pathogens affecting bovines in the U.S.

The total U.S. cattle population in 1993 was about 101 million head. This amounted to an increase of one percent over the 1992 estimated population (Drain & Bierstadt 1993). Texas ranked first among the 50 states with 14.3 million followed by Nebraska (5.90 millon) and Kansas (5.89 million).

Byford et al. (1991) recently stated that horn flies were the primary ectoparasite of cattle. However several authors have estimated economic losses to livestock by mosquitoes. Steelman (1976) estimated annual losses to livestock production caused by mosquitoes at $25 million. Drummond (1981, 1987) and Scholl et al. (1985) raised that amount to $38-39 million. The most recent estimate place the economic losses caused by mosquitoes at $50 million (Kunz et al. 1991). For the most part, subsequent estimated economic losses on cattle production, due to mosquitoes, were largely inflationary adjustments based on Steelman's field studies during the mid 1970s.

Agricultural communities are receptive to mosquito abatement activities if it can be demonstrated that such a tax-supported service is worthwhile to the community as a whole and to the agricultural producer. A scientific survey of an agricultural/riceland community (pop. 11,000) in Arkansas was conducted by Farmer et al. (1989) to determine satisfaction, economic benefits and general support for a community-wide, tax-supported, mosquito abatement program. Results indicated that a very high level of satisfaction existed among the citizens of the community, and the cost/benefit ratio far exceeded the levied tax to support the program. The main economic benefit was that for every one dollar spent on the program, 3.4 dollars were generated in return (e.g., increased retail sales of outdoor recreational equipment and supplies). Education and income of the citizens surveyed were positively associated with their level of support of the mosquito abatement program, whereas advanced age of the respondents (i.e., retired, fixed income citizens) was inversely related to the level of support.

Methods of Control.

(1) Insecticides. The 450 organized mosquito abatement districts (MAD) throughout the U.S. remain the primary centers for professional mosquito management activities.

Although the heavier populated districts have their responsibilities directed toward urban and suburban areas, a substantial number of the districts have extensive rural and agricultural territories. Consequently these MADs use chemical pesticides and pest management activities in rural and agricultural lands to prevent the subsequent emergence and dispersal of mosquito adults into nearby cities and communities.

The vast majority of tax-based, county or multi-county MADs use an integrated approach to abatement activities. Biorational agents (e.g., bacteria, fish and insect growth regulators) continue to compose greater proportions of MADs' budget and use in the field. Currently, however, chemical larvicides/adulticides remain substantial components of their programs. It is the aerial and ground ULV pesticide applications over these farms and ranch lands that pastured livestock receive the greatest relief from pestiferous mosquito populations.

<u>(a) Adulticides.</u> As the chlorinated hydrocarbon pesticides were replaced by organophosphate pesticides, so are the organophosphates being replaced by pyrethroids, insect growth regulators (IGR), and environmentally compatible biorational agents in organized mosquito abatement districts across the United States. Malathion and naled continue to be used as mosquito adulticides; however, their role in management of mosquito populations in the U.S. has diminished substantially in the last several years due, in part, to the availability of pyrethroids. Permethrin and resmethrin currently predominate the mosquito adulticiding market in the United States relative to pyrethroids. Other adulticides, such as lambda cyhalothrin, await registration for mosquito control and subsequent release to the mosquito control market.

Effective mosquito control was obtained on cattle herds in Louisiana after area-wide ULV applications by parish-operated mosquito program (Steelman & Schilling 1977). More recent studies by Focks et al. (1991) in Louisiana suggested that biweekly treatments of cattle with a pyrethroid reduced *Psorophora* adult populations in nearby areas. Field data used to drive a simulated model indicated too that an 86% reduction of adult populations could be achieved based on biweekly pyrethroid applications.

<u>(b) Larvicides.</u> Larvicides will be discussed here in the context of those chemical compounds that have been synthesized in the laboratory and not biologically generated within an organism (i.e., bacterial toxin). Methoprene, an IGR, is used extensively in professionally operated MADs. Fenoxycarb is an IGR candidate also but has not been released commercially to abatement personnel. Other chemical pesticides such a temephos, chlorpyrifos and certain surfactants are also major components in a given MAD larviciding program. All are effective against those mosquito species that feed profusely on cattle.

(2) Biological Control. *Bacillus thuringiensis* var. *israelensis* (B.t.i.) has been the primary biorational workhorse in progressive mosquito control activities in the United States since it was registered in 1980. Although debate among scientists have yet to gain full acceptance for either side as to whether B.t.i. is a true biological control agent or a mere source of a produced toxin that degenerates the alimentary canals of mosquito larvae after the bacteria have been ingested. For purposes of discussion B.t.i. will be considered as a biorational agent. B.t.i. has gained considerable acceptance in abatement programs throughout the world and, particularly, in the United States. B.t.i. is an effective larvicide, specific to mosquito larvae, easily applied, relatively safe to animals and available in several adaptable formulations. Applications of B.t.i. are most efficacious when directed toward early instar larvae.

In recent years, a fungal agent, *Lagenidium giganteum*, has been intensely evaluated in the laboratory and in California riceland field plots against mosquito larvae (Kerwin & Washino 1988). Results demonstrated a substantial efficacy against mosquito larvae. Based on these data, the U.S. Environmental Protection Agency issued a registration for use about 2 years ago. However, it has yet to be formally commercialized and made available to mosquito control agencies. Mosquito control managers recognize it to be a beneficial component to their programs due to its efficacy and, most importantly, its high potential for recycling in nature. This latter factor lessens the need for retreatment thereby reducing product and labor costs to the agency. Unfortunately, this characteristic also tends to suppress repetitious sales of the product and, understandably, inhibits substantial investments by profit-oriented businesses to make it available to mosquito control personnel. The future of this agent in operational mosquito control programs currently remains uncertain or for at least a time until industry can expect a reasonable return on its investment.

There is a wide range of other candidate biorational agents that have been investigated as mosquito larvicides. According to Beier & Craig (1985) gregarine parasites are not suitable for significantly reducing natural populations of mosquito larvae. *Coelomomyces* fungi will not be used in mosquito control programs because more bionomical and efficacy data are needed (Lucarotti & Federici 1985).

In summary, the availability of B.t.i. and its comparable low cost will prevent many other biorational agents (e.g., mermithid parasites) from being use because of excessive product costs including labor to application costs (Finney-Crawley 1985). Early reports by Lacey & Singer (1982) indicated that *Bacillus sphaericus* was comparable to B.t.i. (H-14) in controlling *Culex* mosquitoes. Currently *B. sphaericus* is not available to front line mosquito control operations. As pointed out by Mulla (1985), *B. sphaericus* will be another valuable tool for controlling a variety of mosquito larvae once it is marketed.

(3) Genetic. As noted in the 1979 livestock IPM workshop, genetic manipulation of selected mosquito species has shown merit in controlled laboratory conditions. However, limitations of its application to a few mosquito species and the failure of field-oriented projects to successful carry through with plans (due, in part, to funding and political difficulties) are primary reasons this technique has not gained substantial acceptance (Grover 1985, Sharma 1985).

(4) Source Reduction. MADs across the United States use one or more source reduction methodologies in their daily activities (e.g., ditching, flushing, impoundments) to suppress mosquito larval production. In agricultural areas, disruption of mosquito larval habitats by means of ditching and draining, land leveling to promote uniform flooding and draining patterns of irrigated croplands and subsequent adherence to managed irrigation schedules are types of source reduction that are adaptable to agricultural lands. Unfortunately, the cost factor prohibits many MADs from implementing or coordinating such procedures with agricultural producers on a large scale basis.

(5) "Trap-Crop" Techniques. This technique first gained notoriety regarding its use in agricultural croplands. The use of pesticide-treated cattle herds to attract and expose blood-seeking mosquitoes to the toxicant has been successfully demonstrated recently in reducing adult mosquito populations proximal to the herds. (Focks et al. 1991). The procedure is costly based on the labor intensive nature of the biweekly herding of cattle and subsequent pesticide applications. It may not been fully accepted by farmers for those reasons to permit a significant reduction of pest mosquito populations.

(6) Other Methods. Host resistance to blood-seeking female mosquitoes is a viable consideration in cattle production, particularly the more tolerant Brahman breed. Rotation of cattle herds from a pasture has been shown to severely reduce *Psorophora* egg populations in the vacated pasture. At the same time the *Psorophora* egg population in the new pasture is markedly increased following the arrival of the bloodmeal source, i.e., cattle (Meek & Olson 1977). McLaughlin & Vidrine (1987) observed the same relationship with *Psorophora* larval density in Louisiana ricelands.

Needs for Insect Pest Management.

(1) Economic Injury Level. According to Wright (1985) the interim period since the Steelman (1976) economic study, no definitive economic threshold experiment has been published. Currently, the lack of ample research funding is the primary reason why updated, substantive, economic injury data are not available regarding mosquito attacks on cattle.

(2) Sampling. Sampling equipment (e.g., 400 ml dippers for mosquito larvae and light traps for adults) and surveillance programs (i.e., landing rate counts, census of standardized resting sites by adults in daytime shelters and light trap samplings) in MADs for mosquito immatures and adults have remained relatively unchanged. These operational procedures are very adaptable for monitoring the variability and intensity of mosquito populations associated with cattle herds.

(3) Biological-Ecological. There are adequate field studies on mosquito bionomics to at least initiate an IPM program for controlling mosquito adults on pastured cattle. However, critical gaps do exist in the total spectrum of mosquito bionomics for most of the United States (e.g., adult dispersal, natural frequency of blood feeding and adult longevity of certain pest species). Natural mortality rates have been determined for *Ps. columbiae* larvae in riceland habitats (Andis & Meek 1985). Results indicated that about 2% of the *Psorophora* eggs deposited matured to emergent adults. Weathersbee & Meisch (1990) have documented *Anopheles quadrimaculatus* dispersal behavior in Arkansas ricelands. Studies by McAllister & Meek (1991) indicated that initial short range (<1 mile) dispersal of *Ps. columbiae* adults from their former larval habitats demonstrated no predominant directional orientation.

(4) Pest-Host Models. Reliable computer models have been developed for certain *Culex* and *Anopheles* species (Weidhaas 1974). Focks et al. (1988a, 1988b) and Focks & McLaughlin (1988) developed and tested a *Ps. columbiae* / riceland model addressing population predictions. However, reliable models are lacking for certain pest *Aedes* and *Psorophora* species and their influence on cattle production. These models are needed to assist in forecasting mosquito population events using life tables of pest and vector mosquito species, their blood host selection behavior, flexible pest management strategies and selected biotic and abiotic parameters.

Research Needs.

(1) Basic Research.

(a) MADs have ample documentation relating to the species of pest mosquitoes that use cattle as blood hosts. There are significant regions of the United States where organized mosquito control does not exist, and, therefore, surveys need to be conducted to determine the most common species of mosquitoes adversely affecting cattle in these geographical regions.
(b) In addition to identifying the species composition of the mosquito populations feeding on cattle, it is vital that the seasonal occurrence and relative abundance of those populations be known.
(c) It is imperative also that primary larval habitats be identified and periodically monitored. Selected bionomical factors should be investigated (i.e., availability of secondary blood hosts, adult dispersal capabilities and diurnal blood feeding patterns).
(d) A coordinated economic injury study among USDA and SAES researchers is needed to identify pest/vector mosquito species that use cattle as the primary blood source in various climatic zones and regions of the United States, and to evaluate at least those bionomical factors stated immediately above in Item (1) (c).

(2) Control Components Research. A continuum of efficacy tests should be maintained to field-evaluate currently labelled chemical adulticides and larvicides and biorational control agents (i.e., bacteria and IGRs) including innovative application methodologies. This work is required, in part, to detect the development of tolerance or resistance to the compounds by any or all life stages of mosquitoes. Secondly, efforts are needed to evaluate promising candidate pesticides, new formulations of labelled pesticides, biorational agents and enhancement of natural biotic or abiotic factors that serve to suppress mosquito populations.

Some of the larger budgeted MADs should employ the newer advances regarding remote sensing and/or computer-generated geographic informational system technologies. Primarily, these technologies can be used for identifying and defining the extent of mosquito larval habitats over wide areas to include the delineation of other bionomical information on mosquitoes.

These technologies can be immediately implemented in the larger MADs due to the impending federal legislation (i.e., Endangered Species Act) which will directly impact the current chemical application strategies of MADs--particularly, when habitats (i.e., open terrain or pastures) are jointly occupied by mosquito larvae and certified or proposed endangered species. Concurrent habitat use may result in elimination of chemical pesticide applications.

Research Needs.

(1) Basic Research. Three primary factors relating to this category are: (a) determine the species composition, relative abundance and seasonal variability of suspected primary pest/vector mosquitoes of cattle; (b) determine the factors affecting dispersal and host-seeking of the primary species; and (c) conduct these surveys in the various geographical regions of the United States for comparative studies.

(2) Control Components Research. Based on initial estimates of injury thresholds by mosquitoes, and refined subsequently, efforts should be continuously made to implement new biorational agents and their new formulations to include efficacious chemical pesticides, innovative formulations and updated application techniques to improve efficiency and safety of application.

Most citizens living within an area associated with a professionally operated MAD are not aware of the immense records kept of budget expenditures, mosquito surveillance activities, spray equipment calibration, pesticide and biorational application rates and locations and source reduction efforts. There data have not been fully utilized in providing a basis to implement and subsequently evaluate a wide area economic study to determine the cost/benefit ratio to livestock producers.

Not since the frequently referenced work of Steelman (1976) has there been a substantive, long-term field trial assessing the economic injury levels of mosquito feeding on cattle herds. A multi-regional project is needed to document the impact of mosquito feeding on the health and productivity of livestock and subsequently determine cost/benefit ratios of organized mosquito abatement efforts on selected cattle breeds. This project should be a joint venture involving personnel and appropriate funding among USDA-ARS and SAESs, mosquito research personnel, industry, MADs and farmers/ ranchers.

Applicable extension personnel should have a good working knowledge of the current commercial products, surveillance techniques and mosquito management strategies of MADs. They should routinely transfer notable events and other information of MADs to the public through appropriate public health-related printed and voice media. Additionally, extension specialists need to recognize and publicize the allied benefits of MADs to the agricultural community regarding the cost/benefits of protecting livestock to improve weight gains, marketability, etc. and improve the production efficiency of outdoor agricultural workers.

REFERENCES CITED

Andis, M. D. & C. L. Meek. 1985. J. Am. Mosq. Control Assoc. 1: 357-362.

Beier, J. C. & G. B. Craig, Jr. 1985. pp. 167-184. In Laird, M. & J. W. Miles [eds.] V2. Academic Press, London. 369 pp.

Byford, R. L., M. E. Craig & B. L. Crosby. 1992. J. Anim. Sci. 70: 597-602.CSRS/USDA, 1991. Inv. Agricul. Res. FY 91.

Cooperative State Research Service. U.S. Department of Agriculture. Washington, D. C. p. 2.

Drain, C. & D. Bierstadt. 1993. TASS, Aug. 1, 1993 issue, Austin, TX. p. 9.

Drummond, R. O. 1987. In Leaning, W. H. D. & J. Guerrero [eds.] Proc. MSD-AGVET Symp. XXIII World Vet. Congr., Aug. 1987, Montreal, PQ, Canada.

Farmer, F. L., J. M. Redfern, M. V. Meisch & A. Inman. 1989. J. Am. Mosq. Control Assoc. 5: 335-338.

Finney-Crawley, J. R. 1985. pp. 287-303. In Laird, M. & J. W. Miles [eds.] Vol. 2. Academic Press, London. 369 pp.

Focks, D. A. & R. E. McLaughlin. 1988. J. Am. Mosq. Control Assoc. 4: 399-413.

Focks, D. A., R. E. McLaughlin & R. S. Nasci. 1991. J. Am. Mosq. Control Assoc. 7: 1-6.

Focks, D. A., R. E. McLaughlin & B. M. Smith. 1988a. Part 1. J. Am. Mosq. Control Assoc. 4: 266-281.

Focks, D. A., R. E. McLaughlin & B. M. Smith. 1988b. Part 2. J. Am. Mosq. Control Assoc. 4: 282-299.

Grover, K. K. 1985. pp.79-116. In Laird, M. & J. W. Miles [eds.] Vol. 2. Academic Press, London. 369 pp.

Kerwin, J. L. & R. K. Washino. 1988. J. Med. Entomol. 25: 452-460.

Kuntz, K. J., J. K. Olson & B. J. Rade. 1982. Mosq. News 42: 202-210.

Kunz, S. E., K. D. Murrell, G. Lambert, L. F. James & C. E. Terrill. 1991. pp. 69-98. In Pimentel, D. P. & A. A. Hanson [eds.], Vol. 1 (2nd ed.). CRC Press, Boca Raton.

Lacey, L. A. & S. Singer. 1982. Mosq. News 42: 537-543.

Laird, M. & J. W. Miles (eds.). 1985. Vol. 2. Academic Press, London. 444 pp.

Lehane, M. J. 1991. Harper Collins Academic, London. 288 pp.

Lucarotti, C. J. & B. A. Federici. 1985. pp. 251-268. In Laird, M. & J. W. Miles [eds.] Vol. 2. Academic Press, London. 369 pp.

McAllister, J. C. & C. L. Meek. 1991. Southwest. Entomol. 16: 185-192.

McLaughlin, R. E. & M. F. Vidrine. 1987. J. Am. Mosq. Control Assoc. 3: 633-635.

Meek, C. L. & J. K. Olson. 1977. Environ. Entomol. 6: 161-166.

Mulla, M. S. 1985. pp. 227-250. In Laird, M. & J. W. Miles [eds.] Vol. 2. Academic Press, London. 369 pp.

Scholl, P. J. & J. J. Petersen. 1985. In Williams, R. E., R. D. Hall, A. B. Broce & P. J. Scholl. John Wiley & Sons. 335 pp.

Sharma, V. P. 1985. pp. 177-140. In Laird, M. & J. W. Miles [eds.] Vol. 2. Academic Press, London. 369 pp.

Steelman, C. D. 1976. Ann. Rev. Entomol. 21: 155-178.

Steelman, C. D. & P. E. Schilling. 1977. J. Econ. Entomol. 70: 15-17.

USDA. 1979. USDA; E.P.A. and Kansas Agri. Exp. Sta. 322 pp.

Weathersbee, A. A., III & M. V. Meisch. 1990. Environ. Entomol. 19: 961-965.

Wiedhaas, D. E. 1974. J. Econ. Entomol. 67: 19-23.

Wright, R. E. 1985. In Williams, R. E., R. D. Hall, A. B. Broce & P. J. Scholl. John Wiley & Sons. 335 pp.

Tabanidae: Horse Flies and Deer Flies

Description and Biology. These flies (tabanids) are known by a large number of common names, but in the United States the large species which frequently attack livestock are most commonly called horse flies. The smaller species of the genus *Chrysops* which frequently attack man are often called deer flies.

Tabanids oviposit cylindrical eggs in masses (less than 100- 1000/mass) on objects above the larval habitat. The eggs hatch within 5-7 days during the summer months. The larvae are free-living in diverse situations ranging from moist sod to aquatic habitats.

Larvae feed on organic debris and/or small invertebrates. The larvae undergo rapid growth during the summer and fall, but are quiescent during cold weather. The larvae mature and pupate in drier soil; the adults emerge 1 to 3 weeks later. Life cycles require from 2 months to 2 years depending upon the species and geographical location. In most regions of the United States, adults of most of the species occur for only about a month, but a succession of species often is seen. The result is that livestock may be attacked by one or more species of Tabanidae throughout all or most of the warm months of the year. Most tabanids begin to feed shortly after emergence. Males feed on nectar, honeydew and other liquids and females feed on these and blood. Females of many species must obtain a blood meal prior to the development of each batch of eggs; however, several species lay one batch of eggs before they seek an animal host. Approximately 300 species are known to occur in North America; the relative importance of most of the species varies temporally and geographically.

Tabanids are pool-feeders that cut large, freely-bleeding wounds in host animals. Their numerous bite wounds serve as secondary feeding sites for other facultative blood-feeding flies (e.g., *Hippelates* and *Hydrotaea* species), and thus complicate the healing of these wounds. The bite wounds also serve as oviposition sites for myiasis-producing flies whose larvae feed in flesh wounds. Horse and deer flies also are important vectors in the epidemiology of several important livestock diseases.

Economic Importance. The annual losses in production and control cost due to horse and deer fly attacks on beef cattle are estimated by the USDA to be $40 million annually, of which $30 million were attributed to reduction in weight gains. Reductions in cattle body weight have averaged 100 lb/animal during massive outbreaks. In New York, for example, Tashiro & Schwardt (1949) reported that attacking *Tabanus quinquevittatus* occurred on cattle at a rate of 90 flies/h and *T. sulcifrons* at 40 flies/h during an attack period of 8-9 h/day. In northern Minnesota, Philip (1931) stated that in July from 40 to 50 flies could be seen on an animal at any time under most conditions, and in California and Nevada, Webb & Wells (1924) estimated that 25 to 30 *Hybomitra sonomensis* and *Tabanus punctifer* feeding on a host for 6 h would deprive that animal of at least 100 cc of blood. Weight deficits of about 100 lbs of normal increase have been estimated in cattle under heavy attack (Webb & Wells 1924), and Bruce & Decker (1951) in Illinois reported that beef cattle protected by repellents gained 20 to 30 lbs more than untreated cattle over the period of study.

Methods of Control. No satisfactory control methods are currently available. However, application of high pressure (100-200 psi) pyrethroid sprays can impact tabanid feeding success. The feeding time was significantly lower on treated cows for the 3 predominant horse fly species observed. The feeding times were reduced by 26% for *T. pallidescens*, by 33% for *T. fuscicostatus*, and by 39% for *T. lineola*. The amount of blood consumed by *T. fuscicostatus* was also significantly reduced by 29.3% for flies feeding on treated cows. Reducing the feeding time of a population of horse flies feeding on livestock by 35% would result in a 35% reduction of flies on the animal at any time. Reducing the amount of blood losses by approximately 30% would also be possible. A 44% reduction in daily blood loss to tabanid feeding for cattle treated with fenvalerate would be predicted from the combination of reduced subsequent feeding and reduced blood meal size (Foil et al. 1990, Leprince et al. 1991).

Needs for Insect Pest Management.

(1) Economic Injury Threshold. Few data are available relative to the effects of horse fly and deer fly attack on range cattle. Research is needed to show whether weight loss occurs due to tabanid attacks and whether compensatory gains in cattle body weight occur after periods of attack have ended. Studies are also needed to determine the effects of tabanid attack on animals raised on high and low energy rations.

(2) Control. Two types of control methodologies need to be developed. These are (a) areawide controls, and (b) individual animal protection techniques.

(3) Sampling. Sampling techniques for adults have been developed. However, the efficiency of these methods needs to be examined. Egg and larval sampling techniques need development. Population estimates of larval and adult sampling strategies should be compared. Basic biological and ecological data on larval habitat, adult flight and feeding behavior are needed for the majority of species which may be pests of range cattle.

(4) Disease Transmission. Horse flies may be important vectors of the organisms that cause several animal diseases (Krinsky 1976). They are known to transmit *Anaplasma marginale* (anaplasmosis in cattle), as well as the agents of Lyme disease, cutaneous anthrax, tularemia, equine infectious anemia, bovine leukemia, vesicular stomatitis, potomic horse fever, and hog cholera. Biological transmission of *Elaeophora schneideri* (arterial worm of deer) by tabanids to elk, deer and sheep has been described.

Research Needs.

(1) Basic Research.
(a) For most of the species, the basic biology-ecology needs to be studied. Seasonal activity, daily feeding cycles, larval habitats, etc. need to be known in order to develop control strategies.
(b) Laboratory rearing techniques, if only for 1 or 2 selected species, are needed to allow for basic physiology studies and the development of more efficient control.
(c) The role of tabanids in transmission of different diseases needs to be elucidated to help determine the economic impact of these pests.

(2) Control Components Research.
(a) Development of control technology including insecticide and application techniques needs to be developed. The pyrethroids have been shown to reduce feeding time and blood meal size of tabanids, but there are no accepted effective controls for tabanids.

Techniques to evaluate potential repellents of tabanids as well as other hematophagous diptera should be developed.
(b) The entire area of biological, ecological and genetic control needs to be investigated.
(c) The economic impact of tabanids from both the annoyances and disease transmission needs to be determined.

REFERENCES CITED

Bruce, W.N. and Decker, G.C. 1951. Tabanid control on dairy and beef cattle with synergized pyrethrins. J.Econ.Entomol. 44:154-160.

Foil, L.D. 1989. Tabanids as vectors of disease agents. Parasitol.Today 5:88-96.

Foil, L.D. and Hogsette, J.A. 1994. Biology and control of tabanids, stable flies, and horn flies. Rev.sci.tech.Off.int.Epiz. 13(4):1125-1158.

Foil, L.D. and Issel, C.J. 1991. Transmission of retroviruses by arthropods. Ann.Rev.Entomol. 36:355-381.

Foil, L.D., Leprince, D.J., and Byford, R.L. 1990. Sublethal effects and mortality of tabanids (Diptera: Tabanidae) induced by fenvalerate treatment of cattle. J.Entomol.Sci. 25:294-302.

Foil, L.D., Adams, W.V.,Jr., Issel, C.J., and Pierce, R. 1984. Tabanid (Diptera) populations associated with an equine infectious anemia outbreak in an inapparently infected herd of horses. J.Med.Entomol. 21:28-30, 1984.

Foil, L.D., French, D.D., Hoyt, P.G., Issel, C.J., Leprince, D.J., McManus, J.M., and Seger, C.L. 1989. Transmission of bovine leukemia virus by Tabanus fuscicostatus. Am.J.Vet.Res. 50:1771-1773, 1989.

Foil, L.D., Leprince, D.J., and Church, G.E. 1989. Changes in the parity rate of Tabanus fuscicostatus (Diptera: Tabanidae) populations associated with controlling available hosts. J.Med.Entomol. 26(4):306 310.

Krinsky, W.L. 1976. Review article: animal disease agents transmitted by horse flies and deer flies (Diptera: Tabanidae). J.Med.Entomol. 13:225-275.

Leprince, D.J., Foil, L.D., and Byford, R.L. 1991. Evaluation of pyrethroid ear tag and spray treatment of cattle against horse flies (Diptera: Tabanidae). J.Entomol.Sci. 26:271-280.

Philip, C.B. 1931. The Tabanidae (horseflies) of Minnesota with reference to their biologies and taxonomy. Minnesota Technical Bulletin 80:1-132.

Tashiro, H. and Schwardt, H.H. 1949. Biology of the major species of horseflies of central New York. J.Econ.Entomol. 42:269-272.

Webb, J. L. & R. W. Wells. 1924. U.S. Dept. Agric. Bull. 1218. 36 pp.

Ceratopogonidae (*Culicoides* and *Leptoconops*)

Description and Biology. Culicoides and *Leptoconops* are small (0.6 - 5.0 mm), compact, blood-sucking flies with the accepted common name of biting midges (Werner 1982). Species of *Culicoides* are commonly found throughout the United States, whereas members of the *Leptoconops* occur only in the western United States, where they are commonly called black gnats. Eggs of *Culicoides* are commonly deposited on vegetative matter partially immersed in water, on muck, moist soil or dung in exposed areas, and in moist treeholes. Certain species are found along the shallow edges of still waters of varying sizes, while others are associated with rivers or streams (Blanton & Wirth 1979). Most species of *Leptoconops* oviposit in damp sand, or sandy soil associated with alkaline or saline waters (Wirth & Atchley 1973), but species such as *L. torrens* and *L. carteri* occur in deep cracks in clay adobe soils in California (Whitsel & Shoeppner 1966).

Depending on the species and climatic conditions, the duration of the life cycle ranges from 2 weeks to as long as 6-7 months. The duration of the 4 larval instars for spring/summer generations requires 10 days to 2 months, with most species overwintering in the larval stage, and a few in the egg stage. The larval stage of certain species, such as *L. torrens* and *L. carteri*, may last 2 years. In drought periods these larvae may remain in diapause for up to 6 years.

The adults of *Leptoconops* spp. attack their host diurnally, with morning and afternoon peaks common; hosts include domesticated livestock and humans. *Culicoides* spp. adults are crepuscular and nocturnal, with peaks of activity at dusk and dawn. These flies may disperse in search of a blood source from a few yards to several miles from the point of emergence. They may disperse over long distances on weather fronts. Some species have 1-2 generations per year, but 7 or more are common in other species, depending on climate. In many parts of the United States, there are 2 peaks in the adult populations, a larger one in the spring and a smaller one in the fall.

Some 25 arboviruses have been isolated from species of *Culicoides*, including those causing such important livestock diseases as bluetongue, African horse sickness, akabane, eastern equine encephalitis, Venezuelan equine encephalitis, bovine ephemeral fever, and epizootic hemorrhagic disease (Karabatsos 1985). Akabane (in Japan and Australia) and bluetongue (worldwide), in addition to causing disease due to primary infection of the bitten ruminant host, can also infect the foetus causing various abnormalities (Murray 1977). The presence of bluetongue viruses in a country results in restrictions on livestock and germplasm trade with bluetongue-free countries.

Culicoides are also important vectors of filarial worms of the genus *Onchocerca*, including *O. gibsoni* in cattle in Europe Australia and Africa, and *O. cervicalis* in horses in North America. *Culicoides*, notably *variipennis* in North America, are also responsible for an allergic reaction in livestock that can cause intense itching, loss of coat, and wounds from rubbing.

Culicoides variipennis has received the most attention in the United States, as the principal vector of the bluetongue (BLU) viruses of livestock. This 'species' is actually a species complex, originally thought to have 5 subspecies (Wirth & Jones 1957); current research indicates that there may be only 3 subspecies, which may be different species (Tabachnick 1992, Holbrook, unpublished). There is a growing body of evidence to indicate that only one of its members, *C. v. sonorensis*, is implicated in the transmission of the 4 serotypes of BLU that are common in North America. The known distribution of *C. v. sonorensis* corresponds closely to the area in the U.S. where there is epidemiologic evidence of BLU transmission. Those area occupied solely by *C. v. variipennis,* i.e., the north central and northeastern states, are free of BLU transmission, and are being considered for establishment of a free trade zone for the international movement of cattle and their genetic byproducts (Tabachnick & Holbrook 1992, Walton et al. 1992).

Economic Importance. The annual losses to the livestock industry due to biting midges have been estimated to exceed $125 million yearly. A study of a bluetongue (BLU) outbreak in Mississippi in 1979 documented a loss of $35,000 in 194 herds, and an estimated $6 million throughout the state (Metcalf et al. 1980). The biting midges have received attention as pests and vectors of organisms affecting domestic livestock, and, to a lesser extent, wildlife. Little is known concerning effects on domestic range animals.

Methods of Control. Compounds registered for aerial and ground applications for the control of adult mosquitoes can provide temporary local reductions in biting midges. A number of chemicals have been evaluated against *C. variipennis* adults in wind tunnel tests (Holbrook 1986a), but there are no adulticides specifically registered for control of biting midges. Ear tags containing synthetic pyrethroids can be effective for at least 75 days on cattle but may not act fast enough on blood-feeding females to prevent pathogen transmission (Holbrook 1986b). Of the chemicals tested for larval control of *C. variipennis* (Holbrook 1982), only temephos is still registered, and has been used successfully in wide-area suppression (Holbrook 1984). Integrated management systems have been developed (Holbrook 1984, Mullens 1992). For *C. variipennis*, source reduction and larval control, in conjunction with adult monitoring, can be assimilated into ongoing mosquito control programs at a very cost-effective level (Holbrook et al. 1994). Information is available on survey tools for adult *Culicoides*, including recommendations for targeting different segments of the female population (Holbrook 1985, 1994, Holbrook & Bobian 1989).

Since *Leptoconops* spp. utilize damp soil along the edges of streams, lakes, ponds and marshy areas, impoundments can eliminate some of the breeding areas. However, if livestock have access to these impoundments, they can easily create favorable sites for

Culicoides infestation and development. Water and waste management can be effective but must be utilized judiciously.

There are a number of viral, bacterial, protozoan and nematode parasites of *Culicoides* (Wirth 1977, Blanton & Wirth 1979), and a number of predaceous ceratopogonid species can be found in *Culicoides* developmental sites (Holbrook & Grogan, in press), but there is little knowledge of the impact of these parasites and predators on the host populations. Genetic control methods for biting midges are currently being examined but have not been developed.

Needs for Integrated Pest Management.

(1) Economic Injury Levels. The effects of *Culicoides/Leptoconops* attack on range cattle has not been determined, and almost no data are available relative to these species and their population dynamics.

(2) Sampling. Although sufficient information is available on the uses of traps with various combinations of light sources, carbon dioxide, and suction (Holbrook 1994), the possiblilities of using pheromone or other attractant traps for sampling biting midges need to be investigated. There is little information regarding *Culicoides* other than *C. v. sonorensis*, and none relative to biting midges as they affect range cattle.

(**3) Biological-Ecological.** For support of IPM programs, basic biological and ecological data is available only for *C. v. sonorensis* (i.e., Barnard 1980, Barnard & Jones 1980a, 1980b, Mullens & Lii 1987, Mullens & Rodriguez 1988, 1990), and that information may be applicable only in limited situations.

(4) Pest-Host or IPM models. Sufficient information is available to initiate a crude model only for *C. v. sonorensis* but not to create the complex, interactive model necessary to propel IPM systems. A review of larval ceratopogonid biology and feeding behavior is available (Mullen & Hribar 1988).

Research Needs.

(1) Basic Research.

(a) Identify and determine the regional importance of species affecting beef production on range.
(b) Develop database on biological and ecological information on economically important species (developmental sites, seasonal abundance, host range, vector potential, sampling).
(c) Improve mass-rearing and colonization procedures, particularly for vectors such as *C. variipennis*.
(d) Improve methodologies for identifying and separating species and species complexes, particularly through the use of molecular biologic techniques.
(e) Amass genetic libraries of most important pest and vector species for use in genetically characterizing vector and/or pest potential.

(2) Control Components Research. In the case of known vector species such as *C. v. sonorensis*:
(a) Develop vaccines (for bluetongue).
(b) Develop water and waste management systems to reduce localized developmental sources.
(c) Develop genetic control methods.
(d) Develop insecticides and insecticide delivery systems.
(e) Develop economic threshold data to include losses due to reactions of sensitized animals, production losses, health (export) aspects.

REFERENCES CITED

Blanton, F. S. & W. W. Wirth. 1979. Florida Dept. Agric. Consumer Serv., Gainesville. Vol. 10, 204 pp.

Holbrook, F. R. 1986a. J. Florida Anti-Mosq. Assoc. 57: 1-3.

Holbrook, F. R. 1986b. J. Econ. Entomol. 79: 1127-1129.

Holbrook, F. R. 1982. J. Econ. Entomol. 75: 736-737.

Holbrook, F. R. 1984. Progress in Clin. and Biol. Res., Alan R. Liss, New York, 178: 617-620.

Holbrook, F. R. 1985. J. Am. Mosq. Contr. Assoc. 1: 379-381.

Holbrook, F. R. 1994. Bull. Soc. Vector Ecol. (In press).

Holbrook, F. R. & R. J. Bobian. 1989. J. Am. Mosq. Contr. Assoc. 5: 558-562.

Holbrook, F. R., S. V. Romney & R. M. Sessions. Wingbeats, Am. Mosq. Contr. Assoc. 4: 19-24.

Karabatsos, N. (Ed.) 1985. 3rd ed., CDC, Ft. Collins, CO, 1147 pp.

Metcalf, H. E., J. Lomme & V. C. Beal, Jr. 1980. Proc. U.S. Anim. Hlth. Assoc. 84: 186 202.

Mullens, B. A. 1992. Proc., CRC Press, Boca Raton, 896-905.

Murray, M. D. 1977. Mosq. News 37: 276-289.

Tabachnick, W. J. 1992. Ann. Entomol. Soc. Am. 85: 140-147.

Tabachnick, W. J. & F. R. Holbrook. 1992. Proc. U.S. Anim. Hlth. Assoc. 94: 207-212.

Walton, T. E., W. J. Tabachnick, L. H. Thompson & F. R. Holbrook. 1992. Proc., CRC Press, Boca Raton, 952-960.

Werner, F. G (Comm. Ch.). 1982. Entomol. Soc. Am.

Whitsel, R. H. & R. F. Shoeppner. 1966. Calif. Vector News 13: 17-25.

Wirth, W. W. 1977. WHO Bull. 54: 197-204.

Wirth, W. W. & W. R. Atchley. 1973. Tex. Tech. Press, Lubbock, No. 5, 57 pp.

Wirth, W. W. & R. H. Jones. 1957. U.S. Dept. Agr. Tech. Bull. 1170, 35 pp.

Ticks (Acari: Ixodidae) (Acari: Argasidae)

Description and Biology. Ticks (Acari: Ixodidae & Argasidae) are bloodfeeding ectoparasites in each postembryonic stage of larva, nymph(s) and adult. They attack livestock, poultry, equines and all taxa of wildlife. Their life cycles are composed of a variety of combinations of on-host and off-host periods and each species may be further characterized by seasonal activities, degrees of host specificity, generation times, and environmental requirements (Teel 1985, Sonenshine 1991, 1993). The proportion of the life cycle spent off of the host often exceeds 90% of the generation time (Needham & Teel 1991), thus the suitability and distribution of habitats on landscapes which support tick survival are considerations in the maintenance and growth of tick populations. Complex ecologies involving the interactions of habitat, livestock and wildlife, both native and exotic, form the basis for the development of pest populations and maintenance of zoonoses. These complex systems often pose challenging impediments to the management of tick populations and/or tickborne disease.

Concentration of the bloodmeal enables ticks to consume large quantities of whole blood. The total blood volume consumed and damage to the skin resulting from feeding lesions may result in reduced growth (rate of gain), weight loss, decreased reproductive efficiency, damaged hides, anemia and death. Transmission of tickborne disease agents is facilitated by tick salivary glands which function in the development and maintenance of the feeding lesion as well as the elimination of excess water from the bloodmeal. Tickborne bacterial, protozoal, rickettsial and viral diseases continue to negatively impact livestock production in the United States and globally.

Eighty species of the approximately 800 tick species known worldwide are found in the United States, and, of these, about 20 species are of veterinary importance. Several of these are of particular importance to the health and economic well being of range and pasture cattle production systems. The lone star tick, *Amblyomma americanum*, is found throughout much of the southeastern United States. All 3 stages will attack cattle and nymphs, and adults may be found in large numbers in spring and summer. The cayenne tick, *Amblyomma cajennense*, is found from southern Texas into Mexico. All stages attack cattle, and it is known to transmit spotted-fever group rickettsia. The Gulf Coast tick, *A. maculatum*, has two disparate populations, one distributed along the coastal regions of the South Atlantic and Gulf of Mexico, and the other in southeastern Kansas and north central Oklahoma. In the coastal regions of Texas, adult ticks reach peak populations on cattle in August and September; however, in Oklahoma peak adult populations occur in April-May. Of these species, *A. cajennense* and *A. maculatum* have been identified in laboratory transmission studies as potential vectors of *Cowdria ruminantium*, causal agent of heartwater in ruminants (Uilenburg 1982). This is significant due to the expansion of the tropical bont tick, *Amblyomma variegatum*, in the Caribbean and the potential for this tick to become established on the mainland of either North or South America (Barre' et al. 1987). The tropical bont tick is a principal vector of heartwater, benign African theileriosis, and is known to facilitate the dissimination of bovine streptothricosis. The cattle ticks, *Boophilus annulatus* and *Boophilus microplus*, and bovine babesiosis remain endemic to northeastern Mexico (Teclaw et al. 1985). Periodic reintroductions of these ticks into Texas continue. These ticks were successfully eliminated from the southeastern United States through a program of quarantine and eradication; however, new circumstances threaten reestablishment, including the discovery of organophosphate-resistant ticks in Mexico, the growing involvement of native and exotic ungulates on quarantined rangeland, and the limiting utility of effective pesticides in dip vats. The winter tick, *Dermacentor albipictus*, found from Canada throughout much of the United States and into Mexico, can build large populations on cattle during winter months when forage is limited and cold weather stress is high. In some areas this tick may be responsible for transmission of *Anaplasma marginale*. The Rocky Mountain wood tick, *Dermacentor andersoni*, is found in the northwestern United States; is responsible for tick paralysis and is a vector of anaplasmosis and Rocky Mountain spotted fever. The American dog tick, *Dermacentor variabilis*, is generally distributed throughout the eastern half of the United States. It attacks a variety of livestock including cattle. It is the principle vector of Rocky Mountain spotted fever in this region, is known to cause tick paralysis and has been implicated in the transmission of bovine anaplasmosis. Distribution of the Pacific Coast tick, *Dermacentor occidentalis*, is limited to the Pacific coastal region, where it is a vector of anaplasmosis and known to cause tick paralysis. The black-legged tick or deer tick, *Ixodes scapularis* (=*dammini*), may be found throughout most of the eastern half of the United States, including the Great Lakes states and extending down into Texas and is collected from cattle during winter and spring months. The spinose ear tick, *Otobius megnini*, may be found feeding as immatures deep inside the outer ears of cattle from Canada, throughout the western and southwestern regions of the United States, and into Mexico. The pajaroello tick, *Ornitodoros coriaceus,* is distributed from southern Oregon through California and into Mexico. It has been associated with the bedding grounds of deer and cattle and is incriminated as a vector of Epizootic Bovine Abortion (EBA) (Schmidtmann et al. 1976), whose putative causal agent is a spirochete (Lane et al. 1985).

Economic Importance. Production losses associated with tick parasitism in the United States were estimated at $275.7 million by Drummond et al. (1981). This estimation does not include the cost of tick control, losses associated with tickborne diseases, nor the cost of the cattle fever tick eradication program. Data from controlled studies of lone star and Gulf Coast ticks parasitizing preweaners, stockers and steers, were used to estimate annual losses of $82 and $58 million for these species, respectively (Drummond 1987). Kunz et al. (1991) revised these values to $29 million for lone star and $75 million for Gulf Coast ticks. These estimates were based upon production and sales values and appropriate cattle census data.

Methods of Control. Control of ticks on range cattle in the United States is largely dependent upon treatment of infested animals with registered acaricides (Drummond et al. 1988). Appropriate formulations enable animals to be treated by immersion in dipping vats, by whole-body, full volume, high-pressure spraying, or by dusting. Tick species which feed primarily in the ears may be controlled by applying acaricides formulated in dusts, ointments, sprays, aerosols, or sustained-release ear tags. Acaricides registered for use in the United States are predominantly organophosphates, synthetic pyrethroids and amidines. The increasing cost of registering new compounds relative to cost recovery is a deterrent to the registration of new compounds and formulations for delivery. Nevertheless, research with promising systemic compounds, such as the avermectins, or with new chemistry such as the juvenile hormone mimics, in combination with novel delivery and sustained-release systems hold promise for the future.

The effectiveness of acaricides applied to range cattle on the tick population largely depends on the role range cattle play in the ecological processes driving the maintenance and growth of the tick population. Wildlife hosts ranging from deer to ground dwelling birds may maintain the population of ticks, or as in the case of *O. corieaceus*, acaricide effectiveness may be limited due to an extremely short feeding period.

The greatest proportion of tick life cycles are completed off the host in microenvironments which provide suitable conditions for development and longevity (Needham & Teel 1991). Altering habitats to render them less suitable for tick development and survival by mechanical means, herbicides or through use of prescribed fire has been conducted with varying degrees of success (see reviews by Warren et al. 1987, Drummond et al. 1988, Schmidtmann 1994). Studies of these treatments all too frequently measure only the acute impact on the pest population, or may extend to the first vegetative regrowth season. Often the initial benefits of reductions in the target population are determined to be nullified by the end of the first regrowth season due to interactions of vegetative regrowth and recruitment of tick infested animals on treated sites. Warren et al. (1987) discuss the time frames and changes in vegetative succession of rangeland following prescribed fire which lead to new steady states and suggest that long-term studies (2-3 yrs) of treated sites are indicated to accurately assess the new states of arthropod populations and their systems. Host-tick contact is a critical parameter in ecological processes regulating the dispersal and maintenance of tick populations (Barnard 1991, Teel et al. 1993). Temporal and spatial relationships of foraging behavior of cattle and certain wildlife species relative to resource/habitat utilization on landscapes are the basis of rotational grazing systems in tick management. Coupled closely to this are accurate tick density and spatial estimates of free-living populations.

Cattle breeds of the *Bos indicus* genotype are most resistant to ticks while breeds composed of genotypes mixed with the most susceptible *Bos taurus* tend to be intermediate in tick resistance. In some tropical and subtopical regions of the world, tick resistant cattle have been incorporated into tick management programs. Investigations of the basis for host acquired immunity to ticks have revealed that antigens on tick gut digest cells, normally concealed from the host, can be used to vaccinate cattle against ticks (Tellman et al. 1992). Ticks feeding on vaccinated cattle imbibe antibodies to gut cells which ultimately disrupt the integrity of the gut, resulting in death or reduced feeding and reproductive capacity. Several technologies for vaccine development have evolved and this avenue of control holds exciting promise in future tick management strategies.

A variety of predators, parasites, pathogens, and tropical grasses are known to cause mortality of ticks (Drummond et al. 1988, Sonenshine 1993). To date, successful avenues to enhance the use of these organisms in biocontrol strategies against ticks have not been discovered.

Hybrid sterility in ticks produced from cross mating *B. annulatus* and *B. microplus* has been evaluated under field conditions and determined to be of limited practical value.

Pheromones used by ticks for aggregation and mating have been used alone and incombination with acaricides to attract and kill ticks both on animals and on the ground (Sonenshine 1993). This was demonstrated for *A. maculatum* in the United States and *A. hebraeum* in South Africa. Recently, male aggregration-attachment pheromone was added to carbon-dioxide baited traps to successfully collect freeliving adultA. hebraeum and *A. variegatum* in Africa (Norval et al. 1992). Carbon dioxide had previously been shown to only exite these species but not attract them in substantial numbers to the traps; the addition of the pheromone provided the needed attractancy. Various combinations of excitors and attractants may improve sampling for a variety of species.

Development and assessment of integrated pest management strategies against ticks have necessitated the development of simulation models to evaluate potential impacts various components and combinations of components might have on tick populations. Simulation models designed for application to tick control and IPM have been developed for *A. americanum*, *Boophilus* sp., and *D. variabilis* (Mount et al. 1989, 1991, 1993, Teel et al. 1994).

Native and exotic deer are frequently involved in the maintenance of tick populations affecting livestock and humans. In such cases exclusion fencing, population reduction/relocation, self-treatment devices and feeding of systemically active acaricide-ladened supplements have been attempted to suppress tick populations.

Needs for Integrated Pest Management. Sustained suppression of tick populations below limits which cause economic loss must rely on strategies of integrated treatments. The complex life cycles of ticks affecting livestock involve the interactions of hosts and habitats at landscape levels and only rarely will a single treatment-type provide satisfactory suppression. The goals of Integrated Pest Management strategies for ticks must be compatible with production goals of the ranching enterprise which often may include production and harvest of wildlife in addition to cattle. In many areas of the United States, interest in exotic animals, especially hoof-stock, has given rise to new animal industries. The diversity and density of exotics on landscapes with and without livestock species present new and unique tick problems whose solutions will require multi-faceted approaches inherent to IPM.

In the 1970's limited economic injury data were collected in controlled studies for 2 species, the lone star tick and Gulf Coast tick, on preweaners, stockers and steers, and have been beneficial in developing economic impact assessments (Drummond 1991). Comparable data are not available for any of the other tick species attacking cattle. Virtually no data are available on the impact of ticks on mother cows and calves, yet cow-calf production systems make up the largest share of the cattle industries throughout much of the United States. Estimating losses in these systems presents unique difficulties due to the reproductive cycle. Changes in body condition scores, calf weight, milk production, and return to estrus will be among the parameters to be measured.

Progress in assessing impacts of habitat modifications both to tick suppression and progress in developing simulation models for tick population dynamics and control strategies have both indicated the need to better estimate free-living populations of ticks, how hosts interact with disturbed and nondisturbed landscapes, and how levels of tick resistance in cattle respond to changing field challenges of ticks.

Research Needs.

(1) Basic Research.

(a) Determine the biotic and abiotic factors regulating survivial and fecundity of ticks.

(b) Determine the effects of hosts and host-landscape interactions on abundance, survival and fecundity of ticks.

(c) Investigate more precise sampling methods for estimating both host-seeking and non-host seeking ticks and correlate estimates of tick populations on host animals with field populations.

(d) Evaluate tick-host mediated and artificially induced resistance to ticks.

(e) Investigate within species genetic variation of ticks for geographic strains.

(f) Define the physiological and ecological relationships associated with zoogeography of ticks.

(g) Develop and enhance population and systems simulation models for evaluation of IPM strategies.

(h) Evaluate the biological and economic impact of IPM strategies on tickborne diseases.

(I) Investigate and develop novel pesticide delivery systems.

(2) Control Components Research.

(a) Document economic losses to each tick species in appropriate breed, sex and age class of cattle to assess the cost-benefit analyses of IPM strategies.

(b) Evaluate economical and ecological value of resistant cattle breeds and induced tick resistance in IPM strategies.

(c) Validate predictive and simulation models developed for population dynamics and IPM strategies.

(d) Combine mangement practices for cattle and wildlife that can be used for tick control; determine their practicality for cattle production systems.

(e) Determine the biological significance of subeconomic levels of tick populations.

(f) Evaluate the suitability of native and exotic wildlife hosts and the potential ecological roles they may have in tick population dynamics.

(g) Improve effectiveness and reduce application costs of acaricides to control ticks.

REFERENCES CITED

Barnard, D. R. 1991. J. Med. Entomol. 28: 557-564.

Barre', N., G. Uilenburg, P. C. Morel & E. Camus. 1987. Onderstepoort J. Vet. Res. 54: 405-417

Drummond, R. O., J. E. George & S. E. Kunz. 1988. CRC Press, Boca Ration. 245 pp.

Drummond, R. O. 1987. pp 9-26. *In* Proc. [Eds.] W. H. D. Leaning & J. Guerrero. Vet. Learning Systems Co., Inc., Lawrenceville, N.J.

Drummond, R. O., G. Lambert, H. E. Smalley, Jr. & C. E. Terrill. 1981. pp. 111-127. Vol. I. [Ed.] D. Pimmentel CRC Press, Boca Raton.

Kunz, S. E., K. D. Murrell, G. Lambert, L. F. James & C. E. Terrill. 1991. pp. 69-98. Vol. I. [Ed.] D. Pimmentel. CRC Press, Boca Raton.

Lane, R. S., W. Burgdorfer, S. F. Hayes & A. G. Barbour. 1985. Science 230: 85-87.

Needham, G. R. & P. D. Teel. 1991. Ann. Rev. Entomol. 36: 659-681.

Norval, R. A. I., T. Peter & M. I. Meltzer. 1992. Exp. & Appl. Acarol. 13: 179-186.

Russell, L. H., Jr., P. D. Teel & D. E. Bay. 1991. pp. 623-635. Vol. I. 2nd Ed. [Ed.] D. Pimmentel. CRC Press, Boca Raton.

Schmidtmann, E. T., R. B. Bushnell, E. C. Loomis, M. N. Oliver & J. H. Theis. 1976. J. Med. Entomol. 13: 292-299.

Schmidtmann, E. T. 1994. In Chapt. 8 [Eds.] D. E. Sonenshine & T. M. Mather. Oxford Univ. Press, London.

Sonenshine, D. E. 1991. Vol. I. Oxford Univ. Press, N.Y. 447 pp.

Sonenshine, D. E. 1993. Vol. II. Oxford Univ. Press, N.Y. 465 pp.

Steelman, C. D. 1991. Vol. I. 2nd Ed. [Ed.] D. Pimmentel. CRC Press, Boca Raton.

Teclaw, R. F., S. Romo, Z. Garcia, M. Castaneda & G. G. Wagner. 1985. Prev. Vet. Med. 3: 403-415

Teel, P. D. 1985. pp 129-149 In Livestock Entomology. [Eds.] R. E. Williams, R. D. Hall, A. B. Broce & P. J. Scholl. John Wiley & Sons, N.Y.

Teel, P. D., O. F. Strey, M. T. Longnecker & D. E. Bay. 1994. J. Med. Entomol. (In press).

Tellman, R. L., D. Smith, D. H. Kemp & P. Willadsen. 1992. pp. 303-331. In Animal Parasite Control Utilizing Biotechnology. [Ed.] W.K. Yong. CRC Press, Boca Raton.

Uilenburg, G. 1982. Am. J. Vet. Res. 43: 1279-1282.

Warren, S. D., C. J. Scifres & P. D. Teel. 1987. Agric. Ecosys. & Environ. 19: 105-130.

Mange Mites (*Psoroptes ovis*)

Description and Biology. Psoroptic scabies mites live on the surface of the skin of cattle where they feed on lymph or plasma obtained by abrading the skin (Rafferty & Gray 1987). The activity and secretions of the mite cause intense itching that results in scratching, biting, or rubbing by the host. The lesions caused by the mites become crusty and increase in size as the mite populations increase. The heavier infestations occur in the winter months and can spread rapidly throughout a herd.

The irritation caused by the mites results in considerable physical damage of the withers, shoulders areas of the cattle. Mite infestations can interfere with weight gains (Meleney & Fisher 1979, Fisher & Wright 1981a, Cole et al. 1984), and calves under 1 year of age may die if not treated (Kemper & Peterson 1953). A serious problem, however, is related to the restriction or embargo placed on the movement of cattle due to the quarantine for scabies. Scabies is considered by State and Federal regulatory agencies to be a quarantinable disease, and any known or suspect cases are required by law to be reported to the nearest animal health officer.

Besides cattle, sheep, horses, goats, rabbits, Bighorn, and wapiti are eligible hosts for one or more of 4 species of *Psoroptes* in North America (Sweatman 1958). Microscopic examination is necessary to differentiate between *Psoroptes* and other parasite mites.

Economic Importance. The annual losses in production costs are estimated to be $58 million (Drummond 1987). Most of the losses are in the Great Plains region. Most important regions appear to be the Texas and Oklahoma Panhandles, eastern New Mexico and Colorado, western Kansas, and all of Nebraska (Meleney & Christy 1978). Cattle scabies now seems to be firmly established in native California cattle (Meleney & Christy 1978). Although the monetary losses are less than for other pests and parasites, mange or scabies mites are potentially a very important parasite.

Methods of Control. USDA Veterinary Services Memoranda No. 556.1 and 556.10 currently permit 4 products for use against psoroptic scabies (scab) of cattle. These are (1) hot lime sulfur (95-105oF) 2% polysulfides of sulfur, (2) coumaphos 0.30% aqueous suspension, (3) phosmet (Prolate®) 0.25% aqueous suspension with 10 lbs of super phosphate or triple super phosphate fertilizer (0-45-0) added per 100 gallons of dip (the phosphate is necessary as a buffer so that the phosmet will not deteriorate in the vat upon standing due to excess acidity), and (4) ivermectin 200 µg per kilogram of body weight.

Cattle that are known to be infested or directly exposed to scabies are quarantined and dipped twice at 10- to 14-day intervals in the first 3 compounds. The double dipping was originally required to reduce the possibility of missing some animals, but since coumaphos and phosmet were added to the permitted list, 2 dippings are known to be necessary to kill all mites on animals dipped in those 2 products. Several dippings may be necessary in hot lime-sulfur before eradication of mites is achieved. Federal Memorandum 556.1 specified the treatment of cattle infested with or exposed to scabies in an immersion type (plunge or cage) vat or in a spray-dip machine, but some states do not allow the use of the spray-dip machine. Cattle are given one subcutaneous injection of ivermectin at a dose of 200 µg/kg (10 mg/110 lbs) of body weight. A herd or lot of animals that has been treated with ivermectin must be allowed to commingle with other animals for at least 14 days after treatment. Regulations are different for each state. Federal regulations supersede State regulations unless the State regulations are more stringent than the Federal regulations.

Hot lime-sulfur is the only material that can be used to treat lactating dairy cows. There is no withholding period for cattle prior to slaughter for hot lime-sulfur and coumaphos, but cattle treated with phosmet and ivermectin must be withheld from slaughter for 21 or 35 days, respectively.

The Memorandum now in force calls for animals that are dipped to be completely submerged, and the head of the animal submerged at least once after it has been plunged into the vat. Animals should be held in the vat until "wet to the skin." Guidelines for the use of spray dip machines can be found in Veterinary Services Memorandum No. 556.5 and Supplement No. 1.

As far as the Federal regulatory agencies are concerned, quarantines may be lifted after the cattle have been dipped a second time. However, animals treated with ivermectin must be isolated and held for 14 days. Some states require negative post dipping examinations before quarantine can be lifted.

Finally, there are Federal and State regulations governing the movement of scabies infested or -exposed cattle after treatment.

Little research has been conducted in the past to develop control methods other than chemical. Studies on host resistance (Stromberg & Fisher 1986) offer hope for the development of a vaccine to control *Psoroptes* on the host. Brahman-cross cattle have been observed to be more resistant than European breeds to psoroptic infestations (Fisher & Wright 1981b).

Needs for Insect Pest Management. Because of the nature of the psoroptic mite and the legal ramifications of its presence in a herd of cattle, it is doubtful if insect pest management *per se* can be used against this pest. Once an infestation is detected, its management is directed, if not conducted, by regulatory agencies for its control. Sampling consists of examining skin scrapings which require checking under magnification. For quarantine purposes, samples are either scored negative or positive. Any positive finding, regardless of the numbers involved, results in quarantine of the herd from which the sample was taken.

Research Needs.

(1) Basic Research.
(a) Biological-ecological studies to elucidate the population dynamics of psoroptic mites through the various seasons of the year.
(b) Basic studies on the physiology of psoroptic mites.
(c) Basic studies on the development of the immune response of animals to psoroptic mite infestations.

(2) Control Components Research.
(a) The development of vaccines to induce or increase an animal's resistance to scabies.
(b) The development of methods of treatment such as systemic chemicals to replace the laborious and expensive dipping procedures currently in use.
(c) The development of sound economic data showing monetary losses will greatly enhance the success of any future efforts to convince the beef industry of the need for eradication attempts. This data should not include only the direct cost of the pest and its control but costs of quarantine premises, added feed costs, cost of reduced grade due to improper timing of slaughter for feedlot animals, etc. This will require the expertise of an economist to develop the whole package.

REFERENCES CITED

Cole, N. A., F. S. Guillot & C. W. Purdy. 1984. J. Econ. Entomol. 77: 390-393.

Drummond, R. O. 1987. pp. 9-24. In The Economic Impact of Parasitism in Cattle. W. H. D. Leaning & J. Guerrero [ed.] Veterinary Learning System Co. 111 pp.

Fisher, W. F. & F. C. Wright. 1981a. J. Econ. Entomol. 74: 234-337.

Fisher, W. F. & F. C. Wright. 1981b. Southwest. Entomol. 6: 57-61.

Kemper, H. E. & H. O. Peterson. 1953. USDA Farmers' Bull. No. 1017. 26 pp.

Meleney, W. P. & J. E. Christy. 1978. J. Amer. Vet. Med. Assoc. 173: 1473-1478.

Meleney, W. P. & W. F. Fisher. 1979. Proc. 83rd Ann. Meet. U.S. Anim. Health Assoc. 83: 340-346.

Rafferty, D. E. & J. S. Gray. 1987. J. Parasitol. 73: 901-906.

Stromberg, P.C. & W. F. Fisher. 1986. Am. J. Vet. Res. 47: 1551-1560.

Sweatman, G. K. 1958. Can. J. Zool. 36: 905-929.

Texas Animal Health Commission Regulations, Chapter 39. 1993.

USDA, APHIS, Veterinary Services, Memoranda 556.1, 556.1-Suppl. 2, 6 & 8, 556.5, 556.5-Suppl 1, 556.10.

Cattle Lice

Description and Biology. Cattle in North America may be infested by four species of sucking lice, (1) *Haematopinus eurysternus* (Nitzsch), the shortnosed cattle louse, (2) *H. quadripertusus* Fahrenholz, the cattle tail louse, (3) *Solenopotes capillatus* (Enderlein), the little blue cattle louse, and (4) *Linognathus vituli* (L.), the longnosed cattle louse; and one species of biting louse, *Bovicola bovis* (L.), the cattle biting louse. The cattle tail louse occurs along the Gulf Coast in the United States and is most abundant on cattle in late summer. The remaining 4 species are most abundant in late winter and are common on cattle throughout the United States. Although there may be regional variations in the relative abundance of the species, these have not been verified accurate surveys. *L. vituli* is probably the most common sucking louse on young cattle throughout the country.

Cattle lice are small (the adults are only a few millimeters in length) wingless insects that spend their entire existence in the hair coat of the host. Nymphal lice resemble the adults but are smaller in size. The eggs are individually glued to hairs. With the exception of the cattle tail louse, cattle lice oversummer at low population levels on cattle. The importance of carrier animals in maintaining summer populations within a herd has not been determined.

The damage usually attributed to sucking lice infestations is blood loss and irritation. In addition to actual blood loss, the toxic effect of louse feeding appears to play a role in development and maintenance of louse anemia in cattle. Although very little is known about the immune response of cattle to lice, the response of the host's immune system to other parasites has been demonstrated to result in reduction in feed intake, nutrient use, and growth.

Infestation of the cattle biting louse are considered less important economically, and from an animal health view, however, this species can be extremely irritating to the host. The result of irritation by both sucking and biting lice is rubbing and hair loss.

Economic Importance. Parasitism by sucking lice results in direct economic losses such as reduced weight gains and, possibly, reduced vigor and resistance to diseases. The effect of lice becomes more important when cattle are under the stress of cold, inclement weather or poor nutrition. Heavily infested animals may die from anemia and pregnant cows may abort. Annual losses in control costs and production losses due to lice on beef cattle have been estimated to exceed $100 million annually (Anonymous 1976). Drummond (1981) estimated annual losses in cattle production due to lice to be $126.3 million.

Damage, which has been attributed to insects by the American Leather Chemists Association, costs the tanning and beef industries in the United States millions of dollars a year. The primary hide defects are "pitting" (presumably resulting from insect bites) and scratches. It is estimated that upwards of two-thirds of all hides could be affected, and downgrading hides could cost as much as $40 to $50 per hide.

Methods of Control. A variety of insecticides, including organophosphates, chlorinated hydrocarbons, pyrethroids, amitraz and ivermectin, is used effectively for cattle lice control. Cattle lice control may be achieved through preventive, fall treatment with systemic insecticides for control of cattle grubs.

Though cattle may be treated with whole body sprays and dips, disposal of used dip solutions is an environmental concern. The development of injectable and pour-on formulations has aided, considerably, in the control of cattle grubs and cattle lice. These methods are labor saving and are less stressful to the cattle, especially under winter-time conditions.

New developments that may eventually have a place in lice control include various new insecticides (systemic as well as nonsystemic) and improved application technology. Promising pyrethroids, organophosphates, avermectins, milbimycins and insect growth regulators are under development. New delivery systems will include devices such as insecticide ear tags, neck bands, etc.

Little research is being performed on non-insecticidal approaches to cattle lice control. The isolation of *Bacillus thuringiensis* strains against the sheep biting louse may lead to the development of this bacterium for control of lice in livestock (Drummond et al. 1992).

Lice populations on cattle differ considerably in severity and species composition, even within a single herd of the same breed of cattle. Little is known of the underlying host resistance mechanisms. Basic knowledge of these mechanisms will be necessary for the development of novel molecular approaches to the protection of cattle from lice infestations.

Little definitive research has been done with genetic control of cattle lice. Since some breeds, and animals within breeds, seem to be more refractory to lice, breeds or lines within breeds of animals might be selected that would be more resistant to louse attack.

Animal nutrition appears to play an important role in the dynamics of louse populations. Several workers have suggested that louse populations increase with decreased nutritional status (Jones 1965, Ely & Harvey 2969, Utech et al. 1969, Tweedle et al. 1977). Possibly the role of dietary factors could be important in reducing cattle lice to subeconomic levels.

Needs for Insect Pest Management.

(1) Economic Injury Level. Little information is available on economic injury levels of louse infestations on cattle. Much of the information relative to effects on economic injury levels, weight gain, and feed efficiency that is currently available is equivocal. Extremely variable lice populations - including various combinations of from 1 to 4 species of lice - and variations in methods of scoring or indexing lice infestations in different geographic regions make comparisons between studies difficult. Gibney et al. (1985) found significant differences between weight gains of cattle heavily infested with multiple species of lice and weight gains of uninfested cattle. Shemanchuk et al. (1960) found that control of heavy louse burdens improved weight gains by 0.41 lb per day. Collins & Dewhirst (1965) reported that heavily infested cattle lost significantly more weight in the winter than did animals with lighter infestations. Scharff (1962), Kettle (1974), and Tweedle et al. (1977) were unable to demonstrate differences in live weight gain between treated or untreated cattle, yet Scharff (1962) and Tweedle (1977) noted animals in poor condition, some of which were suffering severe anemia, and eventually died. This problem needs to be elucidated under carefully controlled experimentation.

In practice, winter and spring treatments for cattle lice control are, generally, applied by producers when animals appear to be rubbing and losing hair and is not based on actual observation of lice. The actual relationship between lice infestation and cattle self-grooming has not been studied. There is a need for knowledge of this animal behavior in response to lice infestations.

(2) Sampling. Various numerical rating systems have been used to estimate louse populations on animals based either on individual animal counts or on herds using the individual counts to assess an average herd infestation. These ratings have not been correlated to levels of damage or losses. An assessment of the various index methods and determination of the most efficient method based on the distribution of each species is necessary. Watson (1984) provided a statistical analysis of an indexing system with actual counts of lice removed from cattle after slaughter.

(3) Biological-Ecological. Very little biological-ecological data have been generated in recent years. The oversummering biologies of most lice remain unknown. Treatments applied to more susceptible, oversummering lice populations could contribute to or eliminate the need for control of the higher wintertime populations. Identification of summer "carriers" and their treatment or removal from the herd, might reduce the severity of winter infestations.

(4) Pest-Host Models. Models, *per se*, for cattle lice are not available. Information on seasonal dynamics, based on systematic sampling of the different species is lacking from most regions of the country. There is little or no information on annual variations in lice populations. Development of preliminary models could well serve to define the research needed to develop IPM programs. A study of host-parasite relationships of mouse lice (reviewed in Nelson et al. 1977) has produced basic information that may provide a model for cattle louse population dynamics.

Research Needs.

(1) Basic Research.
(a) The basic biology-ecology the most common louse species needs to be studied.
(b) Elucidate the underlying host resistance mechanisms.
(c) The oversummering dynamics of lice need to be elucidated to determine how these low level populations can be manipulated to reduce or eliminate infestations in the winter seasons.
(d) Develop alternatives to chemical control measures, e.g., vaccine, nutritional supplementation, herd management practices, biological control, etc.
(e) Develop serological detection methods.
(f) Standardize survey techniques and correlate numerical ratings with total populations.

(2) Control Components Research.
(a) The economic thresholds and animal health effects of louse populations need to be developed for various climatic zones.
(b) The effect of louse control on identified summer carriers in wintertime infestations needs to be evaluated.
(c) Evaluate the effectiveness of the new insecticides and drugs in controlling lice.
(d) Develop population models which could be used to implement IPM for louse control.

REFERENCES CITED

Anonymous. 1976. NRP No. 20480, USDA, 99 pp.

Butler, J. F. 1985. pp. 101-127. In Williams, R. E., R. D. Hall, A. B. Broce & P. J. Scholl [eds.] John Wiley.

Chamberlain, W. F., D. E. Hopkins & A. R. Gingrich. 1976. Southwest. Entomol. 1: 1-8.

Collins, R. E. & L. W. Dewhirst. 1965. J. Amer. Vet. Med. Assoc. 146: 129-132.

DeVaney, J. A., L. D. Rowe & T. M. Craig. 1988. Southwest. Entomol. 13: 125-129.

Drummond, J., D. K. Miller & D. E. Pinnock. 1992. J. Invert. Pathol. 60: 102-103.

Drummond, R. O., G. Lambert, H. E. Smalley, Jr. & C. E. Terrill. 1981. pp. 111-127 In Pimentel, D. [ed.] Vol. 1. CRC Press.

Ely, D. G. & T. L. Harvey. 1969. J. Econ. entomol. 62: 341-344.

Geden, C. J., D. A. Rutz & D. R. Bishopp. 1990. J. Econ. Entomol. 83: 1435-1438.

Gibney, V. J., J. B. Campbell, D. J. Boxler, D. C. Clanton & G. H. Deutscher. 1985. J. Econ. Entomol. 78: 13-4-1307.

Jones, L. D. 1964. Cornell Vet. 55: 399-405.

Kettle, P. O. 1974. N. Z. Vet. J. 22: 10-11.

Meleney, W. P. & I. H. Roberts. 1975. J. Parasit. 61: 956-959.

Nelson, W. A., J. A. Shemanchuk & W. O. Haufe. 1970. Exper. Parasitol. 28: 263-271.

Nelson, W. A., J. F. Bell, C. M. Clifford & J. E. Keirans. 1977. J. Med. Entomol. 13: 389 428.

Scharff, D. K. 1962. J. Econ. Entomol. 55: 684-688.

Shemanchuk, J. A., W. O. Haufe & C. O. M. Thompson. 1960. Can. J. Compar. Med. 24: 158-161.

Shemanchuk, J. A., W. O. Haufe & C. O. M. Thompson. 1963. Can. J. Anim. Sci. 43: 56 64.

Tweedle, N. E., L. J. Cummins & J. F. Graham. 1977. Proc. 54th Ann. Conf. Aust. Vet. Assoc. pp. 89-91.

Utech, K. b. W., R. H. Wharton & L. A. Wooderson. 1969. Aust. Vet. J. 45: 414-416.

Watson, D. W. 1984. M. S. Thesis, Univ. Wyoming. 127 pp.

Screwworm (*Cochliomyia hominovorax)*

Description and Biology. Screwworm is an obligate parasite of warm-blooded animals. Female flies oviposit masses of about 200 eggs on or near open wounds caused by mechanical injury, insect or tick bites, and on the moist umbilicus of new born animals. The eggs hatch and the larvae (or worms) enter the wound and feed upon the living flesh, increasing the size of the wound as they feed and grow. If the wound is untreated, the animal will die as a direct or indirect result of the infestation. After 5-7 days the larvae crawl out of the wound, drop to the ground, and pupate in the soil. Adults emerge after 7-10 days and mate about 3-4 days later. The females then begin their ovipositional cycle, which depending on fly longevity, may occasionally result in as many as 10 egg masses over 30 days.

Screwworm distribution is primarily restricted by weather, movement of infested animals, control efforts of the Joint Mexico-United States Commission for the Eradication of Screwworms, and cooperative eradication programs between the ministries of agriculture of each Central American country and the U.S. Department of Agriculture. Winter freezes limit overwintering ability, but screwworm can survive throughout the year in the warmer climate of the southern United States. Although screwworm was eradicated from the United States in 1966 and Mexico in 1991, vigilance must be maintained due to potential reintroduction via modern rapid transportation of animals with undetected infestations. The goal of the USDA-APHIS cooperative eradication programs is to eradicate screwworm from Central America and maintain a permanent barrier zone in the Darien Province of the isthmus of Panama, where sterile flies will be dispersed to prevent immigration of this pest from South America.

Economic Importance. Presently, the cost to conduct the eradication program is $35 million annually. Based on 1992 figures and half of the U.S. livestock population being at risk, the annual benefit to U.S. livestock producers (all species) is estimated to be $715,334,880. Producer benefits and its linkage effect on the economy are estimated at $2.5 billion annually. Doubling that figure to account for consumer benefits leads to an estimated $5 billion total benefits annually. Establishment and maintenance of the barrier zone in Panama will yield an estimated $23.8 billion in benefits to the United States, Mexico and Central American economies annually to perpetuity. The cost benefit ratio is estimated to be $11:1.

Methods of Control. Registered insecticides are available for screwworm control in animals when applied as sprays, dips, or wound treatments. However, the sterile insect release technique is the driving force of the eradication programs.

Using the sterile insect release technique, screwworm was eradicated from the United States and Mexico, as previously noted. Sterile flies were first released into Guatemala in 1988, and the last case was reported in May 1992. Sterile flies were first released in Belize in August 1989, and the last case was reported in October 1991. In El Salvador, sterile flies were first released in July 1991, and the country was declared free of screwworm in October 1993. Release of sterile flies in Honduras began in November 1991. Except the eastern tip, screwworm is considered eradicated from Honduras (west of 84° longitude). Presently (May 1994), approximately 115 million sterile flies are released weekly in Honduras. Sterile flies were first released in Nicaragua in July 1993, and about 69 million are currently released in an approximate 80 km swath along the Honduran border. Eradication of screwworm from Nicaragua is expected by 1996. An agreement between the governments of Costa Rica and the United States has been signed to eradicate screwworm from Costa Rica. Pre-release activities are scheduled to begin in 1994 with eradication expected by 1997. An agreement was signed in February 1994 between the governments of Panama and the United States concerning the construction of a sterile fly production plant and to establish a barrier zone across the isthmus of Panama in the Darien Province.

Reintroduction of screwworm into the United States has occurred 4 times since 1980. Three times were in infested dogs, 1981 from Mexico; April and August of 1987 from Venezuela and Honduras, respectively. Sterile flies were dispersed for 6 weeks in an 80 km radius of where the dogs had been in the United States. The fourth introduction was in a soldier returning from Panama in the winter of 1989. The potential for reintroduction into the United States or introduction into nonendemic areas is exemplified by screwworm's introduction into Libya, North Africa.

Screwworm, supposedly introduced on infested sheep from South America, was detected in Libya during the spring of 1988 and identified by British entomologists in March of 1989. Countries adjacent to Libya were considered at immediate risk of infestation, and all African countries, with the exception of Namibia, Botswana, and South Africa, were considered at risk, if the infestation was not contained. Additionally, it was reported that southern and southeastern Europe and the Middle East (countries along the Mediterranean and Red Seas) were also at risk, if the infestation was not contained in Libya. In Libya, screwworm was present along an approximate 400 km strip in the coastal zone, extending west from within 20 km of the Tunisian border to Misurata in the east, an area of approximately 24,000 km.A multinational effort lead by the Food and Agriculture Organization (FAO) of the United Nations eradicated screwworm from Libya (officially declared screwworm-free 22 June 1992) using sterile insects flown from the sterile fly production plant of the Joint Mexico-U.S. Commission for the Eradication of Screwworm in Tuxtla Gutierrez, Chiapas, Mexico. Estimated cost of the eradication program was $31 million. Screwworm was eradicated from Libya approximately one year ahead of schedule and $2 million dollars under budget.

Intense surveillance programs have been part of the reason for the success of the screwworm eradication program. However, many years have passed since U.S. livestock producers have dealt with screwworm as part of their management operations. Complacency and a generation of producers without hands-on experience with screwworm infestations characterizes the current situation in the United States. Introductions into Libya and Australia (via an infested tourist who visited South America) and multiple reintroductions into the United States illustrate the potential for recurrence of this pest. It is essential that surveillance to prevent reintroduction into previously eradicated areas must continue as long as native screwworm populations exist in South America and the Caribbean.

Needs for Integrated Pest Management.
(1) Continued research to more effectively integrate the use of the sterile insect technique, insecticide use, and animal husbandry practices to optimize efficiency of control and eradication efforts will be required.
(2) Since eradication of screwworm is ongoing, no specific implementation plans are identified. The screwworm eradication programs continue to use the elements of an ongoing IPM program (e.g., sterile insect release, livestock management, and chemical control).

Research Needs.

(1) Basic Research.

(a) Continued research in Central America and in the proposed barrier zone of Panama will be required to support the eradication program progressing southward and to establish and maintain the barrier zone. To determine population dynamics, responses to biotic and abiotic factors, longevity, reproductive capacity, host reservoirs and dispersal and migration of adults, habitats will be examined as a factor in measuring population densities and responses to efficacy of trapping. Wild animal reservoirs will be identified through the ELISA technique where blood proteins from animal blood meals will be dissected from the fly's digestive tract and tested against known mammalian and avian samples. Laboratory and field studies of remote sensing technology and collection of ground truth data and correlation with ecological studies and predictive modeling will be carried out in habitats in Panama and possibly other Central American countries.

(b) Continued research will be required to genetically characterize screwworm cultures derived from different geographical areas and assess the genetic compatibility of the cultures. Identify quantitative genetic traits of cultures and determine heritability and genetic influence on expression and control of the traits. Use established methods and develop new laboratory parameters for assessing the degree of laboratory adaptation, such as changes in mating behavior, rate of reproductive development, and other heritable traits to study the genetic factors which control the traits.

(c) Continued research will be required to improve screwworm diet and rearing techniques to increase yield and quality of screwworms in the production plant.

(2) Control Components Research.

(a) Continued research will be required to provide data on the possibility of varying release sterile fly release rates by habitat and season in order to reduce costs in the eradication program and maintenance of the future barrier zone.

(b) Continued research will be required to provide protocol and objectives for outbreak situations where screwworm has been reintroduced into previously eradicated or introduced into nonendemic areas. Data on more efficient methods of controlling outbreak populations and identification of geographic source of outbreak populations are needed by the eradication program.

(c) Develop improved surveillance and detection techniques using remote sensing technology and Geographic Information Systems. Incorporate these techniques with screwworm population modeling and estimation efforts for use in eradication, barrier zone, and outbreak area situations to provide a more efficient and economical methods of application of sterile flies.

(d) Continued development of new strains for mass rearing in the production plant. Also, various methods of selecting and maintaining broodstocks will be evaluated for maintaining genetic variability while maintaining suitable production characteristics.

REFERENCES CITED

Brenner, R. J. 1985. Distribution of screwworms (Diptera: Calliphoridae) relative to land use and topography in the humid tropics of southern Mexico. Ann. Entomol. Soc. Amer. 78: 433-439.

Graham, O. H. 1985. Symposium on eradication of the screwworm from the United States and Mexico. Misc. Pub. Entomol. Soc. Amer. 62, 68 pp.

Mackley, J. W. 1986. Incidence of screwworm, *Cochliomyia hominivorax*, and the secondary screwworm, *C. macellaria* (Diptera: Calliphoridae), in the central highlands of Chiapas, Mexico.

Mackley, J. W. and H. E. Brown. 1984. Swormlure-4: A new formulation of swormlure-2 mixture as an attractant for adult screwworm, *Cochliomyia hominivorax* (Diptera: Calliphoridae). J. Econ. Entomol. 77: 1264-1268.

Mangan, R. L. 1985. Reproductive responses of native screwworm (*Cochliomyia hominivorax* [Coq.]) populations to sterile fly release. Proc. Entomol. Soc. Wash. 87: 717-739.

Parker, F. D. and J. B. Welch. 1991. Field comparisons of attractants for the screwworm fly (Diptera: Calliphoridae) in a tropical dry forest in Costa Rica. J. Econ. Entomol. 84: 1189-1195.

Parker, F. D. and J. B. Welch. 1992. Monitoring adult populations of the screwworm (Diptera: Calliphoridae) with feeding stations baited with liver. J. Econ. Entomol. 85: 1740-1753.

Thomas, D. B. and R. L. Mangan. 1992. Persistence of sterile screwworm (Diptera: Calliphoridae) flies at a release site. J. Econ. Entomol 85: 441-444.

Welch, J. B. 1988. Effect of trap placement on detection of *Cochliomyia hominivorax* (Diptera: Calliphoridae). J. Econ. Entomol. 81: 241-245.

Welch, J. B. 1990. A detector dog for screwworms (Diptera: Calliphoridae). J. Econ. Entomol. 83: 1932-1934.

****
****

End of Range Beef Cattle section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****
****

CONFINED BEEF CATTLE SUMMARY

1. The number of cattle marketed from the 13 leading cattle feeder states during 1992 was 22 million.

2. The yearly total value of confined beef in the 13 leading feeder states during 1992 was $17.6 billion.

3. The major insect pests of confined beef are the stable fly and house fly.

4. There are three minor pests: cattle grubs, lice, and scabies.

5. Four major areas identified as research needs for the major pests are: the determination of larval development sites; migration/distribution of adults; biological control (this includes foreign exploration); further definition of pest management strategies that rely less on insecticides but include sanitation, feedlot design, manure management (emphasis should be placed on area wide pest control).

6. Extension needs were related to research needs and include: enhance information on the economic significance of flies to confined cattle; refine techniques to sample adult and immature stable flies and house flies; improve pest-host computer models and; continue Pilot Tests and other research that enhances area wide control.

7. Related major problems also outlined included: manure management; water management; public perception; insecticides; loss of extension and research personnel; and others.

CONFINED BEEF CATTLE

Committee Members

Dr. John B. Campbell, Chair
Dr. Ivan L. Berry
Dr. Jan Chirico
Dr. Bill C. Clymer
Dr. Gerald L. Greene
Dr. Robert D. Hall
Dr. Steven R. Skoda
Dr. Gustave D. Thomas
Dr. Richard A. Weinzierl
Dr. John B. Welch

Economic Significance of Confined Beef

The number of fed cattle marketed from the thirteen major feedlot states in 1992 was 22 million: the total yearly value of confined beef in these states was $17.6 billion (Neb. Cattlemen 1993). The top thirteen feeder states that year, in descending order, were Texas, Nebraska, Kansas, Colorado, Iowa, Oklahoma, Idaho, California, South Dakota, Washington, Illinois, Minnesota, and Arizona.

Arthropod Pests of Confined Cattle

The two primary pests of beef cattle in feedlots are stable flies and house flies. Three secondary pests are cattle grubs, lice and scabies mites. The stable fly and house fly will be discussed in detail. The secondary pests will be briefly discussed.

Stable Fly, *Stomoxys calcitrans* (L.), and House Fly, *Musca domestica* L.

These pests will be considered together because their habitat, biology and behavior are similar and control measures are generally effective for both species (Morgan et al. 1983, West & Peters 1973).

Description and Biology. The stable fly and house fly are typical of the muscoid Diptera. The stable fly has piercing-sucking mouthparts, and both sexes feed on blood. The house fly has sponging-type mouthparts and feeds on a wide range of organic wastes, including mucous secretions and wounds of animals.

Both species breed in decaying organic matter in and around feedlots consisting of soil, manure and/or feed mixed with moisture. House flies may also breed in fresh manure. The stable fly is more restricted than the house fly, as far as confined beef animals are concerned, because it does not breed in fresh manure. Feeder areas in California, Oklahoma, Texas, New Mexico, Arizona, parts of eastern Colorado, Wyoming, Western Kansas, and Nebraska may be too dry most years to support stable fly breeding.

Females of either species may deposit several hundred eggs in their lifetime. The eggs hatch under summer conditions in 12-24 hr dependent upon temperature, Lysyk (1998) and species. The house fly can develop from egg to adult in 15-16 days. The stable fly takes about a week longer than the house fly. Adults may live about two weeks in nature.

Economic Importance. The economics of stable fly infestations on feeder cattle, in terms of weight loss and feed efficiency, are fairly well documented (Berry et al. 1983, Campbell et al. 1987). In all likelihood, the effect of stable fly feeding on weight gains is dependent on an accumulation of stress factors.

Stable fly feeding on the front legs of cattle cause cattle to bunch as each animal tries to protect its front legs from fly attack. If the bunching occurs during hot weather it will increase the effects of heat stress (Wieman et al. 1992). Research with low population levels of stable flies indicates that, in a situation with both stable flies and heat stress, the direct effect of stable flies accounts for 28.5% of the weight gain reduction (Catangui et al. 1993). These losses are fairly predictable and can be modeled with a high degree of accuracy.

Research with house fly populations of 49 per animal showed no effect on feeder cattle (Campbell et al. 1981). The economic effects of house flies are complicated because of the potential of the fly to transmit diseases and the increasing threat of nuisance lawsuits (Thomas & Skoda 1993).

An economic evaluation of the stable fly and house fly must, of course, consider the reduction of feed efficiency and weight loss (Campbell & Berry 1989). The commercial feeder also must consider the aesthetic value of fly control in terms of potential feeder customers. Also, the cost of control that will keep fly populations below an acceptable level must be included. In a well-managed production system, control costs include waste management as well as an insecticide program. A cost benefit analysis for sanitation (animal waste management) is difficult. Consideration must be given to the cost of labor, equipment, fuel, and so forth, on the debit side. The difficult aspect is to assign value to waste as fertilizer, increased animal comfort, and increased animal production. If modification of the cattle holding facilities, such as mound improvement, better drainage, improvements in debris basins and holding ponds, and so forth, are instituted, the cost should be prorated over the number of years the modifications last.

Insecticide costs may have little relationship to fly populations. In feedlots where the most insecticide is used, there may be fly levels above the economic threshold. The benefit of the insecticide is dependent on the insecticide system used and the degree of sanitation and animal management employed in conjunction with the insecticide.

Methods of Control.

Chemical. Insecticides most commonly employed at confined beef units are applied as space (knockdown) sprays. These are generally applied with tractor-mounted power takeoff units. They have squirrel cage fans which create an air blast that dispenses particles of insecticide. There are methods such as aircraft, foggers, and self-powered units that do basically the same thing.

Other chemical control methods include: (a) use of residual sprays applied to fly resting surfaces, more commonly used at small lots; (b) baits, which are not effective on stable fly and questionable for house fly population management; (c) larvicides, not widely used in feeder areas; and (d) feed additives, not effective on stable flies and only possibly effective on house flies in drier areas where most breeding is in fresh manure (Mock & Greene 1989).

Cultural. Sanitation is the main non-chemical method of fly reduction employed. It is not employed as extensively as it could be for reasons including: (a) much labor and equipment is required to do an adequate job; (b) crop production takes priority; (c) there is no place to put the manure after crops have emerged; (d) lack of knowledge on economics of flies on cattle; and (e) reliance on pesticides. Thomas et al. (1996) determined that sanitation decreased stable fly numbers by 50.9% and 36.2% respectively in a two-year study. The authors indicated that had they been able to get the feedlots cleaned earlier the first time (June 20 and June 29), that stable fly reductions would have been even greater.

Several types of traps have been used to reduce fly populations in feedlots (Pickens et al. 1994). Guo et al. (1998) used alsynite traps to obtain a population profile of stable flies moving in a feedlot area. Schofield (1998) evaluated electrified target traps baited with different colors and designs for trapping stomoxys spp. and the traps were evaluated with or without Co2 releases. Further research of this nature is needed on trap use (trap type, placement, numbers and other factors).

Environmental modifications such as sanitation, stocking rates and mechanical modification of breeding areas will reduce fly breeding (Campbell & Thomas 1993). The use and value of feedlot wastes for fertilizer, methane gas production, and cattle feed is being explored.

Biological. Several species of fly parasites have been sold to feedlot managers with undocumented benefits. Releases of one naturally occurring species have shown up to 50% stable fly reduction and has doubled pupal parasitism in Kansas (Greene 1990). Similar releases in Illinois (Weinzierl & Jones 1998) and Nebraska (Andress & Campbell 1994) have been less successful and too costly for implementation. As with insecticides adequate sanitation will be required if parasite releases are to be successful.

In recent years several studies have shown predators have an impact on fly populations, but to date have not been manipulated to control flies (Hall et al. 1989).

There are some pathogenic agents that infect muscoid flies (Rutz & Patterson 1990), but none have proven effective against stable flies and house flies.

Genetic. Genetic control shows promise for the future but considerably more research on stable flies and house flies is needed. Examples include sterile male techniques and genetic loading (Steiner et al. 1982).

Host Resistance. Research by Steelman et al. (1991) has shown resistance of cattle to the horn fly. Studies by Catangui et al. (1993, 1995) indicate that Brahma X English Continental crossbred calves may be slightly tolerant to stable flies while in the growing mode but not in the finishing stage of feeding. Overall, the Brahma crossbreds had as much reduction in weight gain and feed efficiency due to stable flies as did the English X Continental crossbreds and were 10-15% lower in weight gain and feed efficiency performance in the absence of stable flies. As was the case in the horn fly research of Steelman et al. (1991) some animals in either crossbred group always had more flies feeding on them than others. It is therefore possible that this mechanism could be used in the future for the stable fly but much more research is needed.

Although the development of vaccines against muscoid flies has not been documented, further research may show it is possible (Yon, 1992).

Cattle Grubs

In the past, cattle grubs (also called heel flies as adults) have been considered one of the most important pests of cattle. The "grub", or larva, of the heel fly spends about 8 months as an internal parasite. Eventually it migrates to the loin area of the back, completes larval development, cuts a breathing hole in the skin from which it exits, falls to the ground, and pupates. The greatest economic impact from grubs is at slaughter. Trim loss and reduced value of hides from grubby cattle usually results in the packer reducing the price 5 cents or more per pound. Other losses include weight gain reduction in the growing animal.

The development of systemic insecticides provided a method of efficient and cheap grub control. The newer biologicals, such as Ivermectin®, may not be as inexpensive but they are very effective.

Scabies

A skin condition caused by mites, scabies has been a periodic problem since the early 1900's and is the only livestock pest in the U.S. that has been the subject of federal legislation. In the years between 1970 and about 1987, scabies outbreaks were common in the Great Plains. At that time, toxaphene was the treatment of choice. However, EPA has restricted toxaphene use. This probably hastened the registration of Ivermectin®, which is very effective for the control of scabies.

There is, of course, no economic threshold for scabies because of the law. Scabies has ceased to be a problem since Ivermectin® has been registered for its control. It may be that, for all practical purposes, this pest has been eradicated.

Cattle Lice

There are four species of cattle lice of major importance to cattle. Three of these are blood feeding and the fourth feeds on skin debris. Their life cycles are similar in that the entire life cycle is spent on the animal. The reproductive rate of lice usually increases in the winter and declines in the summer.

The economics of cattle lice have been studied to some degree but not extensively. It generally takes a moderate population to impact cattle weight gains. However, old or young animals with heavy lice populations are more susceptible to respiratory diseases and may die during stress periods from severe winters. Death may be from a respiratory disease but the underlying cause was probably the stress caused by the lice.

Summary of Research and Extension Needs

Research Needs

1) Refine/develop information on the relative contributions of various larval development sites (in livestock facilities and elsewhere) and dispersal (long distance and short range) to house fly and stable fly populations in feedlots and nearby urban areas.

2) Continue to investigate biological control. Points should include foreign exploration and identification of candidate predators, parasitoids, and pathogens; improved methods of assessing natural enemy populations and their impacts on fly populations; optimization of indigenous parasites (release methods and release rates); the use of molecular genetics to "improve" the impacts of natural enemies, especially pathogens.

3) Develop/refine preventive pest management practices that rely on least-toxic and still economically feasible practices to limit fly populations. These practices include feedlot design, sanitation, manure management, and the use of insecticides and application methods that present reduced risks to human health and the environment. Expert systems and modelling will be beneficial additions to prioritizing and developing research needs and impacts.

4) Initiate area-wide pest management programs. These can be improved/modified as more information becomes available from the previous 3 points.

Extension Needs

1) Information is available for economic injury level for the stable fly in Nebraska. However, this information needs to be developed in other areas of the country because of the interaction of temperature and humidity. This is because much of the existing work was done in Nebraska where there is low humidity. Little work has been done on the house fly (Campbell et al. 1981) and more should be done. House fly is often incriminated in disease transmission but no information is available under feedlot conditions.

2) Studies have shown the relationship between the three major sampling methods of stable fly adults (Thomas et al. 1989). Further studies may be needed to correlate this to prediction models. Also, more work on sampling adult house flies is needed. Studies have also shown (Meyer & Peterson 1983, Skoda et al. 1991) how to best predict immature levels of stable fly in feedlots but more work is needed to correlate this to adult levels. Similar needs exist for immature house flies (Skoda et al 1993).

3) There are models of the hosts; some attempts to combine the economic information available for the stable fly with these host models have been made (Feddes et al. 1985) and were very efficient. More refinement is needed for the stable fly and nothing has been done with the house fly.

4) Nebraska has completed two pilot projects of IPM in feedlots. Other pilot projects are underway in Kansas and Illinois. Further work and refinement so to implement area-wide control should be implemented.

Related Major Problems

1) Manure management as it relates to the possibility of nuisance litigation (flies, dusts and odors), pollutants (nitrates and phosphates), composting procedures and their potential benefits (mechanical alterations).

2) Water management relating to retention ponds, diversions, pollution, possible insect breeding, and the use of bacteria to reduce odors.

3) Improving the public's perception of confined beef operations. This will include improving education, aesthetics, and tolerance.

4) Immature fly development in urban settings including animal hobbyists (dogs, horses, etc.) and composting. Flies in the urban environment can become a major source for litigation against commercial feedlots and confined swine or poultry units.

5) Livestock insecticides are a minor component of the pesticide industry. Therefore, companies are unable to invest because of the large costs to develop new or reregister existing insecticides. Changing the public's perception, through education, of the cost-benefit of judicial insecticide use is a part of this problem. Withdrawal periods for drugs of all types, assuring safety, are now law (sometimes unreasonably low tolerance of chemicals are in place).

6) Insecticide resistance is a major problem, particularly with so few chemicals registered for use in confined animal facilities. This is exacerbated because few new chemicals are produced for use with livestock. If used correctly, chemicals may be a major component of a pest management system.

7) Large hay bales contribute significantly to fly breeding in pastures. This contributes to stable fly problems; this contribution is to problems of flies in the feedlots, particularly when pastures adjoin.

8) Testimonials instead of reliable research data are used to support parasites for fly control and bacteria for odor control.

9) Relating fly problems to the entire economics of confined animal feeding operations. This includes such things as backgrounding vs finishing, the farmer feeder vs commercial feeders, the effect of animal rights groups, legislation and the farm bill, "romantic image" of the farm, the trend toward large feeders (less than 200 feedlots marketed over 1/2 of the fed beef in 1989 [Nebraska Farmer 1990]).

10) There has been a reduction in the number of "traditional" County Extension Personnel. This has contributed to the reduction of overall extension education, partly because of lack of personnel and partly due to redirection (educate pesticide applicators for restricted use chemicals or prescription use of pesticides).

References Cited

Andress, E.R. & J.B. Campbell. 1994. Inundative releases of pteromalid parasitoids (Hymenoptera: Pteromalidae) for control of stable flies, *Stomoxys calcitrans* (L.), (Diptera:Muscidae) at confined cattle installations in west central Nebraska. J. Econ. Entomol. 87:714-722.

Berry, I. L., D. A. Stage & J. B. Campbell. 1983. Populations and economic impacts of stable flies on cattle. Trans. Am. Soc. Ag. Eng. 26: 873-877.

Campbell, J. B. & I. L. Berry. 1989. Economic threshold for stable flies on confined livestock, pp. 18-22. *In* J. J. Petersen & G. L. Greene (eds.), Current status of stable fly (Diptera: Muscidae) research. Misc. Pub. No. 74, Entomol. Soc. Am., College Park, MD.

Campbell, J. B., I. L. Berry, D. J. Boxler, R. L. Davis, D. C. Clanton & G. H. Deutscher. 1987. Effects of stable flies (Diptera: Muscidae) on weight gain and feed efficiency of feedlot cattle. J. Econ. Entomol. 80: 117-119.

Campbell, J. B., D. J. Boxler, J. I. Shugart, D. C. Clanton & R. Crookshank. 1981. Effects of house flies on weight gains and feed efficiency of yearling heifers on finishing rations. J. Econ. Entomol. 74: 94-95.

Campbell, J. B. & G. D. Thomas. 1993. Control of flies at confined production units. Agri-practice 14: 38-45.

Catangui, M. A., J. B. Campbell, G. D. Thomas & D. J. Boxler. 1993. Average daily gains of Brahman-crossbred and English X exotic feeder heifers exposed to low, medium, and high levels of stable flies (Diptera: Muscidae). J. Econ. Entomol. 86: 1144-1150.

Catangui, M.A., J.B. Campbell, G.D. Thomas & D.J. Boxler. 1995. Average daily gains of Brahman-crossbred and English X exotic feeder heifers during long-term exposure to stable flies (Diptera: Muscidae). J. Econ. Entomol. 1349-1352.

Feddes, J. J. R., J. A. DeShazer, I. L. Berry & J. B. Campbell. 1985. Modelling effects of stable flies on beef cattle. Am. Soc. Agric. Eng. Tech. Paper 85-4007.

Greene, G. L. 1990. Biological control of filth flies in confined cattle feedlots using pteromalid parasites, pp. 29-42. *In* D. A. Rutz & R. S. Patterson (eds.), Biocontrol of arthropods affecting livestock and poultry. Westview Press, Boulder, CO.

Guo, Y.J., G.L. Green & M.D. Butine. 1998. Population profile of stable flies (Diptera: Muscidae) caught on alsymite traps in various feedlot habitats. J. Econ. Entomol. 91: 159-164.

Hall, R. D., J. P. Smith & G. D. Thomas. 1989. Effect of predatory arthropods on the survival of immature stable flies (Diptera: Muscidae), pp. 33-40. *In* J. J. Petersen & G. L. Greene (eds.), Current status of stable fly (Diptera: Muscidae) research. Misc. Pub. No. 74, Entomol. Soc. Am., College Park, MD.

Lysyk, T.J. 1998. Relationships between temperature and life-history parameters of *Stomoxys calcitrans* (Diptera: Muscidae). J. Med. Entomol. 35: 107-119.

Meyer, J. A. & J. J. Petersen. 1983. Characterization and seasonal distribution of breeding sites of stable flies and house flies (Diptera: Muscidae) on eastern Nebraska feedlots and dairies. J. Econ. Entomol. 76: 103-108.

Morgan, C. E., G. D. Thomas & R. D. Hall. 1983. Annotated bibliography of the stable fly, *Stomoxys calcitrans* (L.), including references on other species bolonging to the genus *Stomoxys*. North Central Reg. Res. Pub. No. 291, Univ. of Missouri Research Bulletin No. 1049, Columbia, MO.

Mock, D. E. & G. L. Greene. 1989. Current approaches to chemical control of stable flies, pp. 46-53. *In* J. J. Petersen & G. L. Greene (eds.), Current status of stable fly (Diptera: Muscidae) research. Misc. Pub. No. 74, Entomol. Soc. Am., College Park, MD.

Nebraska Cattlemen. 1993. Beef facts. Nebraska Cattlemen 49: 32-35.

Nebraska Farmer. 1990. 200 feedlots finish half of this country's beef cattle. Nebraska Farmer 132: 52-53.

Pickens L. G., E. T. Schmidtmann & R. W. Miller. 1994. How to control house and stable flies without using pesticides. USDA-ARS Agriculture Information Bulletin No. 673.

Rutz, D. A. & R. S. Patterson (eds.). 1990. Biocontrol of arthropods affecting livestock and poultry. Westview Press, Boulder, CO.

Schofield, S. 1998. Responses to electrified targets and daily activity of *Stomoxys. spp.* (Diptera: Muscidae) in Zimbabwe. Bull. Entomol. Res. 627-632.

Skoda, S. R., G. D. Thomas & J. B. Campbell. 1991. Developmental sites and relative abundance of immature stages of the stable fly (Diptera: Muscidae) in beef cattle feedlot pens in eastern Nebraska. J. Econ. Entomol. 84: 191-197.

Skoda, S. R., G. D. Thomas & J. B. Campbell. 1993. Abundance of immature stages of the house fly (Diptera: Muscidae) from five areas in beef cattle feedlot pens. J. Econ. Entomol. 86: 455-461.

Steelman, C. D., A. H. Brown Jr., E. E. Gbur & G. Tolley. 1991. Interactive response of the horn fly (Diptera: Muscidae) and selected breeds of beef cattle. J. Econ. Entomol. 84: 1275-1282.

Steiner, W. W., W. J. Tabachnick, K. S. Rai & S. Narang (eds.). 1982. Recent developments in the genetics of insect disease vectors. Stipes Pub. Co., Champaign, IL.

Thomas, G. D., I. L. Berry, D. R. Berkebile & S. R. Skoda. 1989. Comparison of three sampling methods for estimating adult stable fly (Diptera: Muscidae) populations. Environ. Entomol. 18: 513-520.

Thomas, G. D. & S. R. Skoda (eds.). 1993. Rural flies in the urban environment? North Central Reg. Res. Bull. No. 335, Univ. of Nebraska Agricultural Res. Div., Institute of Agriculture and Natural Resources Res. Bull. No. 317, Lincoln, NE.

Thomas, G.D., S.R. Skoda, D.R. Berkebile & J.B. Campbell. 1996. Scheduled sanitation to reduce stable fly (Diptera: Muscidae) populations in beef cattle feedlots. J. Econ. Entomol. 411-414.

Weinzierl, R.A. & C.J. Jones. 1998. Releases of Spalangia nigroaena and Muscidifurax zaraptor (Hymenoptera: pteromalidae) increase rates of parasitism and total mortality of stable fly and house fly (Diptera: Muscidae) pupae in Illinois cattle feedlots. J. Econ. Entomol. 91: 1114-1121.

West, L. S. & O. B. Peters. 1973. An annotated bibliography of Musca domestica Linnaeus. Dawsons of Pall Mall, London.

Wieman, G. A., J. B. Campbell, J. A. DeShazer & I. L. Berry. 1992. Effects of stable flies (Diptera: Muscidae) and heat stress on weight gain and feed efficiency of feeder cattle. J. Econ. Entomol. 85: 1835-1842.

Yong, W. K. (ed.). 1992. Animal parasite control utilizing biotechnology. CRC Press, Boca Raton, FL.

****

End of Confined Beef Cattle section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****

SWINE SUMMARY

1. The total U.S. swine inventory was estimated at 62,156,000 head as of December 1, 1998. The total value of this inventory was estimated at $4,962,403,000.

2. Ten states account for 80% of the total swine inventory. These states include Illinois, Indiana, Iowa, Kansas, Minnesota, Missouri, Nebraska, North Carolina, Ohio, and South Dakota.

3. The major arthropod pests of swine are the sarcoptic mange mite, hog louse, house fly, and stable fly.

4. Major areas identified as research needs for sarcoptic mange mites and lice are: the development of standardized, reliable, and efficient sampling methodologies; a comprehensive understanding of mite and lice biology and ecology as it relates to varying production systems and environments; further determination of the economic importance

of mites and lice as it relates to different production systems, disease transmission, and the presence of other external or internal parasites and; the development of alternative control methodologies which could fit into an integrated pest management program.

5. Major areas identified as research needs for house flies and stable flies are: determining the effects of varying production systems and waste handling systems on fly dispersal, population dynamics, and behavior; the development of standardized sampling methodologies; a more comprehensive determination of the species composition, biology, and ecology of naturally occurring biological control agents; determining the true relevance of house flies and stable flies as a nuisance and public health hazard; determining the role of house flies and stable flies in the transmission of swine diseases and; the development of alternative control methodologies which could fit into an integrated pest management program.

Extension needs are related to research needs and include: the development of effective and efficient information delivery systems; evaluating the acceptance of implemented pest management programs and; providing feedback as to additional research needs or special problems unique to a geographical area.

SWINE

Committee Members

Dr. Ken H. Holscher, Chair
Dr. Ralph E. Williams
Dr. Cliff E. Hoelscher
Dr. Gene R. Strother
Dr. Darrell E. Bay
Dr. John L. Riner
Dr. William F. Lyon
Dr. Roger D. Moon
Dr. Ib Hagsten

Economic Significance of Swine

Pork production is an integral and important facet of the U.S. livestock industry. According to the National Agriculture Statistics Service, the total U.S. swine inventory was estimated at 62,156,000 head as of December 1, 1998 (Anonymous 1998). The total value of this inventory was estimated at $4,962,403,000. While swine are raised in all 50 states, 10 states account for 80% of the total swine inventory. These states include Illinois, Indiana, Iowa, Kansas, Minnesota, Missouri, Nebraska, North Carolina, Ohio, and Oklahoma.

Arthropod Pests of Swine

The U.S. swine industry has undergone significant changes over the last decade. The trend towards total confinement and vertical integration has greatly impacted the manner in which swine are raised. Today, more and more animals are being produced on fewer and fewer facilities. This trend has not only had a significant impact on our pork production system, but on the arthropod pests associated with pork production as well.

The arthropod pest complex associated with pork production consists of two distinct groups. One group consists of the "on-host" pests that directly attack and infest swine. Of primary importance in this group are the sarcoptic mange mite and the hog louse. These pests are unique to the swine industry and as such pose unique problems and challenges in the development of IPM programs. The other group consists of the "off-host" pests that are directly associated with swine facilities. Of primary importance in this group are the house fly, stable fly, and other associated Diptera. These pests are not unique to pork production but are a problem and concern throughout the entire livestock industry. As such, IPM strategies to combat these pests in swine facilities may be adapted from IPM programs developed for other livestock commodities.

Sarcoptic Mange Mite, *Sarcoptes scabiei*

Description and Biology. Sarcoptic mange mites are tiny parasitic mites that live in threadlike feeding tunnels in the epidermal layer of the host animal. These tunnels are enlarged up to 5 mm per day and are constructed with the aid of digestive enzymes that dissolve the host animal's tissue. Mating generally takes place on the skin surface. After mating, newly fertilized females construct new feeding tunnels in which they lay 1 to 4 eggs per day over a period of about 2 weeks. These eggs hatch into larvae in about 3 to 5 days, although some egg hatch may be delayed as long as 20 days. Larvae continue to expand the feeding tunnels and develop into nymphs in about 2 to 4 days. Nymphal mites also feed and expand the tunnels before developing into adult mites 4 to 6 days later. The entire life cycle of the sarcoptic mange mite takes place on the host animal and can be completed in as little as 10 to 14 days.

Sarcoptic mange mites are host specific and spend their entire life cycle on the animal. Therefore, transmission of mites is primarily the result of direct contact with infested pigs. It is theorized that mites may be able to survive off the animal for several days under ideal conditions. This could possibly result in mite transmission without direct animal contact. However, pig-to-pig contact is the major means of sarcoptic mange mite transmission.

The feeding activity of sarcoptic mange mites causes a tremendous amount of irritation. Infested animals scratch vigorously and rub against objects in an effort to relieve the discomfort. Most often, but not always, an infestation begins on the inner side of the ear before spreading over the head along the neck, and across the body. The skin of infested animals may become inflamed, cracked and thickened, and encrusted lesions may develop in the ears and on the face. Extensive hair loss may also result. The activity of these mites increases as skin is warmed by fever or high environmental temperature. This increases mite feeding rates and irritation and may intensify clinical signs in affected pigs. Mites can be found on pigs throughout the year but are most likely to cause noticeable problems during the winter. Winter adds an additional stress to animals and, in many cases, causes pigs to bunch together thus allowing mites to spread rapidly throughout the herd. In summer, pigs are less likely to bunch together thereby reducing pig-to-pig contact and subsequent mite transmission.

Economic Importance. The presence of sarcoptic mange mites was listed as the most prevalent condition noted by the Minnesota Food Animal Disease Reporting System in their ranking of the 20 most prevalent diseases and syndromes in swine during the period of 1974-1980 (Diesch et al. 1982). Results from the North American National Health Monitoring System (NAHMS) indicated that 50% of the swine herds had moderate to severe skin lesions indicative of sarcoptic mange mite infestations while an additional 30% of the swine herds had mild lesions (Miller 1991). A 1987 survey of swine in Indiana indicated that 25.3% of all hogs surveyed were infested with sarcoptic mange mites while 36.2% of all farms surveyed had mite-infested pigs on the premises (Wooten-Saadi et al. 1987a).

Relatively few studies have been conducted regarding the economic implications of sarcoptic mange mite infestations in swine. While the results of these studies have been somewhat variable, most report significant improvements in animal performance in the absence of mite infestations. Cargill & Dobson (1979), Alva Valdes et al. (1986), and Dalton & Ryan (1988) reported average weight gain reductions ranging from 3.3% to 9.0% in mite-infested growing pigs. Cargill & Dobson (1979) and Dalton & Ryan (1988) also reported reduced feed efficiencies of 3.1% and 9.0% respectively in growing pigs. In a 15-month trial conducted in a four state area, Gaafar et al. (1986) demonstrated a 12 lb per pig increase in weight gain, a 0.1 lb decrease in feed-to-gain, and 0.6 additional pigs weaned per litter in animals subjected to an integrated mange control program. In the only in-depth study conducted on the impact of sarcoptic mange mites on sow performance, Arends et al. (1990) reported that sows treated for sarcoptic mange mites consumed 4.3 lb less feed per weaned piglet and had litter weights that were 9.13 lb heavier than those of untreated sows. In addition, piglets weaned from treated sows were 12.76 lb per head heavier at slaughter and had a 0.11 lb per day superior average daily gain. Using these findings, sarcoptic mange mite infestations could result in annual losses ranging from $84 to $115 per sow.

The aforementioned studies indicate the potential economic impact of sarcoptic mange mite infestations on pork production. These studies, however, do not address the classical definition of economic injury level since measured changes in animal performance were not directly correlated to graduated differences in mite densities or infestation levels. Rather, these studies measured changes in animal performance in the total presence or absence of mites. It is therefore clearly evident that more intensive research is needed before a precise economic injury level can be determined. This research must take into account differences in age, breed, host immunity, management and housing systems, nutritional programs, the presence of other external or internal parasites, climatic conditions, and other production variables that could affect animal performance and subsequent injury level determinations.

Current Methods of Control and Research Needs

Chemical. The most commonly used and advanced technology to control sarcoptic mange mites in swine is the direct application of ectoparasiticide compounds to target animals. Six ectoparasiticide active ingredients are currently registered by EPA/FDA for use on swine to control sarcoptic mange mites. Amitraz, malathion, permethrin, and phosmet are all approved for use as a direct animal spray. Doramectin is approved for use as an injectable while ivermectin is approved for use in both injectable and feed additive formulations.

Ectoparasiticide application is typically recommended on a whole-herd basis when initiating a mange mite control program. Since none of the currently available ectoparasiticide products are effective against the egg stage of the sarcoptic mange mite, a second whole-herd treatment application is recommended 10 to 14 days later to assure a complete break in the mite life cycle. These initial whole-herd treatments can be effective in reducing mite populations to extremely low levels. Unfortunately, these treatments may not completely eliminate a mange problem. Therefore, an ongoing preventive management program consisting of routine ectoparasiticide treatments is generally recommended. The specifics of the preventive management program vary according to the type of production system (i.e. farrow-to-finish, finishing, feeder pig production), product availability, product restrictions (i.e. slaughter intervals, age restrictions), availability of labor and equipment, climatic conditions, etc.

Cultural Control. Cultural or sanitation practices will not eliminate an existing sarcoptic mange mite infestation. However, cultural management practices can be an integral part of an overall management program aimed at preventing the establishment or reestablishment of mange mites within the operation. Specifically, one of the most important cultural management practices involves the isolation of all new stock for a minimum of 30 days. These new animals should be treated for mange mites with an approved ectoparasiticide product as they arrive, followed by a second treatment 10 to 14 days later. Any animals that still exhibit clinical signs of mange following completion of the isolation period should be culled before incorporating the remaining animals into the operation.

Although no published data exist, evidence suggests that sarcoptic mange mites are less likely to have a serious impact in healthy animals raised in a stress free environment. While good husbandry practices should not be viewed as a viable control option, they could aid in preventing or minimizing severe mange impact.

Biological Control, Host Resistance, IGR's. Alternative methodologies such as biological control, genetic control, host resistance, and the use of insect growth regulators are not available for the control of sarcoptic mange mites on swine.

Eradication. Eradication of the sarcoptic mange mite from the U.S. swine industry is not feasible. Eradication of mange mites within an individual operation may be accomplished through the use of intensive management practices and biosecurity measures. While this form of eradication may be feasible in foundation or seed stock breeding herds, it is not practical for commercial swine operations.

Regulatory Issues. The application of ectoparasiticides currently forms the basis of sarcoptic mange mite control in swine. As such, legal issues such as Rebuttable Presumption Against Registration (RPAR), Worker Protection Standards, the Delaney Clause, and the Food Quality Protection Act could limit the availability and continued use of these products. Local, state, or federal legislation regarding animal husbandry practices could also seriously impact sarcoptic mange mite problems and limit the use of currently available control tactics.

Sampling. Basic to the development and implementation of an effective pest management program is a sampling methodology that is reliable and can be quantified to establish economic injury levels and monitor the effectiveness of treatment or management programs. Current sarcoptic mange mite sampling methodologies in swine rely on the extraction of mites from skin scrapings that have been collected through the use of various physical tools or devices (i.e. wood chisel, bone curette, sharpened spoon, etc.). These skin scrapings are generally taken from a 1/2 square inch to 1 square inch area of the inside surface of the ear of suspect animals. While skin scrapings can be used to demonstrate the presence of mites, certain inherent limitations affect the reliability and usefulness of this sampling method in an integrated pest management program. Specifically, skin scrapings can be fairly reliable in determining the presence or absence of mites within a herd if a sufficient number of animals are sampled. However, this sampling method cannot be used with any degree of reliability to determine or estimate total mite densities within individual animals or within a herd. In addition, skin scrapings can be unreliable in determining initial or low grade infestations and in monitoring fluctuations in mite populations within individual animals.

The problem inherent in the skin scraping sampling method has lead to the development and use of other mange monitoring methodologies (Lee et al. 1980, Courtney et al. 1983, Martineau et al. 1984, Wooten et al. 1986, Wooten-Saadi et al. 1987b). Although somewhat variable, these methodologies employ a rating scale based on the extent of visual mange symptomology. Symptoms factored into these rating scales include pruritus, hypersensitivity, lesion development, and rubbing behavior. These methodologies, however, provide little insight into mite population dynamics since the extent of observed symptoms cannot be directly correlated to mite densities.

Biology and Ecology. Very little has been reported concerning the biology and ecology of sarcoptic mange mites on swine (Davis & Moon 1990). Unfortunately, these data are crucial to the development of an integrated pest management program.

Models. Computer simulation can be a valuable aid in the development of effective and efficient pest management programs. The ability to simulate the responses of a pest population to varying management strategies can provide a basis for an optimal decision-making process. Computer models of varying complexity have been developed to describe the population dynamics of crop and forest pests. However, little attention has been devoted to the development of such models for livestock pests. This is especially true in regard to sarcoptic mange mites in swine in which no modeling data is available. As research data is gathered on efficient sampling methodologies, mite biology and ecology, economic injury level determinations and control strategies an integrated pest management program could be designed and incorporated into a modeling system. The ultimate goal would be to develop practical implementation procedures that could be incorporated into a total swine health program. Emphasis must be given to the development of a flexible modeling system that would allow for adjustments based on facilities and management systems and the availability of control alternatives.

Hog Louse, *Haematopinus suis*

Description and Biology. The hog louse is the largest bloodsucking louse infesting domestic animals in the United States. Hog lice are host specific and are restricted to the skin surface of the hog where they feed on blood several times each day. Female lice glue their eggs to the hair shafts of the hog. These eggs hatch into nymphs in about 10 to 14 days, although in cool weather hatching may be extended up to 20 days. Nymphs have the same feeding habits as adult lice and resemble adults except that they are smaller in size. After undergoing several molts, nymphal lice develop into adults in about 10 to 14 days. The entire life cycle from egg to adult can be completed in about 2 to 3 weeks, depending on temperature.

Hog lice tend to feed in clumps during their development. Infestations generally start around the ears before expanding to lower body regions. Preferred feeding areas include the ears, the skin around the neck, folds in the skin, and the inside surface of the legs near the body. Hog lice are host specific and spend their entire life cycle on the animal. While lice can survive for several days in warm bedding, the primary method of transmission is direct contact with infested hogs.

Economic Importance. Hog lice are fairly common external parasites of swine in the United States. A 1987 survey of swine in Indiana indicated that 18.1% of all hogs surveyed were infested with lice while 51.5% of all farms surveyed had lice infested pigs on the premises (Wooten-Saadi et al. 1987a).

Little has been reported relating the economic importance of hog lice in the swine industry other than "estimates" or "guesses." In the only recent studies addressing this topic, Nickle & Danner (1979) and Davis & Williams (1986) investigated the impacts of hog lice on pig growth. Their data did not substantiate expectations that hog lice infestations result in reduced growth rate or feed efficiency. Hog lice have also been incriminated in the transmission of eperythrozoonosis (Von Heinritzi 1992), swine pox virus, and other swine diseases. However, the prevalence with which disease transmission occurs and the economic implications that may result have not been investigated.

It is clearly evident that more research is needed to deduce the economic significance of hog lice on animal performance. If research findings implicate hog lice as an economic factor, more intensive research will be needed to determine precise economic injury levels. Research efforts must take into account differences in age, breed, host immunity, management and housing systems, nutritional programs, the presence or absence of other external or internal parasites, climatic conditions, and other production variables that could affect animal performance and subsequent injury level determinations.

Current Methods of Control and Research Needs

Chemical. The most commonly used and advanced technology to control lice in swine is the direct application of ectoparasiticide compounds to target animals. Ten ectoparasiticide active ingredients are currently registered by the EPA/FDA for use on swine to control hog lice. Amitraz, methoxychlor, and permethrin are all approved for use as a direct animal spray while coumaphos, malathion, phosmet, and stirofos are approved for use in both spray and dust formulations. Fenthion is approved for use as a direct pour-on application. Doramectin is approved for use as an injectable while ivermectin is approved for use in both injectable and feed additive formulations.

Ectoparasiticide application is typically recommended on a whole-herd basis when initiating a hog lice control program. Since none of the currently available ectoparasiticide products are effective against the egg stage of the hog louse, a second whole-herd treatment application is recommended 10 to 14 days later to assure a complete break in the louse life cycle. These initial whole-herd treatments can be effective in reducing lice populations to extremely low levels. Unfortunately, these treatments may not completely eliminate a louse problem. Therefore, an ongoing preventive management program consisting of routine ectoparasiticide treatments is often recommended. The specifics of the preventive management program vary according to the type of production system (i.e. farrow-to-finish, finishing, feeder pig production), product availability, product restrictions (i.e. slaughter intervals, age restrictions), availability of labor and equipment, climatic conditions, etc.

Cultural Control. Cultural or sanitation practices will not eliminate an existing hog lice infestation. However, cultural management practices can be an integral part of an overall management program aimed at preventing the establishment or reestablishment of lice within the operation. Specifically, one of the most important cultural management practices involves the isolation of all new stock for a minimum of 30 days. These new animals should be treated for lice with an approved ectoparasiticide product as they arrive, followed by a second treatment 10 to 14 days later. Any animals that still exhibit lice following completion of the isolation period should be retreated or culled before incorporating the remaining animals into the operation.

Although no published data exists, evidence suggests that hog lice are less likely to have a serious impact in healthy animals raised in a stress free environment. While good husbandry practices should not be viewed as a viable control option, they could aid in preventing or minimizing the severity of impact.

Biological Control, Host Resistance, IGR's. Alternative methodologies such as biological control, genetic control, host resistance, and the use of insect growth regulators are not available for the control of lice on swine.

Eradication. Eradication of hog lice from the U.S. swine industry is not feasible. Eradication of hog lice within an individual operation may be accomplished through the use of intensive management practices and biosecurity measures. While this form of eradication may be feasible in foundation or seed stock breeding herds, it is not practical for commercial swine operations.

Regulatory Concerns. The application of ectoparasiticides currently forms the basis of hog lice control. As such, legal issues such as Rebuttable Presumption Against Registration (RPAR), Worker Protection Standards, the Delaney Clause, and the Food Quality Protection Act could limit the availability and continued use of these products. Local, state, or federal legislation regarding animal husbandry practices could also seriously impact hog lice problems and limit the use of currently available control tactics.

Sampling. Basic to the development and implementation of an effective pest management program is a sampling methodology that is reliable and can be quantified to establish economic injury levels and monitor the effectiveness of treatment or management programs. Current lice sampling methodologies in swine rely on making population estimates based on whole body counts or limited area counts, or in assigning infestation levels (i.e. light, moderate, heavy) based on predetermined index counts. These methodologies are subject to considerable variability. Difficulty often arises in making whole body counts due to an animal's size and color and to accumulations of dried mud or other debris that may adhere to the animal's hair coat. The difficulty with making limited area counts is that there is usually an unequal distribution of lice on the animal (Wooten-Saadi et al. 1987). While index counts provide a measure of infestation, they do not provide a precise measure of lice population densities.

Biology and Ecology. Very little has been reported concerning the biology and ecology of hog lice. Of the only recent study on the subject, Wooten-Saadi et al. (1987a) made observations of the on-host distribution of hog lice in their survey of swine in Indiana. Unfortunately, data on hog lice biology and ecology is crucial to the development of an integrated pest management program.

Models. Computer simulation can be a valuable aid in the development of effective and efficient pest management programs. The ability to simulate the responses of a pest population to varying management strategies can provide a basis for an optimal decision-making process. Computer models of varying complexity have been developed to describe the population dynamics of crop and forest pests. However, little attention has been devoted to the development of such models for livestock pests. This is especially true in regard to hog lice in which no modeling data is available. As research data is gathered on efficient sampling methodologies, hog lice biology and ecology, economic injury level determinations and control strategies an integrated pest management program could be designed and incorporated into a modeling system. The ultimate goal would be to develop practical implementation procedures that could be incorporated into a total swine health program. Emphasis must be given to the development of a flexible modeling system that would allow for adjustments based on facilities and management systems and the availability of control alternatives.

House Fly, *Musca domestica*, And Other Nonbiting Flies

Description and Biology. The house fly is the most common nonbiting fly species encountered in swine facilities. Female flies deposit their eggs in manure, spilled feed, or other decaying organic matter found in and around the facility. These eggs typically hatch in about 1 to 2 days whereupon the larvae burrow into and feed on the breeding material. The larvae complete their development in about 4 to 6 days and then seek a drier environment in which to pupate. Adults subsequently emerge about 5 to 6 days later. The entire life cycle can be completed in as little as 10 to 14 days, depending on temperature. In northern climates house flies are usually present from May through October with numerous generations being produced during this time period. However, in southern climates or in environmentally controlled swine confinement units, conditions may exist that would allow house flies to breed continually throughout the year.

Adult house flies have sponging mouthparts and feed on animal secretions, manure fluids, and other materials commonly found in and around the swine facility. When not feeding, adult house flies tend to rest on the ceiling, walls, roof supports, or other exposed surfaces. Adult flies congregate at night inside open buildings or on the outside of buildings under the eaves.

The trend towards total confinement systems has also created an environment conducive to the development of other nonbiting fly species. These include little house flies (*Fannia* species), dump flies (*Ophyra* species) and moth flies (family Psychodidae).

Economic Importance. The house fly is a cosmopolitan pest that can be encountered in all types of pork production systems. Little house flies and dump flies can also be encountered in all types of production systems but are most abundant in and around confinement housing. Moth flies are most prevalent in confinement facilities that utilize a liquid manure management system.

Direct economic losses in pork production resulting from house fly infestations have not been documented. However, the house fly has been incriminated in the transmission of hog cholera and is suspect in the transmission of other swine diseases such as pig scours. The house fly has also been incriminated in the transmission of numerous human diseases and is therefore considered to be a nuisance and public health hazard. As a result, lawsuits and threats of legal constraint have been instigated against livestock producers for failure to properly control house flies (Hayes 1993). This in itself could have serious economic implications and should be reason enough for pork producers to implement control measures against this pest.

Although house flies have not been shown to cause direct losses in animal performance, considerable expenditures in control costs targeted at this pest may in incurred by pork producers. For example, it has been estimated that over $20 million is spent annually on house fly control by pork producers in the North Central states (Campbell 1993). While these expenditures may not technically be considered "losses", they nonetheless constitute a significant investment above return.

The economic implications of little house flies, dump flies and moth flies in pork production systems have not been addressed. However, in large numbers these flies can constitute a problem due to their annoyance and nuisance behavior.

By definition, economic injury levels for house flies and other nonbiting flies cannot be determined since these pests have not been shown to cause direct economic losses in animal production or performance. A justification could be made, however, for the establishment of an intolerable nuisance level based on the number of flies dispersing from swine or other livestock or poultry facilities. Unfortunately, the scientific establishment of such levels is highly improbable since the tolerance for flies is likely to vary considerably from region to region, state to state, locality to locality, and individual to individual.

Current Methods of Control and Research Needs

Chemical. Current control measures for house flies and other nonbiting flies found in and around swine facilities include the application of conventional insecticides as residual surface sprays, space sprays, feed additives, larvicide sprays, or baits. Residual surface sprays are applied to fly resting areas, such as the framework of swine housing, ceilings, trusses, electric light wires and fixtures, outside walls and fences, and any other surfaces that may attract flies. These surface sprays are applied with hydraulic or compressed air sprayers to the point of runoff and are generally effective for 1 to 3 weeks or longer. Space sprays are applied with foggers or mist blowers around the swine facility to provide a quick knockdown of adult fly populations. These applications have little, if any, residual activity and may need to be reapplied at frequent intervals to obtain maximum effectiveness. Space sprays are most advantageous where an immediate reduction of an adult fly population is necessary. Feed additives function by passing through the animal's digestive system to kill fly larvae developing in the manure. As such, these materials are most effective where fresh manure is the sole or main fly breeding source. However, since these materials will not control existing adult flies or fly larvae developing in other breeding materials, supplemental controls may be necessary. Larvicide sprays applied to manure and other fly breeding materials may, at times, aid in the control of flies. While these sprays may be of benefit in certain situations, total reliance on this form of treatment is not generally recommended. Baits may also help reduce house fly populations but are generally ineffective by themselves as a fly control agent.

A major concern associated with the continued use of conventional insecticides has been the development of insecticide resistance within fly populations. This is particularly true in regard to the prolonged use of residual surface sprays (Scott & Georghiou 1985, Meyer et al. 1987). As a result, more recent emphasis has been placed on the use of quick knockdown space sprays in an effort to avoid or minimize the onset of resistance development.

Cultural Control. Sanitation practices to eliminate or minimize fly breeding materials is the most important and effective approach to house fly management in and around swine facilities. To a large extent, these sanitation practices are dependent on the type of manure management system being employed. In systems designed to handle manure in a solid or semi-solid form, the manure should be mechanically removed from the facilities at weekly intervals during the fly season. Ideally, the manure should be hauled directly to fields and scattered thinly to dry. If land is not immediately available, fly development can be minimized by temporarily stockpiling the manure in steep-sided piles on a well-drained site until it can be properly disposed. Since house flies cannot develop in excessively wet breeding materials, facilities that utilize a liquid manure management system generally have fewer house fly problems. These systems, however, should be closely monitored so that manure is not allowed to collect or accumulate along the edges of flow gutters or above the waterline in storage pits since this material is ideal for fly production.

Cultural or sanitation practices should also take into consideration other potential fly breeding materials or areas found in and around swine facilities. Waste feed, moist bedding material, and other organic matter can support fly production and should be mechanically removed and disposed on a frequent and timely basis. Dead animal carcasses should also be properly disposed. In addition, since house flies require moist environments to complete their development, lots and adjacent areas should be constructed or graded to allow water to drain rapidly and completely.

Animal husbandry practices will not eliminate an existing house fly problem. However, an aspect of animal husbandry that can be effective in preventing or minimizing fly problems is facility design, repair, or alteration. Facilities constructed so as to minimize the accumulation or manure, spilled feed, or other fly breeding material or to allow easy and complete removal of these materials can significantly reduce the potential for fly problems. Continual inspection and maintenance of feeding systems, water systems, and manure handling systems is also of critical importance.

Biological Control. The use of biological control agents to control house flies in and around livestock and poultry facilities has received considerable attention in recent years (Peterson 1993). This attention has primarily been focused on the use of parasitic pteromalid wasps in and around beef feedlots, dairies, and poultry facilities. Surveys have been conducted to determine the species composition of naturally occurring pteromalid wasps at these locations and the level of control being afforded by these naturally occurring parasites. Other efforts have centered on the level of control obtained through the release of commercially available pteromalid wasps or other augmentative practices at these locations. To date, however, the species composition and augmented use of these biological control agents in and around swine facilities has not been investigated.

IGR's. Insect growth regulators are available for use in beef cattle, dairy cattle and poultry to control house flies. However, no insect growth regulators are currently registered for use in swine.

Regulatory Concerns. The application of insecticides continues to remain an important component in the control of house flies and other nonbiting flies. As such, legal issues such as Rebuttable Presumption Against Registration (RPAR), Worker Protection Standards, the Delaney Clause, and the Food Quality Protection Act could limit the availability and continued use of these products. Future local, state, or federal legislation regarding waste handling practices could also seriously impact fly problems and limit the use of currently available control tactics.

Sampling. Various sampling methods have been used to monitor adult house fly populations. These methods include sticky traps, light traps and baited traps (Morgan & Pickens 1978), and spot cards and scudder grids. Emergent cone traps have also been employed to sample house flies emerging from likely breeding sites (Broce 1993). However, standardized techniques for sampling house fly populations in and around swine facilities have not been developed. In addition, sampling methodologies for other nonbiting flies found in and around swine facilities are lacking.

Biology and Ecology. With the exception of the fruit fly, the house fly has probably been the most extensively studied of all insects. As a result, much is already known regarding basic house fly biology and ecology. Germane to this discussion include studies on the dispersal of house flies from livestock and poultry facilities (Lindquist et al. 1951, Schoof & Siverly 1954, Hanec 1956, Eddy et al. 1962, Pickens et al. 1967, Lysyk & Axtell 1986) and the characterization of house fly breeding sites in and around livestock and poultry facilities (Walsh 1964, Meyer & Petersen 1983, Hulley 1986, Stafford & Bay 1987, Fatchurochim et al. 1989, Meyer & Shultz 1990). Unfortunately, most of these efforts have focused on house fly populations associated with cattle feedlots, dairies, and poultry facilities. It is evident that more commodity specific research must be conducted if effective integrated pest management programs are to be developed for swine facilities.

Models. Computer simulation can be a valuable aid in the development of effective and efficient pest management programs. The ability to simulate the responses of a pest population to varying management strategies can provide a basis for an optimal decision-making process. A computer simulation model of house fly management in confined animal production systems has recently been developed (Wilhoit et al.1991). This modeling system was primarily developed for use in poultry facilities but may be adapted for use in other confined livestock systems, including swine. The ultimate goal should be the development of practical implementation procedures that could be incorporated into a total swine pest management program. Emphasis must be given to the development of a flexible modeling system that would allow for adjustments based on facilities and management systems and the availability of control alternatives.

Stable Fly, *Stomoxys calcitrans* And Other Biting Flies

Description and Biology. The stable fly is a common biting fly frequently encountered in swine facilities. Female flies deposit their eggs in decaying organic materials, such as wet bedding, waste or spilled feed, or manure mixed with straw or other bedding materials. These eggs typically hatch in about 2 to 4 days whereupon the larvae burrow into and feed on the breeding material. The larvae complete their development in about 10 to 14 days and then seek a drier environment in which to pupate. Adults subsequently emerge about 6 to 8 days later. The entire life cycle can be completed in about 20 to 30 days, depending on temperature. In northern climates stable flies are usually present from May through October with numerous generations being produced during this time period. However, in southern climates or in environmentally controlled swine confinement units, conditions may exist that would allow stable flies to breed continuously throughout the year. Adult stable flies have piercing-sucking mouthparts and feed on blood several times each day. After each feeding, stable flies rest in shaded areas where they digest their bloodmeal.

The mosquito is another common biting fly that can occasionally be found in large numbers in and around swine facilities. Female mosquitoes feed on blood at 3 to 5 day intervals and deposit their eggs either directly on water or on the ground in low lying areas that will later be flooded by water. In many regions massive populations of mosquitoes are produced by rainfall that accumulates in pasture areas or by irrigation runoff that accumulates in adjacent crop lands. Large numbers of mosquitoes can also be produced from more permanent water sources such as swine waste lagoons. The life cycle and number of mosquito generations produced each year varies between species and the seasonal conditions that exist in different regions.

Black flies and biting midges can also occasionally be found in and around swine facilities. Black flies (family Simuliidae) are bloodfeeding pests that develop in swift flowing water. Adults are strong fliers and may disperse more than 10 miles from their breeding site in search of a warmblooded host. Biting midges include members of the family Ceratopogonidae. These tiny bloodfeeding flies develop in wet or semiaquatic habitats, such as the mud or moist soil around streams, ponds, sloughs, and marshes. Biting midges are also capable of breeding in and around anaerobic swine waste lagoons.

Economic Importance. The stable fly is a cosmopolitan pest that can be encountered in all types of pork production systems. Mosquitoes, black flies, and biting midges are also cosmopolitan pests but are most likely to be encountered where established breeding sites are located in close proximity to swine facilities. Recent studies by Weiner & Hansens (1975) and Prullage (1988) found stable flies and mosquitoes to be the most common bloodfeeding flies encountered in swine facilities.

Direct economic losses in pork production resulting from stable fly infestations have not been documented. However, the stable fly has been incriminated in the transmission of eperythrozoonosis (Prullage et al. 1993) which can be of considerable economic importance. The stable fly has also been incriminated in the transmission of numerous human diseases and is therefore considered to be a nuisance and public health hazard. As a result, lawsuits and threats of legal constraint have been instigated against livestock producers for failure to properly control stable flies (Hayes 1993). This in itself could have serious economic implications and should be reason enough for pork producers to implement control measures against the stable fly.

Although stable flies have not been shown to cause direct losses in animal performance, considerable expenditures in control costs targeted at this pest may be incurred by pork producers. While these expenditures may not technically be considered "losses", they nonetheless constitute a significant investment above return.

The effects of mosquitoes, black flies, and biting midges on swine weight gain, feed efficiency, and other production parameters have not been investigated. However, mosquitoes have been incriminated in the transmission of encephalitis and other human diseases. These pests are therefore considered to be a nuisance and public health hazard. This could have economic implications for pork producers if breeding sites for these pests become established in and around swine facilities.

By definition, economic injury levels for stable flies, mosquitoes, black flies, and biting midges cannot be determined since these pests have not yet been shown to cause direct economic losses in swine production or performance. A justification could be made, however, for the establishment of an intolerable nuisance level based on the number of flies dispersing from swine facilities. Unfortunately, the scientific establishment of such levels is highly improbable since the tolerance for these flies is likely to vary considerably from region to region, state to state, locality to locality, and individual to individual.

Current Methods of Control and Research Needs

Chemical. Current control measures for stable flies found in and around swine facilities include the application of conventional insecticides as residual surface sprays, space sprays, larvicide sprays, or feed additives. Residual surface sprays are applied to fly resting areas, such as the framework of swine housing, ceilings, trusses, electric light wires and fixtures, outside walls and fences, and any other surfaces that may attract flies. These surface sprays are applied with hydraulic or compressed air sprayers to the point of runoff and are generally effective for 1 to 3 weeks or longer. Space sprays are applied with foggers or mist blowers around the facility to provide a quick knockdown of adult fly populations. These applications have little, if any, residual activity and may need to be reapplied at frequent intervals to obtain maximum effectiveness. Larvicide sprays applied to fly breeding materials may, at times, aid in the control of stable flies. While these sprays may be of benefit in certain situations, total reliance on this form of treatment is generally not recommended. Feed additives function by passing through the animal's digestive system to kill fly larvae developing in the manure. However, these materials are generally of limited effectiveness since stable flies rarely develop in fresh manure.

A major concern associated with the continued use of conventional insecticides is the potential development of insecticide resistance within stable fly populations. This is particularly true of residual surface sprays. As a result, more recent emphasis has been placed on the use of quick knockdown space sprays in an effort to avoid or minimize the onset of resistance development.

Current control measures for mosquitoes, black flies, and biting midges include the application of conventional insecticides as space sprays, residual surface sprays, or larvicide treatments. Space sprays provide a quick knockdown of adult populations but may need to be reapplied at frequent intervals during pest outbreaks. Residual surface sprays applied to fly resting areas can aid in the reduction of adult populations. Larvicide treatments in the form of liquid sprays or granular applications can be effective in preventing pest outbreaks if treatments are timed properly and applied to all known breeding sites.

Cultural Control. Sanitation practices to eliminate or minimize fly breeding materials is the most important and effective approach to stable fly management in and around swine facilities. Ideally, these materials should be mechanically removed at weekly intervals and scattered thinly on fields to dry. Eliminating or minimizing temporary standing water sources around swine facilities through draining, filling, or grading practices can be effective in preventing or reducing mosquito populations. Minimizing shoreline vegetation around swine waste lagoons or other permanent water sources can also aid in mosquito control. Cultural control practices for black flies and biting midges encountered in and around swine facilities have not been developed or investigated.

Animal husbandry practices will not eliminate an existing biting fly problem. However, an aspect of animal husbandry that can be effective in preventing or minimizing problems is facility design, repair, or alteration. Facilities constructed so as to minimize the accumulation of breeding materials or to allow easy and complete removal of these materials can significantly reduce the potential for stable fly problems. Continual inspection and maintenance of feeding systems, watering systems, and waste handling systems is also of critical importance. Facilities constructed or altered to prevent or minimize the accumulation of standing water sources can also significantly reduce the potential for mosquito problems.

Biological Control. The use of biological control agents to control stable flies in and around livestock facilities has received considerable attention in recent years (Peterson 1993). This attention has primarily been focused on the use of parasitic pteromalid wasps in and around beef feedlots and dairies. Surveys have been conducted to determine the species composition of naturally occurring pteromalid wasps at these locations and the level of control being afforded by these naturally occurring agents. Other efforts have centered on the level of control obtained through the release of commercially available pteromalid wasps or other augmentative practices at these locations. To date, however, the species composition and augmented use of these biological control agents in and around swine facilities has not been investigated.

Various biological control agents are available for use against pest mosquito species. These agents are largely targeted against the larval stage of the mosquito and include mosquito-eating fish, predatory mosquito species, and bacterial agents. These agents have been shown to be effective against mosquitoes in urban communities when utilized in a pest management program. However, their use in and around swine facilities has not been investigated. Likewise, the area-wide use of bacterial agents applied to larval habitats has been demonstrated to be effective in reducing black fly populations in the poultry producing regions of North Carolina (J.J. Arends, personal communication). Their use and effectiveness in relation to the pork industry, however, has not been investigated. To date, biological control agents for biting midges remain unknown.

IGR's. Insect growth regulators for stable fly control are available for use in beef cattle and dairy cattle. However, no insect growth regulators are currently registered for use in swine to control stable flies. The insect growth regulator methoprene is registered for use as a mosquito larvicide treatment. No insect growth regulators are currently available for use against black flies or biting midges.

Regulatory Concerns. The application of insecticides continues to remain an important component in the control of stable flies and other biting flies. As such, legal issues such as Rebuttable Presumption Against Registration (RPAR), Worker Protection Standards, the Delaney Clause, and the Food Quality Protection Act could limit the availability and continued use of these products. Future local, state, or federal legislation regarding waste handling practices could also seriously impact fly problems and limit the use of currently available control tactics.

Sampling. Various sampling methods have been used to monitor adult stable fly populations. These methods include animal-baited traps, sticky traps, carbon dioxide-baited traps, light traps, emergence traps (Harris 1978) and Alsynite traps (Williams 1973, Broce 1988). However, standardized techniques for sampling stable fly populations in and around swine facilities have not been developed. A variety of sampling methods have also been used to monitor mosquito, black fly, and biting midge populations. These methods include animal-baited traps, sticky traps, carbon dioxide-baited traps, light traps, emergence traps, vehicle-mounted traps, bite counts, and direct netting. While certain of these methods have been used extensively to monitor pest populations in urban areas, their standardized use in and around swine facilities remains largely unexplored.

Biology and Ecology. Much is already known regarding basic stable fly biology and ecology. Germane to this discussion include studies on the dispersal of stable flies from livestock facilities (Bailey et al. 1973, Eddy et al. 1962, Hogsette & Ruff 1985, Scholl 1986, Williams & Rogers 1976) and the characterization of stable fly breeding sites in and around livestock facilities (Broce 1986, Hogsette et al. 1987, Meyer & Petersen 1983, Meyer & Shultz 1990, Schmidtmann 1988, Skoda et al. 1991). Unfortunately, most of these efforts have focused on stable fly populations associated with cattle feedlots and dairies. It is evident that more commodity specific research must be conducted if effective pest management programs are to be developed for swine facilities.

Models. Computer simulation can be a valuable aid in the development of effective and efficient pest management programs. The ability to simulate the responses of a pest population to varying management strategies can provide a basis for an optimal decision-making process. A computer simulation model of house fly management in confined animal production systems has recently been developed (Wilhoit et al. 1991). While this modeling system was primarily developed for house fly management in poultry facilities, it could potentially be modified or expanded to include stable fly management in other confined livestock systems, including swine. Modeling systems for mosquito, black fly, and biting midge populations associated with livestock production have not been developed. The ultimate goal should be the development of practical implementation procedures that could be incorporated into a total swine pest management program. Emphasis must be given to the development of a flexible modeling system that would allow for adjustments based on facilities and management systems and the availability of control alternatives.

Summary of Research and Extension Needs for Swine

Research Needs

(1) The major pests associated with swine and pork production facilities include sarcoptic mange mites, hog lice, and certain species of biting and nonbiting flies. Sarcoptic mange mites and hog lice are unique to pork production. Therefore, the major thrust of research efforts for this commodity should be directed at these pests.

(2) Of critical importance is basic research targeted at elucidating a greater understanding of sarcoptic mange mite and hog louse biology and ecology under varying management systems and environments. A greater understanding of the economic impact of mange and lice is also needed, especially as it relates to differences in management systems, disease transmission, and the presence of other external or internal parasites. The key to progress in this area, however, lies in the development of standardized, reliable, and efficient sampling methodologies for assessing mange mite and lice populations within individual animals or herds.

(3) The application of ectoparasiticides currently forms the basis for sarcoptic mange mite and hog lice control in swine. Until alternative control strategies are developed, the evaluation of potential new ectoparasiticides will remain a necessity. Research is also needed on the development of alternative control approaches and methodologies which could fit into an integrated pest management program for mange mites and lice. These alternative approaches include cultural and management practices, biological control, genetic control, and host resistance.

(4) Regarding house flies and stable flies, a voluminous amount of basic research data currently exists. Evaluation and synthesis of the available literature on fly bionomics, as it relates to the swine industry, will be vital to the establishment of an effective integrated

pest management program. Other research will need to be initiated to fill gaps currently existing in the data base. Of primary importance are basic studies targeted at elucidating the effects of varying production systems and waste handling systems on fly dispersal, population dynamics, and behavior. Standardized sampling methodologies must also be developed and implemented. A greater understanding of the species composition, biology, and ecology of naturally occurring biological control agents is also of critical importance. The true relevance and importance of the house fly and stable fly as a nuisance and public health hazard must be fully explored. In addition, further investigation is needed on the role of these flies in the potential transmission of swine diseases.

(5) Sanitation practices to eliminate or minimize fly breeding materials is the most important and effective approach to house fly and stable fly management. As the trend towards total swine confinement systems continues, control research will be needed to evaluate the direct effects of varying confinement waste handling systems on fly bionomics. These waste handling systems must also be evaluated to determine their effects on naturally occurring fly predators, parasites, and pathogens.

(6) The application of insecticides in the form of surface sprays and space sprays continues to remain a vital component of house fly and stable fly management in swine facilities. Until alternative control strategies are developed the evaluation of potential new surface spray and space spray insecticides will remain a necessity. Research is also needed on the development of alternative control approaches and methodologies which could fit into an integrated pest management program for house flies and stable flies. These alternative approaches include the use of biological control agents, nonchemical baits, traps, sex attractants, and insect growth regulators.

Extension Needs

(1) As pest management strategies become available they must be evaluated to determine which techniques can be used under certain conditions, or in combinations, to provide the most cost effective form of control. It will also be necessary to determine the effects of specific swine management practices on these strategies.

(2) The role of extension in technology transfer and input for specific research needs is critically important for all swine pests. Close cooperation between researchers, extension personnel, pork producers, integrators, and veterinarians is tantamount to the dissemination of pertinent information and in the development and implementation of effective and acceptable IPM programs. Extension expertise is necessary to design effective information delivery systems, evaluate the acceptance of implemented IPM programs, and provide feedback as to additional research needs or special problems unique to a geographical area.

References Cited

Alva-Valdes, R., D.H. Wallace, A.G. Foster, G.F. Ericsson & J.W. Wooden. 1986. The effects of sarcoptic mange on the productivity of confined pigs. Vet. Med. (Food Anim.). 81: 258-262.

Anonymous. 1998. Hogs and pigs. National Agricultural Statistics Service, USDA. Mt. An. 4(12-98). http://usda.mannlib.cornell.edu/usda/usda.html (25 May 1999).

Arends, J.J., C.M. Stanislaw & D. Gerdon. 1990. Effects of sarcoptic mange on lactating swine and growing pigs. J. Anim. Sci. 68: 1495-1499.

Bailey, D.L., T.L. Whitfield & B.J. Smittle. 1973. Flight and dispersal of the stable fly. J. Econ. Entomol. 66: 410-411.

Broce, A.B. 1993. Dispersal of house flies and stable flies. pp. 50-60. *In* G.D. Thomas & S.R. Skoda, eds., Rural Flies in the Urban Environment, North Central Regional Publication No. 335.

Broce, A.B. 1988. An improved Alsynite trap for stable flies, *Stomoxys calcitrans* (Diptera:Muscidae). J. Med. Entomol. 25: 406-409.

Broce, A.B. 1986. Stable fly breeding: Feedlots vs. urban areas. *In* Department Report, Stable Fly Biology and Control in Cattle Feedlots. Kansas Agricultural Experiment Station, 86-362-D.

Campbell, J.B. 1993. The economics of the fly problem. pp. 34-39. *In* G.D. Thomas & S.R. Skoda, eds., Rural Flies in the Urban Environment, North Central Regional Publication No 335.

Cargill, C.R. & K.J. Dobson. 1979. Experimental *Sarcoptes scabiei* infestations in pigs: (2) Effects on production. Vet. Rec. 104: 33-36.

Courtney, C.H., W.L. Ingalls & S.L. Stitzlein. 1983. Ivermectin for the control of swine scabies: Relative values of prefarrowing treatment of sows and weaning treatment of pigs. Am. J. Vet. Res. 44: 1220-1223.

Dalton, P.M. & W.G. Ryan. 1988. Productivity effects of pig mange and control with ivermectin. Vet. Rec. 122: 307-308.

Davis, D.P. & R.D. Moon. 1990. Dynamics of swine mange: A critical review. J. Med. Entomol. 27: 727-737.

Davis, D.P. & R.E. Williams. 1986. Influence of hog lice, *Haematopinus suis*, on blood components, behavior, weight gain, and feed efficiency of pigs. Vet. Parasitol. 22: 307-314.

Diesch, S.L., R.M.H. Reusbech & F.M. Martin. 1982. Identification of the 20 most prevalent diseases and syndromes reported in swine in Minnesota, 1974-1980. United States Animal Health Association. pp. 361-362.

Eddy, G.W., A.R. Roth & F.W. Plapp, Jr. 1962. Studies of the flight habits of some marked insects. J. Econ. Entomol. 55: 603-607.

Fatchurochim, S., C.J. Geden & R.C. Axtell. 1989. Filth fly (Diptera) oviposition and larval development in poultry manure of various moisture levels. J. Entomol. Sci. 24: 224-231.

Gaafar, S.M, J.J. Arends, A. Hogg, K. Holscher & R.W. Williams. 1986. An integrated program using Taktic to control mange in swine. J. Agric. Entomol. 3: 374-381.

Hanec, W. 1956. A study of the environmental factors affecting the dispersion of house flies (*Musca domestica* L.) in a dairy community near Fort Whyte, Manitoba. Can. Entomol. 88: 270-272.

Harris, R.L. 1978. Stable flies and horn flies (Subfamily Stomoxynae). pp. 77-79. *In* R.A. Bram, ed., Surveillance and Collection of Arthropods of Veterinary Importance, United States Department of Agriculture Handbook No. 518.

Hayes, D.K. 1993. Legal aspects of rural flies in the urban environment. pp. 40-45. *In* G.D. Thomas & S.R. Skoda, eds., Rural Flies in the Urban Environment, North Central Regional Publication No. 335.

Hogsette, J.A. & J.P. Ruff. 1985. Stable fly (Diptera:Muscidae) migration in northwest Florida. Environ. Entomol. 14: 170-175.

Hogsette, J.A., J.P. Ruff & C.J. Jones. 1987. Stable fly biology and control in northwest Florida. J. Agric. Entomol. 4: 1-11.

Hulley, P.E. 1986. Factors affecting numbers of *Musca domestica* Linnaeus (Diptera:Muscidae) and some other flies breeding in poultry manure. J. Ent. Soc. Sth. Afr. 49: 19-27.

Lee, R.P., D.J.D. Dooge & J.M. Preston. 1980. Efficacy of ivermectin against *Sarcoptes scabiei* in pigs. Vet. Rec. 107: 503-505.

Lindquist, A.W., W.W. Yates & R.A. Hofman. 1951. Studies of the flight habits of three species of flies with radioactive phosphorus. J. Econ. Entomol. 44: 397-400.

Lysyk, T.J. & R.C. Axtell. 1986. Movement and distribution of house flies (Diptera:Muscidae) between habitats in two livestock farms. J. Econ. Entomol. 79: 993-998.

Martineau, G.P., J. Vaillancourt & J.L. Frechette. 1984. Control of *Sarcoptes scabiei* infestation with ivermectin in a large intensive breeding piggery. Can. Vet. J. 25: 235-238.

Meyer, J.A., G.P. Georghiou & M.K. Hawley. 1987. House fly (Diptera:Muscidae) resistance to permethrin on southern California dairies. J. Econ. Entomol. 80: 636-640.

Meyer, J.A. & J.J. Petersen. 1983. Characterization and seasonal distribution of breeding sites of stable flies and house flies (Diptera:Muscidae) on eastern Nebraska feedlots and dairies. J. Econ. Entomol. 76: 103-108.

Meyer, J.A. & T.A. Shultz. 1990. Stable fly and house fly breeding sites on dairies. Calif. Agric. 44: 28-29.

Miller, M. 1991. They find disease you don't. Pork'91. 11: 38-40.

Morgan, N.O. & L.G. Pickens. 1978. House flies and other nonbiting flies (Family Muscidae). pp. 72-76. *In* R.A. Bram, ed., Surveillance and Collection of Arthropods of Veterinary Importance, United States Department of Agriculture Handbook No. 518.

Nickle, E.A. & G. Danner. 1979. Experimental studies of course and effects of pediculosis in domestic swine. Arch. Exp. Vet. Med. Leipzig. 33: 645-649.

Petersen, J.J. 1993. Biotic agents to control house flies and stable flies. pp. 70-82. *In* G.D. Thomas & S.R. Skoda, eds., Rural Flies in the Urban Environment, North Central Regional Publication No. 335.

Pickens, L.G., N.O. Morgan, J.G. Harstock & J.W. Smith. 1967. Dispersal patterns and populations of the house fly affected by sanitation and weather in rural Maryland. J. Econ. Entomol. 60: 1250-1255.

Prullage, J.B. 1988. Mechanical transmission of *Eperythrozoan suis* Splitter by arthropod vectors. Ph.D. Dissertation. Purdue University, West Lafayette, IN, 73 pp.

Prullage, J.B., R.E. Williams & S.M. Gaafar. 1993. On the transmissibility of *Eperythrozoan suis* by *Stomoxys calcitrans* and *Aedes aegypti*. Vet. Parasitol. 50: 125-135.

Schmidtmann, E.T. 1988. Exploitation of bedding in dairy outdoor calf hutches by immature house flies and stable flies (Diptera:Muscidae). J. Med. Entomol. 25: 484-488.

Scholl, P.J. 1986. Field population studies of *Stomoxys calcitrans* (L.) in eastern Nebraska. Southwest. Entomol. 11: 155-160.

Schoof, J.F. & R.E. Siverly. 1954. Multiple release studies on the dispersion of *Musca domestica* at Phoenix, Arizona. J. Econ. Entomol. 47: 830-838.

Scott, G.L. & G.P. Georghiou. 1985. Rapid development of high-level permethrin resistance in a field-collected strain of the house fly (Diptera:Muscidae) under laboratory selection. J. Econ. Entomol. 78: 326-319.

Skoda, S.R., G.D. Thomas & J.B. Campbell. 1991. Developmental sites and relative abundance of immature stages of the stable fly (Diptera:Muscidae) in beef cattle feedlot pens in eastern Nebraska. J. Econ. Entomol. 84: 191-197.

Stafford, K.C.III & D.E. Bay. 1987. Dispersion pattern and association of house fly, *Musca domestica* (Diptera:Muscidae), larvae and both sexes of *Macrocheles muscaedomesticae* (Acari:Macrochelidae) in response to poultry manure moisture, temperature, and accumulation. Environ. Entomol. 16: 159-164.

Von Heinritzi, K. 1992. Untersuchungen zur Ubertragbarkeit von *Eperythrozoan suis*. Tierarztl. Umschau. 47: 588-599.

Walsh, J.D. 1964. A survey of fly production in cattle feedlots in the San Joaquin Valley. Calif. Vector Views. 11: 33-39.

Weiner, T.J. & E.J. Hansens. 1975. Species and numbers of blood sucking flies feeding on hogs and other animals in southern New Jersey. J. N.Y. Entomol. Soc. 83: 198-202.

Wilhoit, L.R., R.E. Stinner & R.C. Axtell. 1991. Computer simulation model of house fly management in confined-animal production systems. North Carolina Agricultural Research Service Technical Bulletin No. 296.

Williams, D.F. 1973. Sticky traps for sampling populations of *Stomoxys calcitrans*. J. Econ. Entomol. 66: 1279-1280.

Williams, D.R. & A.J. Rogers. 1976. Vertical and lateral distribution of stable flies in northwestern Florida. J. Med. Entomol. 13: 95-98.

Wooten, E.L., F. Blecha, A.B. Broce & D.S. Pollmann. 1986. The effect of sarcoptic mange on growth performance, leukocytes and lymphocyte proliferative response in pigs. Vet. Parasitol. 22: 315-324.

Wooten-Saadi, E.L., C.A. Towell-Vail, R.E. Williams & S.M. Gaafar. 1987a. Incidence of *Sarcoptes scabiei* (Acari:Sarcoptidae) and *Haematopinus suis* (Anoplura:Haematopinidae) on swine in Indiana. J. Econ. Entomol. 80: 1031 1034.

Wooten-Saadi, E.L., A.B. Broce, J.S. Stevenson & J.L. Nelssen. 1987b. Growth performance and behavioral patterns of pigs infested with sarcoptic mites (Acari:Sarcoptidae). J. Econ. Entomol. 80: 625-628.

****
****

End of Swine section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****
****

SHEEP AND GOAT SUMMARY

The economic benefit of management of most arthropod pests of sheep and goats in the United States is unknown. The only pest or disease problems for which economic data appear to be available are *Melophagus ovinus*, the sheep ked, and bluetongue virus which is transmitted by the biting midge, *Culicoides variipennis*. The economic importance of sheep bot fly, *Oestrus ovis*, and various species of sheep lice is unknown, and needs to be documented. The occurrence and the distribution of many of the arthropod pests of sheep and goats are unknown.

Currently, control of arthropod pests of sheep and goats relies heavily on insecticides. It appears that as long as insecticides are effective and relatively inexpensive this approach will continue. Because of the limited market, the number of insecticides registered for use on sheep and goats is small when compared to that for cattle. Not all pests of sheep are satisfactorily controlled through the use of the insecticides currently registered. There is a need for more efficacious insecticides as well as insecticides with alternative modes of action for use in resistance management. There is a need for research on insecticide formulations, delivery systems, alternative insecticides and biorational insecticides.

The knowledge base necessary to develop IPM programs for most arthropod pests of sheep is lacking. Information necessary to develop IPM programs includes: economic injury levels and economic thresholds; basic biology of the pest or vector species; sampling or monitoring techniques; and alternative control technology. To m limited extent, information may be transferable from research on pests or diseases of other domestic and wild animal species and from research performed in other parts of the world where sheep and goats are more important.

A network of specialists involved with sheep and goat parasite work in the United States should be established to exchange information. Educational information needs to be updated. If sufficient research data are not generated in the U.S., sources of information in other parts of the world, e.g., Australia or New Zealand, should be located to update educational information and recommendations for producers.

Research on bluetongue virus will continue, primarily because of its importance to the United States cattle industry. Biological studies include vector distribution, genetics, and ecology to define regions at risk and targets for control. New approaches to control strategies will integrate vaccines with biological, genetic, and cultural control.

Currently, sheep ked is controlled effectively by the use of low volume or pour-on applications of pyrethroid insecticides. Recently, the insecticide diazinon has also been found to be effective as a pour-on and has received label clearance. This provides the sheep producer with an insecticide with an alternate mode of action. There is, however, a need for additional insecticides, including biorationals, with alternative modes of action. Although economic injury levels related to weight gains and wool production have been studied, there is no quantitative information available regarding the relationship between sheep ked and pelt defects, specifically "cockle."

Oestrus ovis, the sheep bot fly, may be a pest of significant economic importance in the United States because of its wide distribution, and the effects of both the adults and larvae on the host. Research is needed on the potential economic benefit of management of this pest.

Although not currently a nation-wide problem, lice on sheep and goats is a serious concern in certain sheep and goat producing regions of the country because of reports of insecticide resistance. Although there are research needs for IPM, current pressing needs are studies of management, especially resistance management, and development of alternative insecticide products for lice control.

SHEEP AND GOATS

Committee Members

John E.Lloyd, Chair
Dennis R. Berkebile
Ronald B. Davey
Terri Erk
Thomas W. Fuchs
Frederick R. Holbrook
Gregory D. Johnson
Bradley A. Mullens
Robert G. Pennington
Walter J. Tabachnick

Economic Significance of Sheep and Goats

The number of sheep in the United States has been in decline since the 1942 when all sheep and lambs reached a peak of 56.21 million animals. The inventory of all sheep and lambs in the United States in January 1, 1996 totaled 8.5 million head, down 5% from 1995 and 13% below 1994 (National Agricultural Statistics Service 1996). About 16.5% of U.S. sheep are feeders. The dollar value of sheep increased from $543 million in 1977 to $666 million in 1992 because the value of the animals themselves has increased by approximately one-third. Gross income from slaughter animals has remained around $400 million since 1977 but the value of wool decreased from $77 million in 1977 to $46 million in 1991 because the value of wool has dropped from 72 to 55 cents per pound (National Agricultural Statistics Service 1992).

Within the United States distribution of the sheep industry is primarily in eleven western states, with most occurring in Texas, California, Wyoming, Colorado and South Dakota. The industry is more predominant in areas where sheep convert native forage that is not consumable by humans and cannot be harvested by traditional methods into products that humans use.

The number of sheep in the United States has been declining for the past 50 years for many reasons: a shift from natural fibers to synthetic; losses to predators; the high cost of labor; a lack of good herders; the inability to compete with imports; and consumers preference for meats other than lamb. Newer reasons that particularly affect the western sheep industry include environmental concerns and potential increases in grazing fees on public lands and removal of price supports for wool. Meat goat production, on the other hand, is expected to become increasingly important as wool and mohair government incentive payments are phased out and meat production becomes a larger proportion of income derived from sheep and goats.

There are approximately two million goats in the U.S. with about 85 percent (1.8 million) being found in a county area of Texas known as the Edwards Plateau. The Angora goat comprises the largest numbers (approximately 1.4 million), however, due to the phase out and eventual loss (1996) of the wool and mohair incentive program, Angora goat numbers have declined appreciably in the last three years. The price of mohair is dependent upon its use in fashions in Europe and can vary a great deal from year to year. The price of mohair has been very low since 1990 with the yearly clip worth approximately 10 to 13 million dollars (not considering the incentive payment).

There are approximately 0.5 million Spanish meat goats in the U.S. The meat goat industry has prospered the last few years due in part to the fact that many ethnic groups like goat meat and it is very lean and low in cholesterol. Meat goats weighing up to 50 to 60 pounds have been worth approximately $1 per pound live weight. The introduction of the Boer goat from South Africa seven years ago has also caused a great deal of excitement with extremely high prices being paid. The use of embryo transfer and artificial insemination has increased tremendously in the goat industry due to the introduction of the Boer goat with many of the dairy breeds being used as recipients and in crossbreeding programs. It will be interesting to follow the Boer goat and its impact on the meat goat industry in the U.S.

Insect Pests of Sheep and Goats

Wool Maggots or Fleece Worms: black blow fly, *Phormia regina* (Meigen), northern black blow fly, *Protophormia terraenovae* (Robineau-Desvoidy), green bottle fly, *Phaenicia sericata* (Meigen), and secondary screwworm fly, *Cochliomyia macellaria* (Fabricius).

Description and Biology. Very probably, on a world-wide basis, the most significant external parasite of sheep is the sheep blow fly, which is particularly damaging in Australia and Africa. Sheep struck by the blow fly frequently die. If they survive, the quantity and quality of their wool are severely reduced. In North America the blow fly problem is less severe. Sheep and, to a lesser extent, goats are subject to parasitization by several species of Calliphoridae (blow flies)(Hall 1948, Zumpt 1965). A few species, such as the primary screwworm, *Cochliomyia hominivorax*, oviposit around or in wounds and feed on living flesh. The greater problem in temperate countries, however, is with species attracted to moist, soiled fleece. Larvae of these flies are known as wool maggots or fleece worms.

The most important wool maggot species in North America are the green bottle fly, *Phaenicia sericata*, the black blow fly, *Phormiaregina*, and the secondary screwworm, *Cochliomyiamacellaria*. *P. regina* is most abundant in cooler spring weather, while the other species are most common in summer. In the northern portion of North America, *P. terra-novae* tends to replace *P. regina*. Another species, *Phaenicia cuprina*, is very serious pest of sheep in Australia and Africa.

The Calliphoridae mainly develop in carrion. Indeed, much of the recent work on biology and ecology in North America has been relative to their role as primary colonizers of human corpses, and they are probably the most useful taxon in determining time of death (Greenberg 1991). Responding presumably to similar, microbially-mediated, olfactory cues, female flies also lay eggs (known as "sheep strike") in moist, soiled fleece. Wet weather, wounds, or diarrhea (scours) tend to predispose sheep to parasitization. It appears that the presence of fly larvae tends to further increase fly oviposition on the animal, leading to a worsening and spreading infestation. Eggs hatch quickly, usually in less than 12 hours, and larvae develop quickly through three instars. They can be ready to leave the host within three to four days and pupate in the soil. Pupal development time varies with ambient temperatures, but commonly is 7-10 days under summer conditions. Multiple generations occur during a year, and overwintering occurs as larvae, pupae, or adults.

Economic Importance. Fly larvae often feed near the skin surface. Infested sheep are very irritated, restless, groom constantly, and lose weight and condition. It is common for portions of the fleece near the infestation to slough off. Infested animals may die without treatment.

Methods of Control. Animal husbandry is key to preventing wool maggots. Lambing and shearing early in the season, before the onset of fly activity, is advisable. Care in handling and shearing the sheep will minimize wound sites which enhance oviposition. Prompt treatment or clipping of soiled fleece on sheep with scours is helpful. Existing infestations can be treated with insecticides as well to eliminate the larvae. Two surgical procedures are common in Australia to prevent fleece being wetted by urine or feces. One, mulesing, involves removal of a portion of skin in the crotch area, while a less common procedure, pizzle dropping, lowers the penis angle to minimize fleece wetting.

A substantial amount of work has been done over the past decade on cyromazine and ivermectin, including studies of both lethal and sublethal effects on the calliphorid targets (Kotz 1992, Mahon et al. 1993). Biological control using microbial agents (protozoans, bacteria, fungi) and nematodes (Steinernematidae, Heterorhabditidae) has also been explored (e.g. Bedding et al. 1983), but prospects for successful control in the field are mixed. The most promising are fleece-adapted strains of *B, thuringiensis,* which colonize the fleece and may give protection for up to 12 weeks (Pinnock 1994).

Population modelling also has received attention, with the goal of predicting fly population levels (Vogt and Morton 1991, Wall et al. 1993) and implementing genetic controls or sterile releases. Most recently there has been quite a bit of work on vaccine development, which offers a possibly attractive solution (East and Eisemann 1993).

Research and Extension Needs. Work on wool maggots, and sheep and goats generally, has been hampered by the relatively minor economic status of these animals in the U.S. We still do not have a clear understanding of how severe or widespread the problem is, of control measures taken and their cost and real or perceived effectiveness, or even which fly species constitute the most severe problem in different regions and seasons.

It behooves livestock entomologists to keep in touch with work being done in forensic entomology which may improve our knowledge of the ecology of these flies. Basic research on chemical cues in host location, interactions with microorganisms in the larval habitat, temperature preferences or microhabitat alteration by fly larvae, mate location and mating behavior, etc. are all critical to successful control. At this juncture, the applied research most needed involves quantitative survey on the extent and distribution of the problem and costs and logistics of addressing those problems with existing technology.

Sucking Lice and Chewing (Biting) Lice: sheep biting louse, *Bovicola ovis* (Schrank), face and body louse, *Linognathus ovillus* (Neumann), foot louse, *L. pedalis* (Osborn), African blue louse, *L. africanus* (Kellogg and Paine), goat sucking louse, *L. stenopsis* (Burmeister), goat biting louse, *B. caprae* (Gurlt), Angora goat biting louse *B. limbatus* (Gervais), hairy goat louse *B. crassipes* (Rudow).

Description and biology. Several species of chewing (biting) and sucking lice infest sheep and goats in the United States. One species of biting lice, *Bovicola ovis* and several species of sucking lice including *Linognathus ovillus, L. pedalis* and *L. africanus* infest sheep in the United States. Meat, dairy and Angora goats are infested by two species of sucking lice, *L. stenopsis* and *L. africanus* and three species of biting lice, *Bovicola caprae*, *B. limbatus* and *B. crassipes*. Each of these species completes its life cycle on the host (Drummond et al. 1988).

The sheep biting louse is one of the most common lice found on sheep. It is a small species, up to 1.8 mm in length, with a pale abdomen, darker thorax and reddish head. It is very active and moves rapidly through the wool usually near the skin.

B. limbatus is somewhat similar to the sheep biting louse in appearance and is the most common biting louse found on Angora goats (Darrow 1973). It lives on the surface of the skin and feeds on scales, skin debris, bits of hair and other debris (Peterson and Bushland 1956, Darrow 1973).

The biting louse, *B. caprae*, as well as the sucking lice, *L. stenopsis* and *L. africanus*, which infest meat goats including Spanish goats may become increasingly important as the meat goat industry further develops in the U. S. This industry is expected to become increasingly important as wool and mohair government incentive payments are phased out and meat production becomes a larger proportion of income derived from sheep and goats.

Economic importance. Biting lice live on scurf and other skin and wool products. They cause intense irritation which sheep and goats relieve by biting or pulling wool or mohair or rubbing against posts and other objects. The fleece of heavily infested sheep may become ragged and torn and reduced in value.

Heavy infestations of lice on goats have been reported to cause discomfort to the host which may result in loss of weight, lowered vitality and reduction in mohair (Peterson and Bushland 1956, Darrow 1973). Results of a more recent study evaluating the effects of a short-term lice infestation on Angora goats on objectively measurable mohair characteristics, however, indicated that effects were negligible (Lupton et al. 1988). Further evaluation on impacts on mohair as well as the animal are needed.

L. africanus is probably the most important sucking louse of sheep and goats. This louse may occur anywhere on the animal's body and emaciated or weak animals are particularly susceptible. The lice suck the animal's blood and can produce severe anemia.

Methods of Control. Lice on sheep are currently controlled primarily with sprays, dips or pour-ons. This is usually done immediately after shearing when there is a natural reduction in the lice population due to physical removal of lice plus the reduction due to the drastic change in the microenvironment.

Current recommendations for control of biting lice on Angora goats indicate that the most effective time to apply insecticides is 4-8 weeks after shearing (Fuchs 1990). Delaying treatments was reported to have several advantages over treating immediately after shearing: 1) takes advantage of natural mortality due to habitat modification following shearing before a treatment decision is made, 2) allows hair to regrow to a sufficient length to retain more insecticide on the animal increasing residual control and 3) reduces time period over which control is required. Insecticides applied as pour-ons, sprays or dips have been shown to be effective (Fuchs and Shelton 1985, Miller et al. 1985). Recent work in Australia indicates that fleece-adapted strains of *B. thuringiensis* (esp. *kurstaki*) may be useful in control (Pinnock 1994).

Research and Extension Needs. Economic injury levels and economic thresholds for lice on sheep and goats have not been established. This is a basic requirement for the development of an IPM program. There is little understanding of the exact nature of damage and the possibilities of genetic selection of sheep and goats for resistance to lice.

A primary need is in determining when control is justified and how treatments are most effectively delivered. Studies to date have had variable results. Insecticide resistance in lice on Angora goats appears to be a problem. Little is known about resistance mechanisms or resistance management in lice, and few alternative insecticides are labelled. Insecticide products are difficult to get labelled on sheep and goats due to the small potential market. While products such as synthetic juvenile hormones and chitin inhibitors have been shown to be effective and would be excellent alternatives in resistance management programs, none have been labelled to date (Chamberlain and Hopkins 1971, Chamberlain et al. 1976, Miller et al. 1985).

There is a need to better define the problem of lice on sheep and goats including the species involved in various geographic areas.

Sheep Ked, *Melophagus ovinus*

Description and Biology. The sheep ked, often called sheep "tick" by sheep producers, is a permanent ectoparasite specific to sheep. It is a wingless, blood-sucking fly, and can easily be distinguished from true ticks because the adult possesses 6 legs.

The sheep ked has an unusual way of reproducing. An egg hatches and the larva develops within the uterus of the female. A single larva develops at a time. The larva is cemented to the sheep's wool and forms the brownish-red, barrel-shaped puparium. Females produce about 15 offspring during a lifetime (Lloyd 1985).

Keds that fall off the host may be able to regain a host. If not, they usually succumb within a week and present little danger of infesting a flock (Strickman et al. 1984). Ked populations build up during the autumn and winter months and reach peak numbers in January and February, then decline until June to low numbers that are carried over the summer (Legg et al. 1991, Pfadt 1976).

Economic Importance. Drummond et al. (1981) estimated that the sheep ked causes an 8% reduction in weight gain, a 15% reduction in wool production, and a 30% reduction in value of sheep skins which are responsible for annual losses in sheep production in the United States of $40.9 million. In addition, losses to sheep ked include "back loss" which is death loss of sheep that roll onto their backs, presumably to relieve irritation, and are unable to right themselves again (Lloyd 1985). Significant differences in production, both weight gains of lambs and fleece production by lambs or ewes have been attributed to sheep ked infestations in Canada (Nelson and Slen 1968) and the United States (Everett et al. 1971). Wool quality is negatively affected due to sheep ked debris. Incidence of the ked-induced defect in tanned hides known as "cockle" makes leather unmarketable. Feeding by the sheep ked results in fibrin deposition in the skin. The fibrin is in dense nodules that are hard and thick and are not easily penetrated by chemicals such as dyes (Everett et al. 1969).

Methods of Control. Several insecticides are used currently to control infestations of sheep ked. The most effective and convenient time to treat is in spring following shearing. Insecticides may be applied as sprays, dips, pour-ons, sprinkles (from a sprinkler can) or dusts (Lloyd 1985). Pyrethroid insecticides, applied as either a low volume spray (Lloyd et al. 1978) or pour-on (Lloyd et al. 1982) have been adopted by many sheep producers as the compounds of choice in state-wide ked free programs in the West. These products are so efficacious that keds may be eliminated from individual flocks thus eliminating the need for annual treatment.

Research and Extension Needs. Statistically sound methods of estimating ked populations have been presented by Nelson et al. (1957) and Legg et al. (1991). No economic thresholds or economic injury levels have been established for sheep ked infestations. There is a need to determine the relationship between ked density and the severity of the cockle defect in feeder lambs. In addition, the length of time required for cockle development and the length of time for the cockle defect to heal once an animal has been freed of keds is important to the feeder, packer, and tanner.

Eradication of keds from individual flocks is possible with conventional pyrethroid-based control methods. There is a need to pursue registration of newer pesticides and biorationals with alternative modes of action to delay the selection for insecticide resistance. With development of these new compounds the producer will have chemicals to rotate with the pyrethroids.

There is a need for extension educational programs dealing with control of sheep ked, particularly avoiding reinfestation of flocks that have been treated.

Sheep Bot Fly, *Oestrus ovis L.*

Description and Biology. The sheep bot fly is a major cosmopolitan parasite which adversely affects the efficiency of production by sheep and goats both directly and indirectly (Lloyd 1985). Rogers and Knapp (1973) and Lloyd and Brewer (1992) have presented the life history of the sheep bot fly in Kentucky and Wyoming, respectively.

The adult nose bot fly is about 1/2 inch long and superficially resembles a honey bee. The adult fly does not feed, however it is very annoying because it repeatedly flies at the face of the host in order to deposit its larvae. The larvae of the sheep bot fly live as parasites on the mucous surfaces of the nasal passages and sinuses of sheep and goats.

The first stage larva is deposited in the nostril of the host. The larva migrates through the nasal passages to a sinus cavity where it continues to feed until it is a fully developed, third state larva and is ready to leave the host and pupate. At this time they emerge from the sinuses and nasal passages and drop to the ground. The pupa is inactive for 3 weeks to a month depending upon soil temperature, and at the end of this period, the adult fly emerges from the pupal case, mates and begins to seek a host.

In Kentucky larvae may complete their development during a period of 4 to 6 weeks in the summer. In Wyoming the larvae overwinter in the first larval stage, and require a year for one generation.

Economic Importance. Damage by the sheep bot fly is two-fold, that produced by larvae living in the nasal passages and sinus cavities and that caused by the annoyance of the adult female fly. An external sign of larval infestation is the appearance of a runny nose which increases in severity as the infestation develops. The adult flies are very annoying, causing animals to stamp their feet, run, duck their heads, and rub their noses into the dust and hold them close to the ground. Adult flies interfere with the normal grazing and resting activities of animals.

It has been estimated that sheep bot fly larvae cause a 4 percent decrease in weight gain. Drummond, in 1981 estimated annual losses in sheep production in the United States due to sheep bot fly to be $13.5 million.

Meleney et al. (1962) reported that more than 91% of Southwestern sheep are infested with *O. ovis*. Lloyd and Brewer, in 1992, reported that over 90% of the sheep in Wyoming and neighboring states were infested.

Methods of Control. Control of the sheep bot fly is difficult because of the location of the parasite in the host. IVOMEC (Ivermectin) Sheep Drench, 0.08% solution, is currently the only product registered for control of the sheep bot fly. IVOMEC®sheep Drench also provides effective control of gastrointestinal roundworms and lungworms. The favored time for treatment is during late fall or early winter, after one or more killing frosts have eliminated adult flies. At this time, larvae harbored by sheep are predominately first instars and are found mostly on the nasal mucous membranes.

In areas of the country where this drug is used extensively for control of internal parasites, the sheep bot fly is reported to be very rare. Based on studies in New Mexico, it would appear that sheep bot can be controlled if all sheep in any given area are treated annually with an effective parasiticide (Meleney and Apodaca 1969, Meleney et al. 1963).

Research and Extension Needs. Basic research is needed on biology and behavior, in order to develop alternate, long-lasting controls. While there is currently good information on development of larval bots in a few areas of the country, information is needed in other regions of sheep production. More research is needed on the adult fly and we suggest studies on dispersal and attraction to the host animal.

Although the nasal bot is a common pest, and a concern to many sheep producers, we know little of its effect on the host. Damage thresholds are necessary for the establishment of economic thresholds. Improved methods of population monitoring may be necessary for an IPM program. Currently, animals are sacrificed so that the nasal passages and sinuses may be examined.

Currently one product is available for safe and effective control of sheep bot fly. Insecticides considered for future evaluation must be those used on other species of livestock because the market in the sheep industry would be relatively small. Research is needed on efficacy, toxicology and residues of candidate insecticides as well as research on formulation and application. We would recommend, also, research on other noninsecticidal factors such as those that might modify behavior of the adult flies.

Studies in New Mexico suggest that eradication of the bot from individual, isolated flocks may be a possibility. Because of the extreme efficacy of ivermectin against the larvae, it would appear that sheep bots might be eradicated from a given area if all sheep are treated annually.

Biting Midges: *Culicoides*, no-see-ums, biting gnats

Description and Biology. Biting midges are small biting flies of the family *Ceratopogonidae*. They are grayish to black in color and resemble black flies, but are smaller, 1.0 - 1.5 mm long, and often appear flat from the top as the wings, which are often mottled, are folded horizontally over the back when at rest.

Eggs are laid in areas of high organic content and moisture. The eggs hatch in a few days. The four larval instars last from a few weeks up to several months, depending on the species, and there may be one to many generations per year. There is no obligate diapause, but winters are generally spent as larvae, with pupation beginning in the spring as temperatures rise. Adults are present during all warm months until killing frosts.

Favored developmental sites of biting midges are slowly-moving streams, polluted ponds or animal waste lagoons, rotting vegetation, treeholes, or any standing water polluted over a long period of time with organic effluent (Blanton and Wirth 1979). The larvae can develop in a wide range of pH and in sunny and shaded conditions (Jones 1961, 1965). Livestock on pasture or rangelands often create favorable sites around watering holes, ponds, or overflowing stocktanks, and effluent holding ponds on confined livestock operations are particularly attractive to egg-laying females.

Economic importance. Biting midges are fierce biters, can cause severe allergic reactions, and are vectors of a variety of animal diseases. Although small, they are the most harmful of the biting flies affecting sheep. This is primarily due to one member of the *Culicoides variipennis* (Coquillett) species complex, *C. v. sonorensis*, currently considered a subspecies, and its transmission of 4 serotypes of the bluetongue (BLU) viruses (Jones and Foster 1971). Other species of *Culicoides* (and, in the West, *Leptoconops*) no doubt feed on and irritate sheep, but their vector capacity for bluetongue is unknown. Outbreaks of bluetongue have been recorded producing mortality rates in excess of one-third in susceptible flocks. Goats are apparently unaffected by the virus but serve as reservoir hosts (Luedke and Anakwenze 1972). Bluetongue has assumed a more important role in recent years, as it serves as a non-tariff trade barrier to the international movement of ruminant animals and their genetic byproducts (Jones et al. 1977). A recent proposal would regionalize the U.S. and create BLU-free zones to facilitate international trade (Walton et al. 1992).

The range of the *C. variipennis* complex is from Canada into Mexico (Wirth and Jones 1957). Bluetongue viruses can affect sheep in all areas within the range of *C. v. sonorensis*, and also infect cattle, deer, bison, bighorn sheep, elk, moose, antelope, and exotic ruminant zoo animals (Bowne et al. 1967; Trainer and Jochim 1969).

Methods of Control. Control technologies currently recommended are aimed primarily at the insect vector. Larviciding with granular formulations of temephos has been shown to be effective (Holbrook 1982) when applied to all known larval sites over a relatively large contiguous area (Holbrook 1984). Integrated management systems have been developed (Holbrook 1984; Mullens 1992). Control and monitoring efforts assimilated into ongoing mosquito control programs can be very cost-effective (Holbrook et al. 1994). Current recommendations also are directed at water and waste management, and avoidance of the periods when adult *C. v. sonorensis* are most active. Recently a mermithid nematode parasite has been shown to parasitize (and kill) over 50% of *C. v. sonorensis* larvae in some situations (Paine and Mullens 1994, Mullens and Lugring 1998), but the effect of this level of parasitism on bluetongue transmission remains to be documented. A vaccine that is licensed for use in California provides protection against serotype 10 (Jones et al. 1977). Survey tools and recommendations for use in monitoring adult populations are available (Holbrook 1985, 1994; Holbrook and Bobian 1989).

Research and Extension Needs. There is a need for research on the basic biology of biting midges. Resting sites, long-range dispersal, short-range directed flight activity, and characterization of developmental sites are all important (Jones et al. 1981). There needs to be continued elucidation of the makeup and distribution of the *C. variipennis* complex, and the relationships of the members of the complex to the distribution and transmission of bluetongue. The possibility of other *Culicoides* vector species, as this may relate to the transmission of both endemic and exotic bluetongue and other *Culicoides*-borne diseases of sheep, requires investigation. An example is the possible role of *C. insignis* in Florida, where BLU serotype 2 was introduced but does not appear to have become established. Studies on the interactions of midges and hosts, including potential chemical manipulation or repellent use, need to be conducted. Gene mapping, and an understanding of the genetics of vector competence for the BLU viruses and other native and exotic pathogens of sheep and goats is crucial. Basic studies on the occurrence of bluetongue, and its distribution and vector(s) should be encouraged in Mexico and Canada.

Data need to be collected on the use of new pesticides or pesticide formulations, delivery systems, attractants, repellents and growth regulators for adult and larval control. These than should be related to the goal of reducing vector capacity. The use of cultural methods needs exploration and exploitation, and naturally-occurring or genetically engineered biocontrol agents need to be screened, developed and tested. Integration of available control technologies into existing pest or disease management systems should be emphasized.

Extension personnel should be trained, and used to help determine the severity of the biting midge/BLU problem in the sheep-producing areas, and a rapid reporting system should be established to bring outbreaks or related problems to the attention of State and Federal action and research agents and agencies. Extension personnel should then educate sheep producers as to existing control measures and other resources that may be available.

Black flies: *Simulium arcticum* Malloch, *Simulium vitattum* Zett.

Description and Biology. Adult black flies, or buffalo gnats, are gray to black in color. These insects are generally 1-5 mm long. The first thoracic segment is enlarged giving the flies a hump-backed appearance, thus the name "buffalo gnat."

Eggs are laid either on aquatic plants or on stones beneath or near the water surface. The number of eggs varies from 100-500/batch and several batches are deposited in a lifetime. The eggs hatch in from 5-30 days, depending on water temperature, motion of the water, and the species of black fly. The larval period ranges from 12 days to 10 weeks, and some species overwinter as larvae. The pupal period ranges from 2 days to 4 weeks depending on the species. The entire life history--egg to adult--ranges from 20 days to 15 weeks and over. They usually disperse some 7-10 miles but some species have been collected up to 25 miles from the nearest breeding site.

Economic Importance. Very little information is available concerning black flies attacking sheep. Black fly attacks on other animals are commonly reported. Jessen (1977) summarized the literature as follows:

"Few blackfly problems associated with sheep have been reported in the literature. Fredeen (1969, 1974) reported that *Simulium arcticum* Malloch attacks in Alberta forced sheep to seek shelter. Rempal and Arnason (1947) reported that this species killed a few sheep in Saskatchewan in 1944; freshly shorn sheep were most seriously affected. Jones (1961) collected blackflies from sheep in Colorado, but did not mention a pest problem."

Another species, *Simulium vitattum* Zett., bothers sheep in the Northwest. This species does not feed much on sheep but its pestiferous behavior causes sheep to bunch and ceases feeding. Substantial reductions in weight gain of from 3-10 lbs/lamb have been reported (Jessen 1977). Also, where the sheep bunch on the ranges, range is destroyed.

Black flies are probably causing damage throughout the distribution areas of *S. arcticum* and *S. vitattum*. This area would cover much of the U.S. and, thus, a large problem could exist. Very little documentation is available at present, however.

Control Methods. Substantial efforts have been directed towards the development of management strategies employing microbial larvicides that are highly efficacious against black fly larvae yet are environmentally benign and have little or no toxicity to nontarget aquatic organisms. Investigations have been conducted assessing the efficacy of *Bacillus thuringiensis* var. *israelensis* de Barjac against *Simulium* sp. (Colbo and O'Brien 1984, Gaugler and Finney 1982, Lacey and Heitzman 1985, Merritt et al. 1989). This particular strain was determined to be relatively effective against black fly larvae when applied at the appropriate stream location and time. Assessment of a mosquito-pathogenic fungus *Culicynomyces claviosporus* against black fly was conducted by Gaugler and Jaronski (1983). Although highly virulent against mosquito larvae, this particular fungal isolate was not pathogenic against *S. vittatum* larvae.

Research and Extension Needs for IPM. Much basic biological information is lacking, for example, population dynamics, distance of insect movements, phenology, and behavior need to be further studied. Sampling methods near sheep need to be developed.

Attractants and/or repellents, as well as novel pesticides, should be investigated for control measures. Also, more emphasis should be placed on developing biological agents such as nematode parasites for larval control.

Ticks: lone star tick, *Amblyomma americanum* (Linnaeus), Gulf Coast tick, *Amblyomma maculatum*, Rocky Mountain wood tick, *Dermacentor andersoni* Stiles, American dog tick, *Dermacentor variabilis* (Say), black legged tick, *Ixodes scapularis* (Say), spinose ear tick *Otobius megnini* (Duges).

Description and Biology. Ticks are distinguished from insects by the presence of four pairs of legs, rather than the three pairs possessed by insects, in the nymphal and adult stages, which are the stages usually observed. There are a number of tick species which, under certain conditions, can become economically important ectoparasites of sheep and goats. The following brief life history of each of the species listed is taken from Strickland et al. (1976).

A. americanum is a 3-host tick with a wide host range. All parasitic stages can be found on large mammals, but generally the immature stages (larvae and nymphs) prefer to feed on birds and small mammals. Females lay up to 8,300 eggs over a 3-week period. Incubation period of the eggs is 23-117 days. Larvae require 3-9 days to engorge and an additional 8-26 days to molt to the nymphal stage. Nymphs engorge in 3-8 days and 13-46 days to molt to adults. Adults require 9-24 days to engorge and additional 5-13 days before oviposition begins. Unfed larvae may survive up to 9 months, whereas unfed nymphs and adults can survive for up to 16 and 14 months, respectively.

A. maculatum is a 3-host tick. The immature stages (nymphs and larvae) feed primarily on ground dwelling birds, although small mammals may also be parasitized. Large wild and domesticated mammals constitute the most common hosts for adult ticks. The rather limited seasonal activity of adults suggests that there is only a single life cycle completed each year under normal conditions. A single female may lay up to 18,000 eggs during a 2-11 week oviposition period. Incubation of eggs is 21-142 days. Larval engorgement and molting ranges from 5-71 days. Engorgement and detachment of females occurs at 14-18 days. Survival of unfed larvae may reach 6 months in duration, while nymphal survival may be slightly longer, Unfed adults can survive for up to 13 months.

Dermacentor andersoni is a 3-host tick. The immature stages engorge mainly on small rodents, while the adult hosts include a wide variety of wild and domesticated large mammals. This species may have an unusual life cycle. Generally, 1-2 years is necessary to complete the life cycle with the 1-year cycle occurring when the small animal host population is abundant. However, at higher altitude or at the more northern limits of its distribution, a 3-year life cycle is not uncommon in *D. andersoni*. Females lay up to 7,400 eggs over a 2-4 week interval with an incubation period for eggs of 15-51 days. Larval engorgement and molting ranges from 2-21 days, while the nymphal stage is completed in 3-19 days. Females engorge and detach in 8-17 days. Survival of unfed larvae, nymphs, and adults may extend for periods of up to 4, 10, and 20 months, respectively.

Dermacentor variabilis is a 3-host tick. The immature stages engorge mainly on small rodents, while the adult hosts include a wide variety of wild and domesticated mammals. Females lay approximately 6,500 eggs during the 14-32 day oviposition period. Incubation of eggs requires 26-57 days. Larval engorgement and molt may range from 3-247 days. Nymphal engorgement and molt requires 3-291 days for completion. The females engorge in 5-27 days. Survival of unfed stages is extremely long. Larval and nymphal survival ranges from 18-19 months, while unfed adults have been known to survive for up to 3 years.

Ixodes scapularis is a 3-host tick. The primary hosts for the immature stages are birds and small mammals, but even lizards may occasionally be parasitized. Adult host preferences include a variety of large wild and domesticated mammals, including attacks on humans. This species shows a distinct seasonality with the immature stages being most abundant in the spring and summer, whereas the adult stage is most abundant from late fall to spring. A single life cycle is completed during each year. Females lay approximately 3,000 eggs. The incubation period of eggs ranges from 48-135 days. Larvae engorge in 3-9 days, then require 22-49 days to molt. Nymphal engorgement and molting ranges from 3-56 days to complete. Females will reach repletion in 8-9 days before detachment occurs. Unfed larvae may survive more than 75 days, whereas unfed nymphal survival is slightly less (greater than 60 days). The survival period of unfed adults of this species has not yet been determined.

Otobius megnini is the sole member of the family *Argasidae* (soft ticks) that has been reported parasitizing sheep and goats. It is an aberrant 1-host tick. Only the larval and nymphal stages feed; the adult completes the life cycle on food obtained during the second nymphal stage. The last molt which produces the adult, occurs away from the host. Females lay up to 1,500 eggs in a period of 14-180 days. Incubation period of the eggs is completed in 10-23 days. Larval engorgement and molt occurs in 7-12 days.

Nymphal development, which includes two molts, occurs in 31-209 days. Unfed larvae may survive up to 80 days, while unmated females can survive up to 21 months.

Economic Importance. In areas where any of the tick species listed become numerous, they may become serious pests of sheep and goats. The economic impact of these species can be manifest in a number of ways. If infestation levels are heavy, they can result in significant blood loss in the host animals with *A. americanum* causing more problems because of its large size. "Tick worry" is another chronic problem that may be caused by any of these species which leads to loss of production in the host animal. "Tick paralysis" is another problem associated with several of the species listed. Another factor to be considered in the economic importance of these ticks is their ability to vector certain disease agents.

D. andersoni is the most important tick species of those listed that affect sheep and goats in the U.S. and Canada. This tick is distributed from extreme western Nebraska and South Dakota to the eastern slopes of the Cascades, and from northern Arizona and New Mexico well into British Columbia and Manitoba (Bishopp & Trembley 1945). The distribution accounts for this species being the most important tick on sheep and goats, because this is precisely the area of highest sheep production in the U.S. and Canada. The Rocky Mountain wood tick is an important ectoparasite in the cause of "tick paralysis" in sheep (Anonymous 1991). The malady may be the result of the bite of even a single tick and if not removed, may cause death of the host in 2-4 days. The tick may also become economically important as a vector of tularemia which can affect sheep (Philip et al. 1935).

D. variabilis is one of the most widely distributed tick species in North America, but primarily it is most abundant in the eastern half of the U.S. (Bishopp & Trembley 1945). It replaces *D. andersoni* as the chief species of concern on sheep and goats in the eastern U.S. Although not specifically reported in sheep, the American dog tick can cause "tick paralysis" and is also reported as a vector to tularemia in humans, which makes it a potentially economically important species in sheep (Anonymous 1991).

A. americanum is distributed throughout the southeastern U.S. It is abundant in states bordering the Gulf of Mexico, and along the Atlantic coast northward through Virginia and westward through Tennessee and Missouri (Bishopp & Trembley 1945). The lone star tick replaces *D. andersoni* as the species of economic importance on sheep in the areas where it is distributed. It is a large tick that in high numbers can cause heavy blood loss in infested hosts. The long mouthparts can cause painful, festering wounds that are predisposed to secondary infections. Even in moderate numbers, the presence of lone star ticks can cause "tick worry" in infested animals which can cause a dramatic loss in production because host animals may stop feeding. Although never reported in sheep, the lone star tick is a known vector of tularemia, thus it should not be discounted in this regard. In addition, it has also been known to cause "tick paralysis" in animals other than sheep (Anonymous 1991).

O. megnini is distributed primarily throughout the arid and semiarid areas of the southwest, although it has been widely dispersed to other areas including British Columbia (Bishopp & Trembley 1945). This distribution places it in areas of high sheep and goat production within the U.S. and Canada. The primary economic importance of this species lies in the fact that it can cause a high degree of "tick worry" among infested animals. By attaching deep in the ears, it can cause considerable pain and irritation. Loss of production in infested animals may result from restlessness, shaking and rubbing of the head, and may cause animals to stop feeding (Anonymous 1991).

A. maculatum is found in the southern states bordering the Gulf of Mexico and along the Atlantic coast (Bishopp and Trembly 1945), but sustaining populations have also been reported from eastern Oklahoma. This tick causes considerable "tick worry" to domesticated livestock, and massive infestations can result in great blood loss, debilitation, and even death. Although not reported specifically from sheep and goats, the Gulf Coast tick has been incriminated as a producer of tick paralysis in dogs and humans in the southeastern United States. Even though this species is less important as an economic pest than the other ticks listed, it does have the potential to produce economic losses in sheep and goats when populations are at a high level.

I. scapularis is distributed in the southeastern United States, westward to Texas and Oklahoma and along the Atlantic coast as far north as Massachusetts (Bishopp and Trembly 1945). The black-legged tick can be considered to be considerably less important as an economic pest of sheep and goats than the other species listed. However, the fact that it possesses a number of potential disease vectoring capabilities demands that consideration be given to its potential as an economic pest. The tick has been incriminated in the transmission of babesiosis to humans, as well as being naturally infected with tularemia, although natural transmission of tularemia has not been shown (Anonymous 1981). Perhaps the most important factor in attributing economic importance to the black-legged tick is the fact that it is a known competent vector of Lyme disease in humans.

Current Control Methods. Sheep may be dipped in approved organophosphate acaricides to kill ticks. Sheep may be sprayed with a variety of the "newer generation" pyrethroids for control of ticks and other ectoparasites.

In British Columbia, Canada lindane and phosmet are recommended for application (spray and pour-on) against *D. andersoni* and *O. megnini* (Costello 1986).

In Texas, the use of ivermectin applied as a daily oral does (20mg/kg) to Spanish goats provided ca. 95% control against the lone star tick (Miller et al. 1989). Thus, the use of avermectin pesticides may hold some promise as a possible method for controlling tick populations on sheep and goats.

Another chemical which is not registered for use on livestock at present, but one that could have potential for use in the future, is the phenylpyrazole agent, fipronil. Laboratory studies have shown that topical application of this pesticide provided an LC90 for psoroptic mange in sheep at a concentration of 10 ppm (Hunter et al. 1994). Field studies using a 1% fipronil pour-on formulation against cattle fever ticks on cattle produced 99% control (Davey et al. 1998).

To control "tick paralysis" it may be necessary to manually remove attached ticks, since the presence of even one tick can cause paralysis.

It should be noted that many of the chemicals and application methods used on cattle for the control of ticks may also be highly effective for use on sheep.

Research and Extension Needs. Although few studies have been conducted on the control of ticks specifically on sheep, there has been considerable research conducted on IPM tick control strategies using cattle as the model system. The applicability of most of these various tick control IPM strategies to the tick-sheep system model is a logical approach. However, there are several areas of research that must be conducted specifically on sheep to elucidate the information that is necessary to progress toward an integrated control approach of ticks on sheep.

Perhaps the most important research need is the establishment of economic injury levels of each of the tick species on sheep. Before IPM technologies can even be implemented, it must be known and established at what density level the ticks begin to adversely impact sheep health and well being.

Beyond the establishment of economic injury levels, research specifically on sheep must be conducted to advance technology on genetic and molecular control of ticks associated with sheep.

The primary areas of research on control components should be to evaluate treatment methodologies that are less stressful on the host animals than presently used technologies (dipping or spraying). Four-on, injections, and habitat modification would seem to be areas that offer the advantage of producing less stress while maintaining adequate levels of control.

There is a need for extension personnel to be informed and aware of the economic threshold levels and economic injury levels of ticks on sheep. Additionally, they should be aware of the various acaricides registered for use on sheep, as well as application techniques. This information can be disseminated to extension through periodic training sessions.

Mites: sheep scab mite, *Psoroptes ovis* (Hering), Chorioptic mange mite, *Chorioptes bovis* (Gerlach), rabbit ear mite, *Psoroptes cuniculi* (Delafond), sheep itch mite, *Psorergates ovis Wormersley*, goat follicle mite *Demodex caprae*.

Description, Biology, and Control. The eggs of *Psoroptes ovis* hatch 3-4 days after oviposition. The immature stage goes through 3 molts before becoming an adult. The life cycle from egg to egg takes 10-19 days under ideal conditions. Adult males inseminate pubescent female nymphs during their last molt. *P. ovis* has been found to remain infective in unoccupied corrals for three days. It is suggested that contaminated enclosures be left unoccupied for at least 2 weeks (Wilson et al. 1977).

The feeding of *P. ovis* causes the secretion of host fluids which accumulate to form scabs. These mites can cause extensive loss of wool and can cause death under heavy infestations. Scabbing makes shearing difficult.

P. ovis has been eradicated from domestic sheep in the United States since 1973 (Graham & Hourrigan 1977). They have been found in infected Rocky Mountain bighorn sheep in Idaho (Foreyt et al. 1985) and desert bighorn sheep in northwestern Arizona (Walsh & Bunch 1983).

Chorioptes bovis are surface dwelling mites that feed on dried epithelial cells and skin secretions. There life cycle is similar to *P. bovis* but last 18-28 days. They normally infest the lower legs, feet and face where little damage to the wool occurs. They are most severe when they infest the scrotum of rams.

C. bovis is the most common ectoparasite of sheep in the United States but causes little economic loss. *C. bovis* on Angora goats can be controlled with .05% Fenvalerate (Wright et al. 1988). Treatments for lice, keds, and *P. ovis* also control *C. bovis* on sheep.

The life cycle of *Psoroptes cuniculi* is similar to that of *P. ovis*. Infestations are common on dairy goats in the southwestern United States but the damage is slight due to usually low populations. They are more damaging on Angora goats due to the biting and licking of the mohair by the goats. Control is achieved by administering acaracides to the ear canal.

Psorergates ovis cause derangement of the fleece due to rubbing and biting by the sheep. They have been found beneath the surface of the outer stratum corneum and ingest epidermal lipids (Sinclair 1990). They are rare in the United States and are of little economic concern. They can be controlled by hot lime-sulfur, systemic and transepidermally administered acaricides (Sinclair 1990).

Demodex caprae infests the skin glands and hair follicles of goats. The infestation leads to the formation of nodules and damage to the skin. They have been found to be widespread on Michigan dairy goats (Williams & Williams 1982). It is possibly transmitted perinatally. There is no control.

Economic Importance. Mites that affect sheep and goats appear to be of little economic importance due to their rarity in the United States or due to the low magnitude of their infestations.

References Cited

Anonymous. 1991. Tick-transmitted diseases. Vet. Clinics North Amer.: Small Anim. Pract. 21: 45-47; 49-50; 125-127; 141-143.

Bedding, R. A., A. S. Molyneux and R. J. Akhurst. 1983. *Heterorhabditis* spp., *Neoaplectana spp* and *Steinernema kraussei*: interspecific and intraspecific differences for insects. Exp. Parasitol. 55: 249-257.

Bishopp, F. C. and H. L. Trembley. 1945. Distribution and hosts of certain North American ticks. J. Parasitol. 31: 1-54.

Blanton, F. W. and W. W. Wirth. 1979. The sand flies (*Culicoides*) of Florida, <u>In</u> Arthropods of Florida and neighboring land areas. Florida Dept. Agric. Consumer Serv., Gainesville. Vol. 10, 204 p.

Bowne, J. G., A. J. Luedke, M. M. Jochim. 1967. Bluetongue of sheep and cattle: past, present and future. J. Am. Vet. Med. Assoc. 151: 1801-3.

Chamberlain, W. F. and D. E. Hopkins. 1971. The synthetic juvenile hormones for control of *Bovicola limbata* on Angora goats. J. Econ. Entomol. 64: 1198-1199.

Chamberlain, W. F., D. E. Hopkins and A. R. Gingrich. 1976. Applications of insect growth regulators for control of Angora goats biting lice. Southwest. Entomol. 1: 1-8.

Colbo, M. H. and H. O'Brien. 1984. A pilot black fly (Diptera: Simuliidae) control program using *Bacillus thuringiensis* var *israelensis* in Newfoundland. Can. Ent. 116: 1085-1096.

Costello, R. 1986. Control of insects and ticks on livestock. Brit. Col. Min. Agric. & Fisheries. 20pp.

Darrow, D. E. 1973. Biting lice of goats: Control with dichlorvos-impregnated resin neck collars. J. Econ. Entomol. 66: 133-135.

Davey, R. B., E. H. Ahrens, J. E. George, J. S. Hunter, III and P. Jeannin. 1998. Therapeutic and persistent efficacy of fipronil against *Boophilus microplus* (Acari: Ixodidae) on cattle. Vet. Parasitol. 74:261-276.

Drummond, R. O., G. Lambert, H. E. Smalley, Jr. and C. E. Terrill. 1981. Estimated losses of livestock to pests. pp. 111-127. <u>in</u> Pimentel, D. ed. CRC Handbook of Pest Management in Agriculture, vol 1, CRC Press, Inc.

Drummond, R. O., J. E. George and S. E. Kunz. 1988. Control of Arthropod Pests of Livestock: A Review of Technology. CRC Press. Boca Rotan, Fla. p. 125.

East, I. J. and C. H. Eisemann. 1993. Vaccination against *Luciliacuprina*-the causative agent of sheep blowfly strike. Immunol. and Cell Biol. 71: 453-462.

Everett, A. L., I. H. Roberts, H. J. Willard, S. A. Apodaca, E. H. Bitcover and J. Naghski. 1969. The cause of cockle, a seasonal sheepskin defect, identified by infesting a test flock with keds (*Melophagus ovinus*). J. Am. Leather Chem. Assoc. 64: 460-476.

Everett, A. L., I. H. Roberts, and J. Naghski. 1971. Reduction in leather value and yields of meat and wool from sheep infested with keds. J. Am. Leather Chem. Assoc. 66: 118-132.

Foreyt, W. J., V. Coggins & T. Parker. 1985. *Psoroptes ovis* (Acarina: Psoroptidae), in a Rocky Mountain bighorn sheep (*Ovis canadensis canadensis*) in Idaho. J. Wildl. Dis. 2: 456-457.

Fredeen, F. J. H. 1969. Outbreaks of the black fly Simulium arcticum Malloch in Alberta. Quaestiones Entomol. 5: 341-372.

Fredeen, F. J. H. 1974. Tests with single injections of methoxychlor black fly (Diptera: Simuliidae) larvicides in large rivers. Can. Ent. 106: 285-305.

Fuchs, T. W. 1990. Timing of treatments for lice control on Angora goats. Proc. Livestock Insect Workers Conf. Ithaca, N.Y.

Fuchs, T. W. and M. Shelton. 1985. Effectiveness of new methods of biting lice control on Angora goats. Southwest. Entomol. 10(1): 15-19.

Gaugler, R. and J. R. Finney. 1982. A review of *Bacillusthuringiensis* var. *israelensis* (serotype 14) as a biological control agent of black flies (Simuliidae) Misc. Publ. Entomol. Soc. Am. 12: 1-7.

Gaugler, R. and S. Jaronski. 1983. Assessment of the mosquito-pathogenic fungus *Culicinomyces clavosporus* as a black fly (Diptera: Simuliidae) pathogen. J. Med. Entomol. 20: 575-6.

Graham, O. H. & J. L. Hourrigan. 1977. Eradication programs for the arthropod parasites of livestock. J. Med. Entomol. 13: 629-658.

Greenberg, B. 1991. Flies as forensic indicators. J. Med. Entomol. 28: 565-577.

Hall, D. G. 1948. The blowflies of North America. Vol. IV of the Thomas Say Foundation, Washington, DC 477 p.

Holbrook, F. R. 1982. Evaluations of three insecticides against colonized and field-collected larvae of *Culicoides variipennis* (Diptera: *Ceratopogonidae*) J. Econ. Entomol. 75: 736-7.

Holbrook, F. R. 1984. Progress in Clinical and Biological Research. Alan R. Liss, Inc., New York, 178:617-20.

Holbrook, F. R. 1985. Research on the control of bluetongue in livestock by vector suppression. Progress in Clin. and Biol. Res., Alan R. Liss, Inc., New York, 178: 617-20.

Holbrook, F. R. 1985. A comparison of three traps for adult *Culicoides variipennis* (*Ceratopogonidae). J. Am. Mosq. Contr. Assoc. 1: 379-81.*

Holbrook F. R. 1995. Collecting *Culicoides*. Bull. Soc. Vector Ecol. (In press).

Holbrook, F. R. and R. J. Bobian. 1989. Comparisons of light traps for monitoring adult *Culicoides variipennis* (Diptera: *Ceratopogonidae*). J. Am. Mosq. Contr. Assoc. 5: 558-62.

Holbrook, F. R., S. V. Romney and R. M. Sessions. 1993. Piggybacking bluetongue vector control and mosuito abatement. Wingbeats, Am. Mosq. Contr. Assoc. 4(4): 19-24.

Hunter, J. S., III, D. M. Keister and P. Jeannin. 1994. Fipronil: A new compound for animal health. Proc. Amer. Assoc. Vet. Parasitol. 39th Ann. Mtg. San Francisco, CA. Pg. 48.

Jessen, J. I. 1977. Black flies (Diptera: Sumuliidae) which affect sheep in southern Idaho. Dissertation Abstracts Intl. 38B, 3, 1041-42.

Jones, R. H. 1961. Observations on the larval habitats of some North American species of *Culicoides* (Diptera: *Ceratopogonidae*) Ann. Entomol. Soc. Am. 54: 702-10.

Jones, R. H. 1961. Some observations on biting flies attacking sheep. Mosquito News 21:113-15.

Jones, R. H. 1965. Epidemiological notes: incidence of *Culicoides variipennis* in an outbreak of bluetongue disease. Mosq. News 25: 217-8.

Jones, R. H. and N. M. Foster. 1971. The effect of repeated blood meals infective for bluetongue on the infection rate of *Culicoides variipennis*. J. Med. Entomol. 8: 499-500.

Jones, R. H., A. J. Luedke, T. E. Walton and H. E. Metcalf. 1981. Bluetongue in the United States: an entomological perspective toward control. World Anim. Rev. 38: 2-8.

Kimberling, C. V. 1988. Chapter 7: diseases of the respiratory system. In Jensen and Swift's diseases of sheep. 3rd ed., Lea and Febiger, Philadelphia.

Kotz, A. C. 1992. Effects of cyromazine on reproduction and offspring development of in *Lucilia cuprina* (Diptera: Calliphoridae). J. Econ. Entomol. 85: 1614-1617.

Lacey, L. A. and C. M. Heitzman. 1985. *Efficacy of flowable concentrate formulations of Bacillus thuringiensis* var. *israelensis* against black flies (Diptera: Simuliidae). J. Am. Mosq. Control Assoc. 1: 493-7.

Legg, D. E., R. Kumar, D. W. Watson and J. E. Lloyd. 1991. Seasonal movement and spatial distribution of the sheep ked (Diptera: *Hippoboscidae*) on Wyoming lambs. J. Econ. Entomol. 84: 1532-1539.

Lloyd, J. E., E. J. Olson and R. E. Pdadt. 1978. Low volume spraying of sheep to control the sheep ked. J. Econ. Entomol. 71: 548-550.

Lloyd, J. E., R. E. Pfadt and R. Kumar. 1982. Sheep ked control with pour-on applications of organophosphorus insecticides. J. Econ. Entomol. 75: 5-6.

Lloyd, J. E. 1985. Arthropod pests of sheep. pp 253-267. in Williams, R. E., R. D. Hall, A. B. Broce and P. J. Scholl, eds. Livestock Entomology. John Wiley.

Lloyd, J. E. and M. J. Brewer. 1992. Sheep bot fly biology and management. University of Wyoming, Cooperative Extension Service Bulletin B-966, 3 pp.

Luedke, A. J. and E. I. Anakwenze. 1972. Bluetongue virus in goats. Am. J. Vet. Res. 33: 1739-45.

Lupton, C. J., T. W. Fuchs and C. S. McCown. 1988. Effects of short-term lice infestation of Angora goats on objectively measurable mohair characteristics. PR-4586 IN Research Reports on sheep and Goat, Wool and Mohair, 1988. Texas Agric. Expt. Sta., Texas A&M University, College Station, TX.

Mahon, R. J., K. G. Wardhaugh, A. C. M. Vangewen and W. A. Whitbet. 1993. Reproductive development and survival of *Lucilia cuprina* Wiedemann when fed sheep dung containing ivermectin. Vet. Parasitol. 48: 193-204.

Meleney, W. P., N. G. Cobbett, and H. O. Peterson. 1962. Am. J. Vet. Res. 23(97): 1246-51.

Meleney, W. P., N. G. Cobbett & H. O. Peterson. 1963. Control of *Oestrus ovis* in sheep on an isolated range. J. Amer. Veterinary Med. Assoc. 143: 986-989.

Meleney, W. P. & S. A. Apodaca. 1969. Regeneration of a population of *Oestrus ovis* in sheep on an isolated range. J. Amer. Veterinary Med. Assoc. 155: 136-138.

Merritt, R. W., E. D. Walker, M. A. Wilzbach, K. W. Cummins, and W. T. Morgan. 1989. A broad evaluation of *B. t. i.* for black fly (Diptera: Simuliidae) control in a Michigan river: Efficacy, carry and nontarget effects on invertebrates and fish. J. Am. Mosq.

Miller, A. J., W. F. Chamberlain, and D. D. Oehler. 1985. Methods for control of the Angora goat biting louse. Southwest. Entomol. (10)3: 181-184.

Miller, A. J., G.I. Garris, J. E. George and D. D. Oehler. 1989. Control of lone star ticks (Acari: Ixodidae) on Spanish goats and white-tailed deer with orally administered ivermectin. J. Econ. Entomol. 82:1650-1656.

Mullens, B. A. 1992. Integrated management of *Culicoides variipennis*: a problem of applied ecology. Proc. 2nd Int. Sym. Bluetongue and Related Orbiviruses, CRC Press, Boca Raton, FL, 896-905.

Mullens, B. A. amd K. L. Luhring. 1998. Age-dependent parasitism of *Culicoides variipennis sonorensis* (Diptera: Ceratopogonidae) by *Heleidomermis magnapapula* (Nematoda: Mermithidae) and considerations for assesing parasite impact. Biol. Cont. 11:49-57.

Mullens, B. A. and J. L. Rodriguez. 1990. Cultural management of bluetongue virus vectors. California Agriculture 44:30-42.

Nelson, W. A. and S. B. Slen. 1968. Weight gains and wool growth in sheep infested with the sheep ked, *Melophagus ovinus*. Exp. Parasitol. 22: 223-226.

Nelson, W. A., S. B. Slen and E. C. Banky. 1957. Evaluation of methods of estimating populations of the sheep ked, *Melophagus ovinus* (L.) (Diptera: Hippoboscidae), on mature ewes and young lambs. Can. J. Anim. Sci. 37: 8-13.

Paine E. O. and B. A. Mullens. 1994. Distribution, seasonal occurrence, and patterns of parasitism of *Heleidomermis magnapapula* (Nematoda: Mermithidae), a parasite of *Culicoides variipennis* (Diptera: Ceratopogonidae) in California. Environ. Entomol. 23:154-160.

Peterson, H. A. and R. C. Bushland. 1956. Lice of sheep and goats. USDA Yearbook of Agriculture. 1956: 411-414.

Pfadt, R. E. 1976. Sheep ked populations on a small farm. J. Econ. Entomol. 69: 313-316.

Philip, C. B., W. L. Jellison and H. F. Wilkins. 1935. Epizootic tick-borne tularemia in sheep in Montana. Amer. Vet. Med. Assoc. 86: 726-744.

Pinnock, D. E. 1994. The use of Bacillus thuringiensis for control of pests of livestock. Agric., Ecosystems and Environ. 49:59-63.

Rempel, J. G. and A. D. Arnason. 1947. An account of three successive outbreaks of the black fly *Simulium arcticum,* a serious livestock pest in Saskatchewan. Sci. Agric. 27:428-45.

Rogers, C. E. and F. W. Knapp. 1973. Environ. Entomol. 2(1): 11-23.

Sinclair, A. 1990. The epidermal location and possible feeding site of *Psorergates ovis*, the sheep itch mite. Aust. Vet. J. 67: 59-62.

Strickland, R. K., R. R. Gerrish, J. L. Hourrigan and G. O. Schubert. 1976. Ticks of veterinary importance. USDA, Agric. Handbk. No. 485. 122pp.

Strickman, D., J. E. Lloyd and R. Kumar. 1984. Relocation of hosts by the sheep ked (Diptera: Hippoboscidae). J. Econ. Entomol. 77: 437-439.

Tabachnick, W. J. 1996. *Culicoides variipennis* and bluetongue-virus epidemiology in the United States. Ann. Rev. Entomol. 41:23-43.

Trainer, D. O. and M. M. Jochim. 1969. Serologic evidence of bluetongue in wild ruminants of North America. Am. J. Vet. Res. 30: 2001-11.

Vogt, W. G. and R. Morton. 1991. Estimation of population size and survival of sheep blowfly, *Lucilia cuprina*, in the field from serial recoveries of marked flies affected by weather, dispersal, and age-dependent trappability. Res. Popul. Ecol. 33: 141-163.

Wall, R., N. P. French and K. L. Morgan. 1993. Predicting the abundance of the blowfly *Lucilia sericata* (Diptera: Calliphoridae). Bull. Entomol. Res. 83: 431-436.

Walton, T. E., W. J. Tabachnick, L. H. Thompson and F. R. Holbrook. 1992. An entomologic and epidemiologic perspective for bluetongue regulatory changes for livestock movement from the USA, and observations on bluetongue in the Caribbean Region. Proc. 2nd Int. Sym. Bluetongue and Related Orbiviruses, CRC Press, Boca Raton, FL, 952-60.

Walsh, G. W. & T. D. Bunch. 1983. Psoroptic scabies in desert bighorn sheep (*Ovis canadensis nelsoni*) from northwestern Arizona. J. Wildl. Dis. 19:342-344.

Williams, J. F. & C. S. F. Williams. 1982. Demodicosis in dairy goats. J. Am. Vet. Med. Assoc. 180: 168-169.

Wilson, G. I., K. Blachut & I. H. Roberts. 1977. The infectivity of scabies (mange) mites, *Psoroptes ovis* (Acarina: Psoroptidae), to sheep in naturally contaminated enclosures. Research in Vet. Sci. 22: 292-297.

Wirth, W. W. and R. H. Jones. 1957. The North American subspecies of *Culicoides variipennis* (Diptera: *Helidae*) U. S. Dept. Agr. Tech. Bull. 1170, 35p.

Wright, F. C., F. S. Guillot & J. E. George. 1988. Efficacy of acaricides against chorioptic mange of Goats. Am. J. Vet. Res. 49: 903-904.

Zumpt, F. 1965. Myiasis in man and animals in the Old World. Butterworth, London. 267 p.

****
****

End of Sheep and Goats section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****
****

HORSES SUMMARY

1. The host-parasite relationships of pest flies and ectoparasites of horses need to be investigated. In particular, the populations of these arthropods should be determined in various regions, including seasonality, population fluctuations, weather effects, and premise environmental conditions.

2. Evaluate disease transmission potential to horses by vector species and by regions.

3. Sampling methods should be developed for each of these groups. Samples should be correlated with absolute populations on the animal.

4. Action thresholds, based on both economic considerations and aesthetic concerns, need to be developed.

5. Novel insecticides and biological control agents should be developed, along with innovative delivery systems for their application. Strategic placement should reduce environmental contamination and minimize human and animal exposure while targeting susceptible life stages.

6. Develop pest management strategies that are socially, environmentally and economically acceptable.

7. Natural and induced host resistance should be investigated.

8. The use of genetic manipulations (Fryxell & Miller 1995) should be investigated for ectoparasites and fly pests of horses.

9. Potential for eradication of each pest should be assessed.

10. Research needs to evaluate the correlation between area-wide pest populations and numbers of the pest found on animals.

11. Information on the biology, ecology and physiology of ectoparasites needs to be worked out for various regions of the country. Information needed includes seasonal locations on the animals, climate induced mortality factors, and the effect of host physiology, breed and diet on populations and on economic injury levels.

12. Host specificity of each group should be determined, along with the suitability of alternative hosts.

13. Species population dynamics in relationship to emergence, dispersal and host seeking by regions should be investigated.

14. Several of these pest species are shared with cattle, sheep, other livestock and wildlife. Coordinated efforts should be established with researchers on these other groups and truly integrated pest management strategies devised.

15. Because many of these pests are dealt with by veterinarians, collaborative efforts of entomologists and members of the veterinary profession should be undertaken.

HORSES

Committee Members

Nancy C. Hinkle, Chair
Philip J. Scholl
Donald E. Mock
William B. Warner

Economic Significance of Horses

The horse population in the United States is estimated between 5.3 (USDA-NAAS 1999) and 6.9 million (American Horse Council (AHC) 1998). This represents a marked decline from 1970s figures of 8.5 million to 10 million (Anon. 1979). Even with this decline, the American horse industry is estimated to contribute $25.3 billion in goods and services, with a total impact of $112.1 billion to the U.S. Gross Domestic Product (AHC 1998). The horse industry portion of the U.S. GDP is greater than that of the motion picture industry, railroad transportation, or tobacco products, and generates the equivalent of more than 1.4 million jobs across the U.S.

Approximately 1.9 million U.S. households, about 2% of all households, owned one or more horses in 1996, with 7.1 million Americans involved in the horse industry (AHC 1998). In 1993 approximately 10,000 sanctioned horse shows were held, generating $223 million. In 1992 approximately 71 million people visited U.S. race tracks and wagered an estimated $14 billion (AHC 1994).

Industry sources estimate that consumers spent between $40 million and $42 million for on-animal equine ectoparasiticides (including repellents) alone in 1997. The costs of prevention and treatment of arthropod borne diseases is no doubt many times this sum.

Arthropods have been implicated as major vectors of two of the top six equine infectious diseases identified by a recent survey of horse owners (USDA-APHIS 1997), with Equine Infectious Anemia ranking as the top infectious disease concern among horse owners.

Arthropod Pests of Horses

House Fly, *Musca domestica* L. (Muscidae)

Description and Biology. The house fly is a non-biting fly having sponging mouthparts. It is gray, 6 to 9 mm long, and has four dark stripes running lengthwise on its thoracic dorsum. This fly breeds in damp organic matter and thrives in horse manure. Each female fly will lay from 75 to 150 oval white eggs per laying, and up to 500-600 in her lifetime. Herms (1911) calculated an average of 685 larvae per pound of horse manure. Since the average horse produces about 40 pounds of manure per day, a single horse could potentially yield 27,400 larvae per day. With close to 5 million horses having the potential of producing this many larvae each day, the subsequent fly production from horse manure is astronomical.

Eggs of house flies hatch in 12-24 hours; the larvae undergo three instars, pupate, and adults emerge in about two weeks. The shortest time required for development may be as little as 7 days, depending on temperature. The determination of this minimum may be an important consideration in fly control.

Economic Importance. As discussed in the sections on cattle pests, the common house fly is of concern both for its annoyance and for its potential as a disease vector. Lawsuits brought by residents of homes bordering stables because of "fly bother" are an increasing concern to horse owners. Lack of fly control may also bring citations from local health officials and result in expensive acute control programs. The common house fly is of concern to both horses and humans living adjacent to horses. The house fly is found inside stables and on the animals themselves. If the farm is located close to residential areas it may be the source of serious legal action for nuisance, as well as providing a breeding habitat for potential mechanical vectors of human disease (Knapp 1985).

House fly populations are most damaging in the summer months with maximum numbers in August in the northern U.S. Beside annoyance, house flies are very important in the transmission of numerous equine pathogens as mechanical and biological vectors. A bimodal population curve is seen in the southeastern and southwestern states. Populations in the southeastern states reach damaging levels in late March with about 10 flies per animal and increase to 20 in November December. In the Southwest, high fly numbers are seen from spring through fall, with a mid-season drop in summer, probably due to the dry season.

House fly damage to horses is from annoyance caused by persistent feeding on the muzzles, eyes and wounds. Animals become nervous and restless, reducing food intake. House flies are also responsible for transmission of the roundworm parasite of horses, *Habronema* spp., serving both as intermediate hosts for the gastrointestinal forms and as transport hosts for the larvae that cause summer sores. It has also been shown that the house fly is capable of transmitting diseases such as bovine mastitis and pinkeye. In addition, house flies are known to be contaminated with more than 100 species of pathogenic organisms.

Methods of Control. Fly control around horse properties is similar to that discussed under other animal groups. Current methods are dependent upon sanitation, chemical and biological control. Sanitation and source reduction are the most effective methods of suppressing house fly production. Traps and insecticides are used extensively, with some success. Pupal parasitoids can help reduce adult emergence in certain situations, as discussed in the poultry section.

Several strains of *Bacillus thuringiensis* have been demonstrated to be effective against house fly larvae and some are in commercial development. Various microbes and their by-products are being investigated for potential use as feed throughs incorporated into horse feed for controlling flies in manure. Fungal pathogens such as Diptera-pathogenic strains of *Beauvaria bassiana* are also being commercialized.

Chemical controls may be applied as larvicides, baits, residual sprays, and space sprays. Feed-through larvicides (e.g. tetrachlorvinphos) may be more acceptable in non-food animals such as horses. They optimize chemical placement by limiting the amount that enters the environment while targeting potential fly larval development sites. Baits, likewise, target a specific life stage and minimize environmental contamination.

Residual sprays are normally applied to surfaces which flies frequently contact. These surfaces can be rafters, beams, structures or any place flies tend to rest. Space sprays are effective in quickly knocking down flies. Since there is no residual effect, insecticides applied in this manner must be applied frequently.

On-animal repellents are often the only efficacious control method where flies immigrating from neighboring facilities are the primary concern. The most effective products contain natural pyrethrins (usually with piperonyl butoxide or another synergist); however these products generally are efficacious for only a few hours to a day. Natural pyrethrins' repellency is extended to several days in products that combine a mixture of pyrethroids and natural pyrethrins/synergists, with other repellents (e.g. MGK 326, butoxypolypropylene glycol) and UV blocking agents. Some products containing high concentrations of permethrin (0.5% to 1%) provide good contact repellency, but do not provide the "vapor barrier" of natural pyrethrins.

Stable Fly, *Stomoxys calcitrans* (L.) (Muscidae)

Description and Biology. Stable flies cause irritation and weakness in animals and account for considerable blood loss in severe cases. Feeding wounds also can serve as sites for secondary infection. Discomfort from their bites interferes with feeding activities of animals. In heavily infested areas animals cannot be pastured during the day. These flies are easily interrupted in feeding and are mechanical transmitters of anthrax, equine infectious anemia (swamp fever), anaplasmosis, and stomach worms, *Habronema* spp. A list of diseases transmitted by *S. calcitrans* to horses and other animals can be found in the 1979 Workshop publication (Anonymous 1979).

The stable fly or dog fly is similar to the house fly in size and color, but its bayonet-like mouthparts differentiate it from the house fly. Both sexes are voracious blood feeders. They are strong fliers and may range many miles from the breeding sites.

Stable fly larvae develop in soggy hay, grain or feed, piles of moist fermenting weed or grass cuttings, ground-piled silage (Scholl et al. 1981), under large round hay bales (Hall et al. 1982), spilled green chop, peanut litter, and in manure mixed with hay. The female, when depositing eggs, will often crawl into loose material, placing the eggs in crevices. Each female may lay a total of 500-600 eggs in 4 separate clutches. Eggs hatch in 2 to 5 days and the newly hatched larvae bury themselves, begin to feed, and mature in 14 to 26 days. While the average life cycle is 28 days, this period will vary from 22 to 58 days, depending on weather conditions. Adult flies are capable of flying up to 80 miles and may be carried hundreds of miles by weather fronts (Hogsette and Ruff 1985).

Economic Importance. This fly is a pest in every region of the country, causing economic damage to horses in every state. Economic effects of stable flies for horses include the diseases they transmit, injuries the animals suffer due to irritation of fly feeding, and the legal consequences of "fly bother" lawsuits (Skoda and Thomas 1992). As backyard composting of grass clippings increases, such sources of stable flies will become more significant, especially in urban and suburban areas.

Methods of Control. Stable flies often are produced in staggering numbers in wet organic matter on the premises; therefore, control at any facility implies source reduction through sanitation (elimination of breeding sites) and water management. After breeding sites are controlled, other suppression tactics may be used to decrease animal annoyance. Use of on-animal repellents, especially when applied to the front legs, is the most effective available method of chemical control. Individual animal spray treatments such as pyrethrin-synergist-pyrethroid combinations provide effective protection for hours to several days. A limitation of pyrethroids is that label restrictions allow their application no more frequently than every two weeks yet, in most circumstances, they do not provide protection from flies for that length of time. The most effective available method of chemical control is the application of persistent residual insecticides to vertical stable fly resting surfaces near animals (Knapp 1985). This method of control often has a very limited chance of protecting the animals at the premises as stable flies often disperse considerable distances after feeding (Broce 1993). As discussed in the cattle sections, trapping and biological control may provide useful ancillary tactics against stable flies.

Horn Fly, *Haematobia irritans* (L.) (Muscidae)

Description and Biology. The horn fly is a small bloodsucking fly about 4 mm long or about one-half the size of a house fly. It resembles the stable fly except that it is small, less robust, has a relatively heavier labium and its palpi are almost as long as its proboscis. Its primary host is cattle. It feeds 24-38 times per day (Harris et al. 1974), causing continuous annoyance. This fly deposits its relatively large eggs (1.3-1.5 mm long) only in freshly deposited cow manure. A single female is capable of laying approximately 400 eggs during her lifetime. Under most conditions, the eggs hatch within 24 hours and the larval stage, which develops in the manure, is usually completed within 4-8 days. Last instar larvae crawl to drier areas, most often under the manure pat, where they pupate. Adults emerge under most conditions within 6-8 more days. They overwinter as diapausing pupae.

Economic Importance. The horn fly (*Haematobia irritans*) is said to be an obligate blood-sucking parasite of cattle. Horn flies become pests of horses when newly emerged horn flies, in their quest for a bovine host, encounter horses instead (Greer and Butler 1973). In the absence of bovine hosts, they may continue to feed on the horses for several days. Horn flies also become pests of horses to the point of interfering with their performance as work animals when the horses are being ridden around horn fly infested cattle. Populations on horses are never as high as those observed on cattle; two hundred horn flies per animal is a heavy infestation on horses. There has been no research to establish the economic impact of horn flies on horses; however, they cause considerable irritation to work or pleasure animals even in very small populations, i.e. 10-25 flies constitutes a significant population. This fly occurs throughout the tropical, subtropical and temperate areas of the northern hemisphere, however their intensity and pest potential varies considerably from one geographical area to another. Host-parasite interaction causes damage in feeding areas, especially along the ventral midline. Damage is seen as open sores and may result in secondary infection and scar tissue.

Methods of Control. The employment of a good horn fly control program on cattle in the vicinity of where horses are stabled or worked is an effective method of reducing or preventing annoyance to horses by horn flies. However, since horn fly control is not practiced by all cattle producers, occasional pest populations on horses will continue to occur. Horn fly populations on animals may be controlled by insecticide treatments. Light spraying of horses with several of the commercially available pyrethrin-synergist combinations or wipe-on applications will protect horses from annoyance when the horses are used to work horn fly infested cattle. The pyrethroids, when labeled for application on horses, will provide extended protection of horses from horn flies.

Face Fly, *Musca autumnalis* DeGeer (Muscidae)

Description and Biology. The face fly has been found in most states with the exception of Alaska, Arizona, Florida, Hawaii, New Mexico, and Texas (Loomis et al. 1975). The face fly has oviposition habits similar to the horn fly. It deposits its eggs in fresh cow manure and the larvae develop in the pat. Mature maggots migrate from the pat and pupate to grey puparia. The total cycle from egg to adult takes 8 to 25 days. Female face flies lay about 200 to 300 eggs in their lifetime (Pickens and Miller 1980). The face fly is about the size and color of a house fly, with a distinctive tuft of setae under the wings. Face flies feed on lacrimal tissue, mucus, or blood from cuts or wounds. They have sponging mouthparts like the house fly; however the microstructure is rough and fitted for rasping tissue, causing irritation and additional lacrimation. Their aggressive feeding habits make them dangerous vectors of disease, as well as producing extreme annoyance to animals (Smith et al. 1966, Smith and Linsdale 1967, Greenberg 1971). The face fly avoids shade, preferring open sunlight, causing animals to avoid sunny areas where excessive fly numbers are present.

The overwintering flies enter a true diapause as a consequence of the exposure of adults that are less than 2 days old to a combination of low temperatures (<16oC) and short days (Stoffolano 1968, Valder et al. 1969). The flies overwinter in barns and buildings, or under tree bark and have been reported to enter houses in such numbers that they become nuisances to homeowners.

Economic Importance. These flies probe the mucous tissue of the eyes, producing an irritation that increases the flow of liquid from the eye. Horses attacked by face flies become nervous, spend their time in deep shade and may lose weight from the constant irritation. Blindness as a result of secondary invasion by various organisms can result. The face fly also serves as an intermediate host and vector of the *Thelazia* sp. eyeworm.

Methods of Control. Because face flies develop on pastured cattle and only secondarily affect horses, suppression of fly populations is seldom within the control of horse owners (Pickens and Miller 1980). Preventing flies from bothering the horses, either mechanically or chemically, is the goal of the horse owner. Exclusionary devices such as fly masks can be useful in keeping face flies from feeding in ocular areas.

Horse Flies and Deer Flies (Tabanidae)

Description and Biology. Horse flies are one of the larger and more diverse groups of Diptera. They are strong fliers, vicious biters, and notorious pests of most livestock. They range in size from slightly smaller than house flies to one inch or more in length. Adult tabanids are intermittent feeders, causing considerable irritation and blood loss from their painful bites, as blood continues to flow after the flies leave.

The larvae overwinter in muddy soils, maturing in the late spring and pupating in dry soil. Adults appear in early summer with males feeding on nectar and females feeding on blood. Eggs are deposited on leaves or stems of aquatic plants. Horse flies occur throughout the United States wherever horses and wooded areas are in close association. Because tabanids are found throughout the country, horses in all states are subject to horse fly attack.

Economic Importance. Horses may lose more than 3 ounces of blood a day to horse fly feeding, resulting in anemia. Horse flies are vectors of equine infectious anemia in the United States and are carriers of surra, a trypanosome disease, in South America. Because of the large amount of blood they take and the intermittent nature of feeding, it is expected that they can physically transmit any blood disease of horses if the right circumstances exist (Krinsky 1976).

Tabanid bites may cause nodular reactions and depigmentation as well as resulting in horses becoming very nervous and unmanageable. Horse flies are extremely irritating to horses and their presence and feeding activities make riding impossible or dangerous in areas with large populations. Their significance as pests of humans is exemplified by the effect "greenheads" and other coastal species have on tourist activity on beaches.

Methods of Control. Control of immature stages of horse flies is not practical (Foil and Hogsette 1994). Area control of adult populations is effective in reducing populations; however flies can reinfest horses from surrounding areas within 2-4 days. Consequently, this type of treatment is seldom if ever cost effective for horses. Pasturing horses away from wooded areas in more exposed, breezy sections of pastures aids in reducing tabanid attacks. Individual animal treatments with pyrethrin based repellent sprays or wipes effectively protect horses for hours to several days.

Mosquitoes (Culicidae)

Description and Biology. Mosquitoes cause severe blood loss and annoyance to horses. In some areas, literally thousands of mosquitoes feed upon a horse during one 24-hour period. Populations may be so heavy as to obstruct the nostrils of the animals. Additionally, mosquitoes throughout the United States are potential vectors of some of the most dreaded diseases of horses. The majority of these diseases also affect humans.

There are many species of mosquitoes that feed on horses. Each species displays its own life history peculiarities. Female mosquitoes lay their eggs in an aquatic environment, such as in shallow pools or in low areas that will be subsequently inundated. The larval stages are aquatic, feeding on small animals and particulate organic matter in the water. They are able to survive only in shallow water free from wave action. The length of the larval period may vary considerably depending upon the species, water temperature, etc., but in summer, is often completed in 6 or 7 days. Pupae (or "wrigglers") are also aquatic and mobile. The adults emerge to become blood-feeding parasites, primarily of warm-blooded animals. Most species are not specific as to blood source and will feed on many host species. Most mosquitoes will feed several times during their lifetime. Some feed primarily in the immediate area where they emerged while others are capable of traveling great distances to find a host.

Economic Importance. Mosquitoes feed in great numbers. The annoyance and blood loss associated with feeding produces significant economic losses and sometimes death (Bishopp 1933). Mosquitoes are of great importance because of their role as vectors of eastern equine encephalitis, St. Louis encephalitis, western equine encephalitis, Venezuelan equine encephalomyelitis and swamp fever. There were an estimated 1 million cases of equine encephalitis resulting in 300,000 equine deaths from 1930 to 1935 (Shahan and Giltner 1945). Equine encephalitis is a widespread disease. For example, it was reported in 25 states in 1935 with death rates running from 10-95% (Shahan and Giltner 1945). It is of tremendous importance in some areas with as many as 10% of the equine population being infected and death rates approaching 90% (Kissling and Rubin 1950).

The importance of equine encephalitis was evidenced by the furor ensuing during an outbreak of Venezuelan equine encephalomyelitis in Texasin 1971. Within 35 days, 2.8 million horses were immunized against the disease and all horses in six states were quarantined. Suppression techniques directed at adult mosquitoes while the immunization program was conducted involved ULV spraying of 13.5 million acres. Approximately 7,500 persons were involved and $15 million was spent during this short period (Phelps 1971, Zehmer et al. 1971, USDA 1972, 1973).

The equine encephalomyelitis complex is also a serious disease of humans that generally builds up during the early summer in horses before it becomes a serious threat to humans. Two and one half percent of horses tested in one survey were positive for this mosquito-borne disease of horses in 1974 (personal communication National Cattlemen's Association staff 1974).

Methods of Control. Mosquito control often involves several suppression techniques as adult mosquitoes may fly considerable distances. Control efforts of individual producers can be effective, but most often are not cost-effective. Mosquito control often involves large-scale operations directed at elimination of larval production areas, abatement of the immature forms and chemical treatment of adult mosquito populations. The size of the areas that must be treated requires organized efforts. These efforts are usually funded, administered and performed by local districts or sometimes state mosquito abatement organizations. These organizations provide efficient mosquito control because they have trained personnel, proper equipment, knowledge of the mosquito fauna and expert use of pre- and post-treatment monitoring techniques. Steelman and Schilling (1977) found that cost-effective protection of cattle was obtained by organized mosquito control programs, whereas the cost of control by aerial or ground ULV treatments by individuals against adults cost more than the economic benefit obtained.

Elimination or routine treatment of larval breeding areas on the horse premises and treatment of weeds and brush near stabled horses are necessary owner contributions and responsibilities without which protection may be impossible. Treatment of local mosquito breeding habitats with *Bacillus thuringiensis israelensis (Bti)*, and other microbial or IGR based larvicides may greatly reduce subsequent adult mosquito populations (Mulla et al. 1985).

Aerial applications of ULV insecticides (usually malathion or pyrethroids) to large areas of pasture land are effective in killing the adult mosquitoes present, and are occasionally economical in livestock production. However, treated areas may be rapidly inundated within 3-4 days with immigrating mosquitoes. These types of treatments are generally uneconomical for protection of horses

Applications of repellent-insecticide combinations are generally effective in preventing mosquito feeding for only a few hours. DEET may severely irritate the skin of some horses and so is not used in equine formulations. Webb and Knapp (1993) showed oil of citronella formulations to be moderately effective mosquito repellents on horses for up to 48 hours.

Currently there is no economical method of effectively controlling mosquitoes by treatment of horses, except as provided by organized mosquito control organizations, whose efforts are primarily to prevent annoyance and disease transmission to humans. The most widely used owner applied method of reducing the level of mosquitoes feeding on horses is the routine application of one of the many commercially available repellent-insecticide combinations to the horses. These applications are effective for a few hours up to two days.

Biting Midges, *Culicoides* spp., *Leptoconops* spp. (Ceratopogonidae)

Description and Biology. These are small (2 mm or less in length) blood-sucking flies that transmit disease organisms to horses as well as producing irritation via their biting activity. They are known as biting midges, punkies, or no-see-ums. Immature stages of these flies are associated with wet or semi-aquatic habitats, such as the mud or moist soil around streams, ponds, sloughs, and marshes.

Their larvae develop in decaying leaves, in small standing bodies of water, and in wet sand or mud. This stage has blood gills to facilitate respiration and the pupal stage also occurs in moist locations. Massive adult emergences may occur, with adults feeding most intensively in the evening (Snow et al. 1958). One to several generations may be produced per year. The life cycles of these flies are largely unknown (Blanton and Wirth

1979). The few species intensively studied breed in saturated soil of swamps or beaches, or in tree holes or other container habitats.

Economic Importance. Ceratopogonoid flies are vicious biters, which cause irritation and annoyance, and may transmit the viral disease bluetongue to both sheep and cattle (Bowne et al. 1966, Luedke et al. 1967). *Culicoides* transmit five species of filarial worms to man and his animals, including *Onchocerca cervicalis,* associated with fistulous withers in horses (Rabalais and Votava 1974) and are suspect as a biological vector of VEE (Jones et al. 1972). Queensland itch is an equine allergic dermatitis resulting from bites of *Culicoides* (Riek 1954, Linley 1985). These insects feed in large numbers where they occur. Their painful bites cause horses to be very nervous and may interrupt the hosts' feeding. Ceratopogonid hypersensitivity, also known as sweet itch or summer itch, is the most serious equine dermatological condition caused by ectoparasites (McMullan 1993). Its symptomatology includes severe seasonal pruritus, alopecia, and abrasions at the mane, tail and ears (dorsal form) or under the belly, chest and groin (ventral form).

These insects are of widespread importance in the United States, especially in the southern United States.

Methods of Control. Some control can be achieved by habitat drainage or modification. This is risky, however, as conditions are often made better for other species. Biting midges are small enough to go through standard window screen. Environmental modification such as making banks of stock tanks and ponds steep sided reduces *Culicoides* breeding habitat (Mullens 1989). Many ceratopogonids only reluctantly enter structures, so stabling horses during crepuscular hours may provide relief. Because ceratopogonids are often very localized, simply pasturing horses a few hundred meters away from infested habitats greatly reduces the frequency of attack. Pyrethrin or pyrethroid on-animal fly sprays can be effective in reducing ceratopogonid bites on treated horses.

Eye Gnats, *Hippelates* spp.

Description and Biology. Eye gnats, *Hippelates pusio*, and other species are common small flies seen around the faces, genitals, and open wounds of horses throughout the summer months. The larvae of many *Hippelates* develop in soil high in organic matter (Harwood and James 1979).

These flies feed with sponging mouthparts which have the labella provided with spines that can cut the ocular surface. Feeding damage and fly worry are the primary problems. These flies are also vectors of all the diseases which can be mechanically transmitted across membranes and into open wounds.

Eye gnat populations in the southeastern states commonly run 100 per animal during the summer months. Reports of landing rates of 600 per minute in the eyes of humans have been made by the Florida State Board of Health.

Economic Importance. This species is considered a filth fly around horse establishments. It produces direct feeding damage on horses and indirect problems in urban areas because of human annoyance.

Methods of Control. Face masks and other physical barriers provide protection from eye gnat annoyance. Equine fly repellent products may be efficacious against eye gnats if properly applied to facial areas. One commercially available fly bait is labelled for eye gnats.

Black Flies (Simuliidae)

Description and Biology. Black flies are day feeding or crepuscular, blood-sucking flies that cause pronounced tissue irritation and itching to horses. The pain, itching and resultant local swelling, together with occasional severe complications, suggest that an allergen is injected while the flies feed. Deaths due to feeding have been recorded in livestock. Toxemia or anaphylactic shock along with loss of blood and suffocation by inhalation of flies are the major contributing factors. Species of major concern on horses in the United States are *S. vittatum*, which is a problem throughout the country, *S. arcticum* Malloch, important in the western United States and Canada, and *Cnephia pecuarum*, which is a severe problem in the Mississippi River Valley (Harwood and James 1979).

Black flies lay their eggs in association with running water. The eggs hatch in varying periods of time, depending upon the fly species involved and the prevailing environmental conditions. Most univoltine species of temperate regions pass the winter as diapausing eggs. The larva lives in moving water with high dissolved oxygen content. It maintains its position by means of a caudal sucker with which it fastens itself to stationary, submerged objects. Larval food consists of small crustacea, protozoa, algae, bacteria,and particulate organic material. The pupal stage is also aquatic, and may be quite short, but its duration is dependent upon species and water temperature. The adults are strong fliers and after emergence may fly for distances of 10 or more miles to find a warm-blooded host.

Economic Importance. Black flies may be severe problems of horses in some areas as certain species readily feed on horses. The location of feeding on the animal is species specific; some feed on body areas where the hair covering is sparse, some by crawling down in the mantle, and some by feeding in precise locations such as inside the ears. The bites cause severe irritation, itching, and lesions that often persist for several days to months. Feeding within the ears may cause scabbing and thickening of the tissue (Townsend et al. 1977) to the point of occluding the auricular canal. Feeding of *Simulium vittatum* Zetterstedt on horses and mules interferes with farm operations (Snow et al. 1958), and the feeding activities make horses fractious and difficult to use as pleasure animals (Townsend and Turner 1976). Large numbers of animal deaths, mainly in cattle but including horses, have resulted from feeding of large black fly populations.

Black flies on horses are important throughout the nation; however, the most severe attacks, i.e., ones resulting in death of animals, occur most often in the Mississippi River Valley and in the Rocky Mountains. *S. vittatum* annoys horses across the country, especially near larger, slow moving rivers.

Methods of Control. Area control directed at adult black flies is impractical for protecting horses. Methods that have proven effective in reducing black fly attacks on horses are larviciding of the streams where they are produced, especially using Bacillus thuringiensis israelensis, and individual animal treatment, specifically ear treatment. Black flies are repelled by natural pyrethrins and pyrethroid-based equine fly repellents. Townsend and Turner (1976) reported that applications of 5 gm of petroleum jelly per ear gave 3 days of protection from ear feeding by *S. vittatum*.

Screwworm, *Cochliomyia hominivorax* (Coquerel) (Calliphoridae)

Description and Biology. The screwworm adult is a deep greenish-blue, metallic colored blow fly, with yellow, orange or reddish face. This fly which is larger and more robust than the common house fly has three dark stripes on the dorsal surface of the thorax.

The adult lays eggs in characteristic "shingled" batches of 10 to 193 eggs at each site of a wound on an animal. A total of 2,800 eggs may be laid by one female (Laake 1936; Harwood and James 1979). The eggs hatch within 11 to 21.5 hours. The larval period lasts for about 3.5 to 5 days, depending on temperature and moisture conditions. The prepupal period requires from a few hours to 3 days and the pupal period lasts for 7 days (DeVaney and Garcia 1975). So, under natural conditions, one generation occurs about every 24 days (Harwood and James 1979).

The adult female screwworm fly is strongly attracted to wounds and sores of animals where she deposits large numbers of eggs (Bishopp 1926). The larvae enter the wound and feed on the healthy tissue. Feeding results in an ever deepening, tortuous cavity which exudes a foul smelling exudate that is even more attractive to other ovipositing flies than the original wound, thus promoting continued reinfestation until the animal is either treated or is killed by the infestation.

Economic Importance. Until the 1950s, the primary screwworm fly (*Cochliomyia hominivorax* Coquerel) was the major cause of myiasis in man and animals in North America. In the 1970s this insect was eradicated from the United States using the sterile male technique, along with complementary strategies (Bushland 1974). Prior to eradication, the annual loss produced by screwworms in all livestock in the southwestern states was estimated at over $100 million (Baumhover 1966, Galloway 1972).

Methods of Control. The sterile male release technique has eradicated this pest from the United States, and continues to push its northern range southward toward the planned eradication maintenance area in Panama. Inspection of imported animals and constant vigilance is necessary to prevent its reintroduction.

The Common Horse Bot Fly, *Gasterophilus intestinalis* (DeGeer), the Throat Bot Fly, *G. nasalis* (L.), and the Horse Nose Fly, *G. haemorrhoidalis* (L.)

Description and Biology. In general, the adult horse bot fly is about 3/5 inch long and is covered with fine brown hairs. Wings are marked by a blackish cross-band. The female fly has an elongated tubular ovipositor whereas the male has a blunt abdomen and is provided with a pair of claspers. Mating occurs at specific aggregation sites near the horses (Catts 1979). The horse bot fly has rudimentary mouthparts. It is active on bright sunny days, depositing eggs on the hair of the horse. Location of egg oviposition varies with the species involved. The number of eggs per female varies between species and within species. These eggs can be viable for several months, especially in cool weather. The eggs of the horse bot fly hatch with the aid of moisture from the horse's tongue whereas the other species' eggs hatch without external stimulation by the host. All larvae are similar and possess various numbers of rows of spines, depending on the species, and end up attached to various parts of the horse's stomach. After about 10 to 11 months, the mature larva passes out with the feces, pupates, and emerges in the summer to repeat the cycle (Broce 1985).

Seasonal distribution varies somewhat from north to south and apparently depends on the fall fly activity before the first killing frost, which stops egg laying.

Economic Importance. Horses throughout the United States are exposed to bot flies, necessitating treatment at least once a year. *G. intestinalis*, the common horse bot fly, and *G. nasalis,* the throat horse bot fly, occur throughout the United States. *G. haemorrhoidalis*, the horse nose bot fly, is found rarely in the Northwestern and Midwestern states.

The horse bot fly damages horses both directly and indirectly. Animals under attack may inflict damage on themselves or on anyone trying to handle them. Fright caused by egg laying adults may result in animals going out of control (Knapp 1985).

The common horse bot is the least annoying of the three species, due to adults laying eggs on the horse away from the head. Horses can be seen attempting to avoid these flies by standing end to end in an effort to protect each other. This behavior limits their grazing, causing them to lose weight and suffer other health effects.

Newly hatched bots produce a severe irritation as they burrow into the horse's tongue, gums, or lips. To relieve irritation, horses may rub or bite on objects, thus injuring themselves.

Direct damage is produced by larvae feeding on the tissue of the horse. The first instar larva enters the mouth tissue, causing irritation (Cogley 1989, Cogley et al. 1982). Work by Nelson (1952) and Tolliver et al. (1974) showed that the first instar larvae are involved in necrosis and formation of pus pockets in the periodontal spaces of the mouth. Damage in the stomach includes peritonitis, ulceration (Dart et al. 1987), obstruction of the flow of food from the stomach to the intestine, and irritation of the stomach due to bot attachment to the lining (Drudge et al. 1975, Shefstad 1978). Bots may cause rupture of the stomach and death of the horse (Blagburn et al. 1991).

Human myiasis caused by *G. intestinalis* has been reviewed by James (1948) and Dove (1937).

Methods of Control. The most effective treatment for horse bots has been treatment with one of the available endectocides (Bello 1989). Several organophosphates are also available for use against horse bots (Bauer and Burger 1986, Drummond 1963, Foil and Foil 1988, Foil and Foil 1990, Frahm 1983, Muylle et al. 1979, Seibert et al. 1986). Less effective control of horse bots can also be achieved by breaking the life cycle of the fly. Insecticides are labeled for external treatment in a warm water wash after eggs have been laid but before hatching. For external insecticide treatment, a warm water wash (110-120oF) should be rubbed or sponged on areas infested with eggs. The larvae will hatch and die upon contact with the insecticide. Treatments should be applied weekly during peak oviposition periods (August-September). Care should be taken to protect hands from insecticide and larvae with rubber gloves during the wash. Grooming may aid in removal of eggs but effectiveness of control is questionable. Prior to the advent of modern chemical techniques many purgatives had been devised for removal of the bot larvae from the horse's digestive tract, such as oil of chenopodium, turpentine, or linseed oil (Belding 1942). Many organophosphate insecticides have been tested in the treatment of *G. intestinalis* infestations. Extensive studies have been carried out, especially by Drummond (1963). Suggested controls include grooming the horse to remove the eggs or using warm water to stimulate hatch of *G. intestinalis* eggs (Sukhapesna et al. 1975). Commercial bot egg removal devices consisting of abrasive blocks and a lubricant solution have also been used to remove unhatched eggs. Keeping horses in a dark barn during the day and allowing them to graze at night will help limit their exposure to ovipositing flies. Treatment of choice is one of the new endectocides or an organophosphate insecticide either as a drench, paste or via the feed. Treatment is usually against the larval stage within the horse (Blagburn et al. 1991).

Cattle Grubs, *Hypoderma lineatum* (Villers) and *H. bovis* (L.)

Description and Biology. Cattle grubs are the larval stage of oestrid flies, the adults of which are known as heel flies, warble flies, bomb flies, or gad flies (Scholl 1993). The second and third larval stages produce furuncles or "warbles" under the skin along the backline of host animals in late winter to early spring. There are two species in the United States. In the U.S. the common cattle grub, *Hypoderma lineatum* (Villers), ranges from northern Mexico to northern Canada, while *Hypoderma bovis* (DeGeer), is found north of a line running west to east from northern California through Kansas to the Carolinas (Bishopp et al. 1926).

Hypoderma adults are 13 mm (*H. lineatum*) or 15 mm (*H. bovis*) long flies that are covered with dense hair arranged in alternating transverse bands. They somewhat resemble small bumble bees.

First stage larvae migrate through various body tissues for about 8 months of the year. When they become encysted in the back, the characteristic lesion they produce is the first indication of infestation. The two species can easily be distinguished in advanced larval stages by examination of the spination on the last two body segments, the last two being devoid of spines in *H. bovis* and only the last one being without spines in *H. lineatum.*

Economic Importance. Cattle grubs are primary, obligate parasites of bovines (Broce 1985). Occasionally the larvae infest hosts other than cattle. Infestations of horses (Baker and Monlux 1939, McDougall 1895, Pillers 1923, Scharff 1973) and humans have been reported. No estimates of the incidence of infestation in horses are available. Infestations do cause irritation to the horses and concern to the owners. The advanced stage larva characteristically opens a breathing hole in the region along the back line of the animal, which results in a cyst being formed around the parasite. The major concern of cattle grub infestation of horses is the presence of these cysts in the area occupied by the saddle or the harness. Few if any cattle grubs are able to complete the life cycle in the abnormal horse host (Scharff 1973) and die within the cyst. As they do not emerge normally, the lesion may persist for longer time periods than is seen in the normal host, resulting in long periods when horses cannot be used as work animals.

The migration route of larval stages through the horse has not been determined. As the horse is an abnormal host, considerable variation from that recorded in cattle may occur. *Hypoderma* spp. larvae have not been observed to reach maturity in the horse, usually being killed by the horse's immune system in the second instar (Knapp 1985). Acute neurological disease and death associated with intracranial migration of the first instar larvae of *H. bovis* in horses (Hadlow et al. 1977, Olander 1967) has been reported. Based on the low populations seen in horses and the lack of information on the effects of migration, it must be concluded that the major damage to horses is the production of cysts in the region of the back. While these cysts may become infected and produce secondary complications, the major effect is the interference with harness or saddle usage. No information on the national or regional importance of cattle grubs in horses is available.

Methods of Control. Probably the best method of treatment for cattle grub larvae encysted in the backs of horses is to gently squeeze the grubs out of the cyst through the breathing holes. The breathing hole should be slightly enlarged with a scalpel to prevent crushing the grubs within the cyst. The area should be disinfected subsequent to removal and then periodically examined for complications. Use of hydrogen peroxide to disinfect may also assist in removal of the larva itself (Scholl and Barrett 1986). Most horses are sensitive to application of systemic insecticides used freely for cattle grub control in cattle. Considering the exceptional control of *Hypoderma* at all stages by treatment with avermectin products (Scholl 1993), standard treatment of the normal equine parasite spectrum with one of these products should resolve the problem.

Area-wide control efforts against the cattle grub have been concerned primarily with cattle. One such program was attempted from 1982 to 1986 in northern Montana and southern Alberta, Canada which combined treatment of all cattle in a large region with release of sterile adults (Scholl et al. 1986). Effective control resulted in much lower infestation levels one or two years following treatment. Cattle grub adults do not fly great distances from where they emerge; therefore, such cooperative efforts of several producers on a county or district level are effective. Such efforts would greatly reduce the incidence of occurrence of cattle grubs in horses.

Despite the limited success of this project, grub control in horses will probably remain a concern of individual horse owners. The widespread use of endectocide products in horses for other parasite control programs will certainly impact the incidence of *Hypoderma*.

Biting louse, *Trichodectes* (Werneckiella) *equi* (Denny); Sucking louse, *Haematopinus asini* (L.) (Haematopinidae)

Description and Biology. Horses, mules, asses and zebras are occasionally infested with lice. There are two species that attack horses (Hall 1917), H*aematopinus asini* (L.) and *Trichodectes equi* (Denny) (Moreby 1978). Heavy populations of blood feeding *H. asini* may seriously weaken the horse, resulting in anemia and stunting of growth. *T. equi* is a chewing louse that does not feed on blood, but its activities on the animal cause irritation (Egri et al. 1995). Infestation with either species can result in loss of condition, alopecia, and open sores from rubbing the affected areas. Horses that are well fed and maintained are less likely to support populations of lice.

Lice are permanent parasites of their hosts, spending the entire life cycle on the host. Most species live only a short time off the animal and are not found on other species of animals. Egg production for different species varies from 50-100 eggs over 4-5 weeks.

Haematopinus asini, the horse sucking louse, found on domestic horse and zebras (Murray 1957) will live only for 2-3 days off the host. The life cycle for this louse takes 4 to 5 weeks for completion from egg to egg with 5 to 14 days required for egg hatch. The female louse deposits 50 to 100 eggs during her lifetime. The horse sucking louse, which typically reaches damaging numbers in the winter, may be found anywhere on the body, but is most common on the head, neck, back and inner surface of the thighs.

The biting or dermal feeding lice on horses include *Trichodectes equi* (Denny) (*Werneckiella equi*) (Moreby 1978). This louse feeds on exudates and dermal debris (Hall 1917), causing itching, irritation and hair loss. The life cycle for these lice takes 27-30 days for development from egg to adult and about 7 days for the eggs to hatch. These lice are most prevalent on the flanks, side of neck, tail base and shoulder area on finer body hairs (Murray 1957). These are usually winter parasites; however, they may be found in

Florida in damaging populations at any time of the year. W. equi has been incriminated as the vector of such diseases as infectious anemia (Moreby 1978).

Lice are transferred from animal to animal by contact, movement on flies and occasionally by contaminated equipment and bedding. All animals should be checked periodically for infestations and all animals in infested premises should be treated for good louse control. Retreatment of animals is required for good control because insecticides will not kill the eggs. With 2 weeks between treatments of biting lice and 3-4 weeks between treatments for sucking lice, good control will be achieved. Both species of horse lice oversummer in low numbers on the host. Populations build up as the weather gets colder, and peak populations are reached in the winter.

Economic Importance. Heavy louse infestations are usually seen in the winter and may cause anemia, unthriftiness, loss of condition, stunting of growth, loss of hair, and even sores from rubbing of the irritation. Infested animals are often difficult to handle. It is estimated that approximately 5% of pastured horses may have visually detectable lice populations during the winter months. Both species of horse lice are potential pests throughout the United States.

Methods of Control. Lice infested horses may be treated with water dilutions of registered insecticides. Many treatments are not effective against eggs so the animals will likely require retreatment at 2-3 week intervals. Control of lice requires nozzle pressure sufficient to provide complete wetting of the skin. Extralabel use of ivermectin products may be effective against lice. Infested animals and their tack should be quarantined from uninfested animals until the infestation is eliminated. Severe louse infestations are frequently indications of predisposing conditions such as immunodeficiency; the underlying condition should be addressed as well.

Mites, *Sarcoptes scabiei* var. *equi* Gerlach (Sarcoptidae), *Psoroptes equi* (Raspail) (Psoroptidae), *Chorioptes bovis* var. *equi* (Her.) and *Demodex equi* Railliet (Demodicidae)

Description and Biology. Mange, itch, or scab are names given to a group of contagious skin diseases of horses caused by small parasitic mites which live within or on the skin of the host. The most common of these is *Sarcoptes scabiei equi* which produces sarcoptic mange. Others are *Psoroptes equi* (Raspail), producing psoroptic mange; *Chorioptes bovis equi* (Her.), producing chorioptic mange; and *Demodex equi* Railliet, producing demodectic mange.

Sarcoptic mites excavate tunnels in the skin in which they deposit their eggs. Developing larvae and nymphs may continue these tunnels or crawl to the surface of the skin. The burrowing activities cause watery papules which break and form small scabs at the site of the infestation (Imes 1926). The presence of these mites is very irritating to infected animals.

Psoroptic mites live on the surface of the skin (Patil-Kulkarni et al. 1967). They do not burrow as do sarcoptic mites. Their feeding causes an exudate that hardens to cover the feeding mites. Most of the mites feed at the edge of the scab, constantly enlarging the affected area. The infestation spreads rapidly, causing intense itching.

Infestations of chorioptic mange are generally restricted to the lower legs, particularly around the top of the hoof. The lesions produced are similar to that of psoroptic mange; however chorioptic mange spreads much slower and is less extensive.

Demodectic mites are the most common mites found in horses in some areas. They live under the skin and are much smaller than the scab-producing mites of horses. *Demodex* does not produce scabs in horses but produces lumps or knots that form just under the skin, resulting from pus due to mite activity. Occasionally these burst and ooze, but are usually absorbed by the body. *Demodex* infestations are normally undetected in horses.

All the mites found on horses spend their entire life on the host. The eggs are laid in burrows (sarcoptic and demodectic mange) or on the hair (psoroptic mange). The eggs hatch in a short period of time, for example in 1-3 days for *Sarcoptes* and *Psoroptes*. The life cycle requires about 2 weeks, 9 days, and 3-4 weeks for sarcoptic, psoroptic and chorioptic mites of horses, respectively. Very little is known about the life history, transmission or control of demodectic mange in horses.

Methods of Control. Extralabel use of ivermectin has proven efficacious against these mites, as well as many other equine parasites. Infested animals and their tack should be quarantined from uninfested animals until the infestation is eliminated.

Ticks (Ixodidae and Argasidae)

Indigenous species:
Anocentor (Dermacentor) nitens (Neumann), tropical horse tick
Amblyomma americanum (L.), lone star tick
Amblyomma cajennense (Fabricius), Cayenne tick
Amblyomma maculatum Koch, Gulf Coast tick
Boophilus annulatus (Say), cattle fever tick
Boophilus microplus (Canestrini), Tropical cattle tick
Dermacentor albipictus (Packard), winter tick
Dermacentor andersoni Stiles, Rocky Mountain wood tick
Dermacentor occidentalis Marx, Pacific Coast tick
Dermacentor variabilis (Say), American dog tick
Ixodes pacificus (Cooley & Kohls) western blacklegged tick
Ixodes scapularis Say, blacklegged tick
Otobius megnini (Duges), spinose ear tick
Rhipicephalus sanguineus (Latreille), brown dog tick

Exotic species:

Amblyomma hebraeum Koch, bont tick
Rhipicephalus evertsi Neumann, red tick

Description and Biology. Ticks of the family Ixodidae are called hard-bodied ticks because of the nature of the scutum. The ticks referred to in this section are all ixodids except *O. megnini* which is in the soft-bodied tick family, Argasidae.

Ticks range in length (adult) from 1 mm to 10 mm with the average being about 4 mm. The body of ticks is not divided into obvious sections as in spiders or insects. However, there are three morphologically discrete body regions, the gnathosoma (mouth parts), podosoma (area where the legs attach) and opisthosoma (area posterior to the legs).

Ixodid ticks feed only once at each life stage with a molt occurring after each of the larval and nymphal feedings. Each female hard tick can deposit from 2,000 to 8,000 eggs. After feeding, the adult female moves away from the host and oviposits under ground litter. Development from egg to adult can be accomplished in as little as 60 days or require as long as 2 years depending upon species, environmental conditions, and availability of suitable hosts.

The life cycle of hard ticks may be 1-host, 2-host or 3-host type with the last type being the most common. *Anocentor nitens*, *Boophilus* spp., *Dermacentor albipictus* and *Otobius megnini* are all 1-host ticks and are considered to be the primary tick pests of horses. *Amblyomma americanum*, *Dermacentor andersoni*, *Dermacentor variabilis*, and *Ixodes scapularis* should certainly be added to the list of ticks frequently causing significant problems on American horses. Other species, regionally or locally, also may be important. The type of life cycle is of fundamental importance in a control program.

Amblyomma americanum (L.), the lone star tick, has a wide host range, including the horse. This tick is found from Texas to southeastern Nebraska and in a wide belt to the Atlantic coast. *A. americanum* is a three-host tick with each life stage utilizing a new mammalian host. After adult females mate and engorge they drop to the soil and deposit 3,000 to 5,000 eggs. This tick usually has one generation per year, typically being most abundant in the spring and summer. In warmer climates, however, all stages of development may occur throughout the year. In Kansas, adults may quest as early as February in warm winters and as late as October; nymphs are found from March through mid-November, and larvae mostly in July and August but occasionally until late October (D.E. Mock, unpublished).

The mouthparts of this tick are capable of deep penetration and make the bite painful to man and animal. Deep suppurating sores may form at the site of attachment, resulting from secondary infection. All stages of lone star ticks may infest any part of the horse's skin but are usually more abundant on thinner-skinned sites, and the juveniles are most likely to bite while still on the legs.

A. americanum is an important vector of the tularemia pathogen to humans (Goddard 1993) and the primary vector of *Ehrlichia chaffeensis* (Anderson et al. 1991, 1993; Lockhart et al. 1996). Several authors independently suspected lone star ticks of transmitting *Borrelia burgdorferi* under natural conditions (Schulze et al. 1984, Masters 1990, Luckhart et al. 1991, Fehrenbach 1992, Mock et al. 1992), but *B. lonestari*, provisionally described as a new species, may be the etiological agent of a Lyme-like disease transmitted by *A. americanum* (Barbour et al. 1996).

Amblyomma cajennense (Fabricius), the cayenne tick, is limited in its U.S. distribution to southern Texas. It is widespread throughout Mexico, Central America, South America and the Caribbean. The adults occur primarily on horses and cattle. They prefer to attach between the legs or on the abdomen. However, on equine hosts, all stages of the tick are frequently found inside the ears and other natural cavities, as well as on the flanks, withers, mane and tail.

Amblyomma hebraeum Koch, the bont tick, is a native of Africa and thrives in a warm, moderately humid climate. It has a number of hosts, cattle being the principal one, but it is also found on horses and other large herbivores. This tick has not yet been reported to be established in the U.S. but several specimens have been taken from the ears of rhinoceroses in two places in the U.S.

The bont tick is a three-host tick with the adults attaching primarily to hairless areas such as the groin, axilla, on the genitals, the teats and udder, under the tail, inside the ears, and beneath the fetlocks. The resulting lesions caused by these ticks are believed to provide entrance to the organisms causing ulcerative and epizootic lymphangitis of horses.

Amblyomma maculatum Koch, the Gulf Coast tick, is well named since it lives only in regions of high temperature, rainfall and humidity and was, historically, found only along the Gulf of Mexico. Conventional wisdom has it that *A. maculatum* is established only within 200 miles of the Gulf Coast. However, an apparently established population near Tulsa, Oklahoma was reported as early as 1944 (Bishopp and Trembley 1945), and the species became a severe pest of cattle in localized areas of northeastern Oklahoma by the late 1960s (Bolte and Coppock 1988). Hall et al. (1992) collected *A. maculatum* in Missouri, for the first time, in 1991. During the 1990s, evidence has been gathered of *A. maculatum* being established in dense foci in several counties of Kansas, up to 600 miles from the Gulf Coast (D.E. Mock, unpublished).

This pest is also a three-host tick, with larvae and nymphs attacking ground-inhabiting birds and adults parasitizing larger animals, including horses. This tick is most frequently found in the concave portion of the pinna, causing the ear to swell which may provide suitable conditions for screwworm attack. In addition, infested horses frequently become "head-shy" and difficult to handle.

Boophilus annulatus (Say), the cattle fever tick, and *Boophilus microplus* (Canestrini), the tropical cattle tick, are two species no longer established in the U.S. but that have the potential of being extremely important. The cattle fever tick was for years a primary problem in Texas, New Mexico, Oklahoma, Colorado, and Kansas because of its capability to vector *Babesia bigemina*, the causal agent of bovine piroplasmosis. The tropical cattle tick is of more importance to horse breeders and owners since it is said to vector *Babesia equi*, causal agent of equine piroplasmosis.

Both ticks are one-host ticks, with all life stages occurring on a single host. The nymphs and adults of both species are found on the flank and belly of the host. The larvae of cattle fever ticks are usually found on the legs, belly, and dewlap, while those of the tropical cattle tick are usually found in the ears. In heavy infestations, the ticks may be found over the entire animal. The parasitic portion of the life span of both species is between 20 and 66 days with a complete life cycle requiring 40 to 300 days.

These ticks have been eliminated from the United States except for occasional foci along the Texas-Mexico border. They pose an ever present problem because both are endemic to Mexico. While movement of these animals from Mexico to the United States is restricted, and dipping is required of all imported animals, smuggling and random drift of cattle, horses and deer back and forth across the Rio Grande border provides adequate opportunity for reinfestation of the border counties of Texas. The border counties of Texas do become infested with these pests from time to time, but constant surveillance, the use of a "buffer" zone and early detection have limited the movement of them into the rest of the country. This task becomes more and more difficult with the movement of large numbers of animals together with the rapid transit available, and will likely become even more difficult as a result of international trade and open borders.

Dermacentor albipictus (Packard), the winter tick, is a one-host tick found throughout the boreal zone of Canada, across the northern portion of the United States from Maine to Washington, and throughout the western states to Texas. Its habitat is restricted to the woods and shrubs of the upland and mountainous parts of the country. The preferred hosts are horse, elk, and moose, but it has been taken from most of the larger herbivores and carnivores.

For attachment sites, the tick prefers the belly and areas between the legs, but in heavy infestations may be found anywhere on the animal. This parasite can complete its life cycle in as few as 71 days or it may take nearly two years under unfavorable conditions. Usually the tick will have two or more generations per year. Not all horses are equally susceptible to attack by the winter tick. Those that are very susceptible will usually carry heavy infestations in the fall and winter. At that time, the hair coat is heavy and long and the infestations may not be easily seen.

The symptoms of heavy infestation are expressed by loss of appetite, depression and eventually debilitation. The loss of blood from severe infestations may weaken the animal or even result in death. An edematous condition known as "water belly" may also result. *D. albipictus* is a vector of anaplasmosis and is suspected of being a vector of Rocky Mountain spotted fever.

Dermacentor andersoni Stiles, the Rocky Mountain wood tick, occurs from British Columbia to west Texas in the Rocky Mountains. This tick is a three-host tick and has a life cycle which usually spans two years, but may be as long as three. It and the American dog tick are the primary vectors of *Rickettsia rickettsii*, the causal agent of Rocky Mountain spotted fever of man (Burgdorfer 1975) and can transmit the causative agent of tularemia (Goddard 1993). *D. andersoni* is not known to be a vector of disease to horses.

The toxin from the bite of this tick can cause paralysis in equines and ultimately death by starvation if the ticks are not removed. Tick bite paralysis is especially important in range animals that are allowed to run free a good portion of the season.

Anocentor nitens Neumann, the tropical horse tick, is a one-host tick usually found in the ears of horses. It occurs in southern Texas and Florida and throughout Mexico, the Caribbean Islands, Central and South America.

The preferred hosts of this parasite are the horse, mule and ass, but it is also found on sheep, goats, deer and cattle. While it prefers the ear as a site of attachment, it may also be found in the nasal diverticula, on the mane and belly, in the anal and inguinal regions and, in heavy infestations, on virtually any part of the body.

This tick is a vector of equine piroplasmosis and so is potentially a serious threat to the equine population. In addition, during engorgement, the female tick voids large quantities of feces in which the male becomes embedded and dies. This causes suppuration and predisposes the animals to screwworm attack. The ears get very tender, the animals become "head shy" and are extremely hard to manage.

Dermacentor occidentalis Marx, the Pacific Coast tick, is very closely related to *D. andersoni* but is found only in the narrow land strip between the Sierra Nevada mountains and the Pacific Ocean in Oregon and California. It is frequently reported as causing tick paralysis and its bite causes local inflammation. *D. occidentalis* is a three-host tick, and only the adults are found on horses.

Dermacentor variabilis (Say), the American dog tick, is widely distributed in the eastern two-thirds of the U.S. from Maine to the Gulf Coast and also is found commonly in Oregon and California. It utilizes a diverse array of mammalian hosts including cattle, horses, deer, dogs, cats, and man. *D. variabilis* is a three-host tick, and only the adults are found on horses. It causes annoyance to domestic animals, but is not a known natural vector of animal disease (Anonymous 1979) except possibly Rocky Mountain spotted fever in dogs (Teel 1985). *D. variabilis* is an important vector of Rocky Mountain spotted fever to humans (Burgdorfer 1975) and can cause tick paralysis in humans (Gothe et al. 1979), dogs, and cats. Parasitism by large numbers of *D. variabilis* may result in extreme emaciation of host animals.

Carroll and Schmidtmann (1986) reported infestations of American dog ticks on horses in Maryland to be of high frequency but low density. They found 78% of the ticks were attached to the tail and 12% in the mane.

Ixodes scapularis Say, the blacklegged tick, has long been found from southern Massachusetts to Florida, and westward across the southern half of the states to Louisiana, eastern Texas, eastern Oklahoma, and Mexico. Literature on *Ixodes dammini* Spielman, Clifford, Piesman & Corwin (mostly published from 1979 to 1993, and much of it relating in one way or another to concerns about Lyme disease) actually relates to *I. scapularis* (Oliver et al. 1993, Keirans et al. 1996). Lyme disease related research in the 1980s and 1990s showed an expanded range in the north to include Ontario, Wisconsin, eastern Iowa and northern Illinois and Indiana, and in the south into eastern portions of Kansas (Keirans et al. 1996). I. scapularis is a three host tick with the adults in greatest abundance in late winter and early spring. This species has been reported on horses (Hooker et al. 1912, Cooley and Kohls 1945). Data presented by Bishopp and Trembley (1945) show adult *I. scapularis* common on horses, and all stages were found on horses in Oklahoma (Clymer et al. 1970). Normal areas of attachment on deer include the ears, head and neck, but on horses more than 90% attach on the lower side of the body, notably the chest, underside of chin, axillae of legs, and inguinal region (Schmidtmann et al. 1998).

This tick is annoying to its hosts and is a known vector of *Borrelia burgdorferi*, the causative agent of Lyme disease. Burgess et al. (1986) reported Lyme disease in a Wisconsin pony. Later research showed a high prevalence of Wisconsin horses were antibody-positive for *B. burgdorferi* and presented with either subclinical or clinical Lyme disease (Burgess 1988). Richter et al. (1996) found *Ixodes pacificus* (Cooley & Kohls) to be a common parasite of horses in northern California and demonstrated its role in transmitting *Ehrlichia equi*, the causative agent of equine granulocytic ehrlichiosis.

Otobius megnini (Duges), the spinose ear tick, is a native of North and Central America but has been dispersed to many other localities in the world. It has been found in South America, South Africa, India, and Hawaii. This tick is found primarily in areas of low humidity and is most abundant in the southwestern part of the U.S.

This is a one-host tick and differs from the other ticks discussed in that only the larvae and nymphs attach to the host, with the adult female mating and ovipositing on the ground. This pest also differs from other ticks because the eggs are laid in small batches and may be deposited over a period of 6 months or more.

The spinose ear tick is a serious pest of horses and cattle. Its attachment, deep in the ear, causes considerable pain and irritation and the resultant wound predisposes the animal to screwworm attack. This attack may, of course, result in disfigurement, loss of the ear, or even death.

Secondary infection may cause perforation of the eardrum and infection of the middle and inner ear resulting in deafness or, in extreme cases, death of the animal from meningitis. Horses infested with ear ticks become unmanageable and can cause serious injury to themselves in their attempts to alleviate the irritation.

The effects of these ticks are especially important in the spring in emaciated horses and in cattle following drought periods. Death losses have resulted from a combination of screwworm attack and secondary infections.

Rhipicephalus evertsi Neumann, the red tick or redlegged tick, is an African species which displays broad adaptiveness. It is found on that continent in all types of forest and grassland, in coastal areas and plains from the Sudan to South Africa. It is apparently limited in distribution to areas with rainfall above 10 to 15 inches rather than by altitude or by low temperature conditions.

The red tick was found in Florida and New York on a number of wild animals on game farms in 1960. From the variety of animals infested and the number of ticks found, it is conceivable that the tick had been established in Florida for several years. Quarantines and cleanup procedures were imposed on both places and elimination was achieved in January 1962.

Horses and other members of the family Equidae are the preferred hosts, but all stages of the tick infest a total of 14 other large game and domestic species. It is a two-host tick with the larvae and nymphs generally clustered in the inner convolutions of the ear canal. The adults typically attach between the hind legs, on the scrotum, and in the perianal area, often hidden in the skin folds or on the membrane of the anus.

This tick is reported to transmit equine piroplasmosis and spirochetosis in horses, mules and donkeys.

Rhipicephalus sanguineus (Latreille), the brown dog tick, is probably the most widely distributed tick species in the world, aside from *Argas persicus* (Oken), the fowl tick. *R. sanguineus* is a native of Africa, but its preference for domestic dogs and for birds has helped it spread throughout the warmer parts of the world. Host records from prior to the 1960s should be accepted cautiously. Morel and Vassiliades (1962) argued persuasively that ticks from Africa and the Mediterranean area identified as *R. sanguineus* actually comprised a complex of five separate species.

R. sanguineus attaches primarily to dogs in the United States, but it has been reported on 33 other hosts. It is a three-host tick, with the larvae attached most often in hairy places on the body of domestic animals. The nymphs attach anywhere on the animals, and the adults are found frequently on and in the ears, although they may also be found along the nape and underside of the neck. The life cycle can be completed in as little as 63 days, but in the northern edges of its distribution, it usually takes a year or more to complete. This species maintains populations on unhoused dogs as far north as Wichita, Kansas, but probably not any farther north (D.E. Mock, unpublished).

R. sanguineus is potentially important to horses because it is a vector of *Borrelia theileri*, causative agent for spirochetosis in sheep, goats, cattle, and horses, but there are no records of *R. sanguineus* having parasitized horses in North America.

Brown dog ticks in the U.S. bite people more frequently than much of the literature would indicate. Goddard (1989) reported on a focus of this phenomenon in Texas and Oklahoma, and Carpenter et al. (1990) reported additional instances. Mock (unpublished) has several records of such bites from Kansas and South Dakota. *R. sanguineus* is most likely to bite people who have a lot of contact with infested dogs or who live in an infested home from which the preferred host has been removed. *R. sanguineus* has long been known as the vector of *Ehrlichia canis* to dogs and is the suspected primary vector of human ehrlichiosis. In humans the disease agent was reported as *E. canis* by Conrad (1989) and Barton and Foy (1989). Additional cases were quickly reported by Dimmitt et al. (1989) and Petersen et al. (1989). These cases are considered to have been caused by a new species of etiological agent, *E. chaffeensis*, of which *A. americanum* is the primary vector (Anderson et al. 1991, 1993; Lockhart et al. 1996).

Economic Importance. In the United States horses serve as hosts for two primary tick species and thirteen secondary species (Anon. 1979). Some of these species are widely distributed whereas others are restricted to certain geographical situations.

Ticks are of importance because they are obligate ectoparasites and transmit diseases to both humans and animals. Ticks are second only to mosquitoes as transmitters of horse diseases. They transmit protozoa, viruses, bacteria, rickettsia and toxins (Graham and Hourrigan 1977).

Ticks are widely distributed throughout the world because of their ability to survive adverse conditions. About 10 percent of the approximately 800 known species of ticks are established in the United States. Of the 80 or so species found in the United States, about 20 are of some veterinary importance (Strickland et al. 1976). No estimate is available for the economic injury of ticks to horses, nor of the diseases they vector.

Methods of Control. Tick control has been attempted primarily with acaricides as space sprays, body sprays, dips and insecticidal ear tags. Ivermectins have shown promise against some species. Additional methods which complement the action of acaricides are hormonal control, pasture resting, environmental alteration and selective trapping. Biological control has been investigated periodically in the past and is currently being scrutinized by several labs in the United States. The success of the screwworm project resulted in investigating the feasibility of using the sterile male release with ticks.

The historical review of tick control (Anon. 1979) reveals that chemical suppression has been the primary strategy and that it has been directed principally at cattle. Occasionally, as with the discovery of ticks' role in transmitting the agents of Rocky Mountain spotted fever and Lyme borreliosis, major research effort is directed toward tick suppression to prevent tick parasitism of humans. Research directed specifically toward protecting horses from ticks has been the infrequent exception. Such strategies as host animal resistance (either innate or induced), environmental modification, and tick genetic manipulation may be transferable to reduce tick attack on horses.

Studies of tick control with acaricides have included laboratory assays, area treatment by aerial and ground-operated methods, host-targeted bait stations, conventional topical application to hosts, and oral and injectable systemic acaricides. The following paragraphs summarize acaricidal tick control investigations since those reviewed in the Proceedings of a Workshop on Livestock Pest Management (Anon. 1979).

Barnard et al. (1981) compared various laboratory bioassay techniques and established baseline susceptibility data of *A. americanum* to some chlorinated hydrocarbons, several organophosphates, a carbamate, a pyrethroid, and a formamidine. Drummond (1986, 1987) screened experimental commercial compounds against engorged females of *B. annulatus, B. microplus, A. nitens*, and *D. albipictus*. Of 19 compounds screened, 6 were effective at 0.01% concentration. Drummond (1988) reviewed methods of controlling *A. maculatum* and screened 15 commercial acaricides against female *A. maculatum.* He analyzed and compared different susceptibilities among *A. maculatum, A. americanum, A. cajennense, A. nitens, D. albipictus, B. annulatus and B. microplus*.

Maupin and Piesman (1994) found permethrin and cyfluthrin more toxic to immature *I. scapularis* than were esfenvalerate or carbaryl, but all four acaricides were effective and required lower concentrations to kill larvae than to kill nymphs. Monsen et al. (1996) reported diazinon, chlorpyrifos, and carbaryl to be highly lethal to *I. pacificus* and *D. occidentalis*. The effects of desiccants and Safer's insecticidal soap on *I. scapularis* crawling over various substrates in a landscape were evaluated by Patrican and Allan (1995a). Teel et al. (1996) and Donahue et al. (1997) reviewed the literature on the effect of juvenile hormone analogs on ticks and reported on the effects of piriproxyfen on eggs, larvae, and nymphs of *A. americanum*. Susceptibility of immature ticks to various plant extracts has been investigated (Miller et al. 1995, Panella et al. 1997).

Area control of *A. americanum* nymphs and adults was obtained with sprays of permethrin, propoxur, diazinon, and naled, and with diazinon granules (Mount 1981a). In a separate trial against nymphal and adult *A. americanum*, air-blast spray treatment of plots with chlorpyrifos and stirofos both provided 88% control (Mount 1981b). Excellent control of *A. americanum* larvae was obtained by area sprays of chlorpyrifos, diazinon, and stirofos, and with 4% diazinon granules (Mount 1983).

In an area treatment, Schulze et al. (1991) obtained a useful degree of *I. dammini (=scapularis)* suppression using granular formulations of carbaryl, chlorpyrifos, and diazinon. Application timing and liquid and granular formulations of cyfluthrin were field tested for control of *A. americanum* and *I. dammini* (Solberg et al. 1992). They obtained excellent long term control of both species, especially with spring application of the liquid formulation. Aerial application of granular diazinon was employed successfully for control of *A. americanum* (Mount 1984). Granular carbaryl provided effective control of *I. scapularis* when aerially applied (Schulze et al. 1994). Schulze and Jordan (1995) used carbaryl granules to obtain 90% or greater reduction in numbers of *I. scapularis* in a New Jersey shrub and forest habitat and found that leaf litter depth did not influence the effect of the acaricide. Short-term control of *I. scapularis* has resulted from application of various desiccants and Safer's insecticidal soap in woodlands in New York State (Allan and Patrican 1995, Patrican and Allan 1995b). Monsen et al. (1996) obtained control of ticks in California with sprays of chlorpyrifos and carbaryl emulsions.

Sonenshine and Haines (1985) were the first to adapt for tick control the bait-box technique used for flea control on feral rodents. Using carbaryl dust in a bait of rodent nesting material, they obtained excellent control of *D. variabilis* the juveniles of which depend on rodent hosts. The method was adapted for control of *Ixodes scapularis* using permethrin-treated cotton balls as the bait and has been developed commercially as Damminix®. Damminix is EPA-registered under Special Local Needs in some states.

Although Damminix applications controlled immature *I. scapularis* in Massachusetts (Deblinger and Rimmer 1991), the product failed to reduce numbers of immature *I. scapularis* in three years of testing in Connecticut (Stafford 1991, 1992). Mejlon et al. (1995) reported that Damminix and other permtherin-treated nesting materials failed to control *I. ricinus* in Sweden. They attributed the failure to the fact that several rodent species serving as hosts of *I. ricinus* did not utilize the nesting material. Similar problems initially reduced the efficacy of permethrin-treated nesting material in bait tubes for controlling ticks associated with dusky-footed woodrats in California (Leprince and Lane 1996). Subsequent modification of the bait tubes resulted in excellent tick control (Lane et al. 1998).

A bait station, with ivermectin-treated feed, was developed for use by large, hoofed animals. Success with such devices has been obtained in treating elk to eliminate *Boophilus* spp. from a ranch in the quarantine zone in southern Texas (Cooke 1994) and in treating white-tailed deer to control *A. americanum* (Pound et al. 1996). A different bait station with a self-medicating applicator to deliver liquid acaricides was used to control *I. scapularis* on white-tailed deer and *A. americanum* on goats (Sonenshine et al. 1996).

Following are examples of continuing research on tick control by conventional sprays, dips, and pour-ons applied to animals. Sprays of seven then-registered acaricides (coumaphos, dioxathion, lindane, malathion, ronnel, stirofos, and toxaphene) were applied to cattle to control *A. americanum.* All gave significant control for 24 hours but none provided control for a week. Coumaphos and, possibly, lindane are the only products from this list that still may be used on cattle or horses. Both free-living and parasitic cohorts of an *A. americanum* population were suppressed by a series of spray treatments applied to cows and calves (Barnard et al. 1983). Both pour-on and spray applications of flumethrin yielded excellent control of *B. annulatus* on cattle (Ahrens et al. 1988).

Either whole-body spray or dipping of cattle with microencapsulated permethrin controlled *Boophilus microplus* (Ahrens et al. 1998). A 1% pour-on formulation of fipronil applied to cattle provided excellent control of *B. microplus* for six weeks (Davey et al. 1998).

Slow-release neckband and tailtag formulations of amitraz and cyhalothrin K were used on cattle for control of *A. americanum.* Both products and both kinds of application devices were more than 87% effective for 3 months (Miller and George 1994). The tailtag with cyhalothrin holds potential for tick control on horses.

Jaffe et al. (1986) explored the effects on ticks from insect steroid analogues released over a period of time via host implants. Drummond (1985) reviewed research on the efficacy of ivermectin via oral and subcutaneous administration to hosts, including sustained-release implants. High mortality of parasitizing ticks thus has been achieved in *A. americanum, A. cajennense, A. maculatum, B. microplus, D. albipictus, D. andersoni, D. variabilis,* and *R. sanguineus*. Ivermectin treatments of cattle (Meleney 1981) and horses (Craig and Kunde 1981) were not effective against the slow-feeding nymphal *O. megnini*. Miller et al. (1998) obtained promising long-term control of *A. americanum* on cattle injected with an experimental formulation of ivermectin in microspheres.

Wilkenson (1977) studied the effect of herbicidal elimination of shrubs on the abundance of *D. andersoni* in British Columbia and determined it had obvious advantages in removing residual populations. A similar study was conducted by Hoch et al. (1971) for the control of *A. americanum* in Oklahoma. Although labor intensive, mechanical removal of leaf litter from a forest floor resulted in 72% to 100% reduction in numbers of immature *I. scapularis* (Schulze et al. 1995). Sutherst (1971) examined the use of flooding pastures on tick survival and found only the eggs and larvae survived such a treatment. Additional studies along this line were conducted by Clymer et al. (1970) in Oklahoma and Wharton and Norris (1980) in Australia.

Nearly complete prevention of turkey parasitism by *A. americanum* occurred on previously burned plots (Jacobson and Hurst 1979). Significantly reduced A. *americanum* populations were obtained by annual and biennial early spring burning in Georgia (Davidson et al. 1994). In Massachusetts, vegetation destruction by either mowing or burning greatly reduced the abundance of questing adult *I. scapularis* for as much as one year post-treatment (Wilson 1986). Density of *A. americanum* populations was negatively correlated with frequency of grassland burning in Georgia (Davidson et al. 1994) and Kansas (Cully 1999).

Irradiation of *Hyalomma anatolicum* with Co60 was investigated by Srivastava and Sharma (1976). They found that 20,000-60,000 rads had an adverse effect on development with inhibition of complete egg laying at 60,000 r. However, these females lived about 10 weeks longer than those females not irradiated. Sternberg et al. (1973) found this technique enhanced the population of *Argas persicus*. Flooding the population with irradiated sterile females resulted in a 10-fold increase in the population and increased mating activity of males which produced more spermatophores than nonirradiated ticks. Similar studies were conducted by Purnell et al. (1973) on *R. appendiculatus*, on *O. tholozani* (Galun et al. 1967), and on *B. microplus* (Kitaoka and Morii 1967).

Potential biological control agents against ticks have been reported. Samsinakova et al. (1974) found an entomogenous fungus associated with the ticks *I. ricinus, D. marginatus* and *D. reticulatus*. Kaaya et al. (1996) measured not only mortality in adult *Rhipicephalus appendiculatus* and *Amblyomma variegatum*, but also reductions in engorgement weights, fecundity, and egg hatchability from the effects of *Beauveria bassiana* and *Metarhizium anisopliae*. Kaaya et al. found no harm to the fungi when incubated in organophosphate acaricides for up to 120 hours, thus suggesting compatability of the fungi with acaricides in an integrated tick control approach. *M. anisopliae* is also highly pathogenic to *I. scapularis* (Zhioua et al. 1997). High mortality of *B. microplus* from *M. anisopliae* was obtained in laboratory assays but, when applied to ticks on stabled cattle, the fungus was ineffective (do Carmo Barcelos Correia 1998). Martin and Schmidtmann (1998) identified 63 isolates of aerobic bacteria from *I. scapularis* and considered that as many as 40 of them may prove useful in biocontrol.

Cooley and Kohls (1945) summarized then-known information on several hymenopterous parasites of ticks, their known hosts, and distribution. Cole (1965) reviewed the use of hymenopterous parasites for tick control but the outlook for this strategy is not highly promising because ticks are apparently not the preferred host (Anon. 1979). However, research has continued on hymenopterous parasites as a possible adjunct to control of *I. scapularis* in the northeastern U.S. (Hu et al. 1993, Hu and Hyland 1998, Lyon et al. 1998) and of *D. variabilis* in eastern Oklahoma. Stafford et al. (1996) found the parasitoid wasp, *Ixodiphagus hookeri* (Howard) in natural populations of *Ixodes scapularis* in Connecticut, reported it to be dependent on high tick density, and concluded that the wasp had little potential in biocontrol of *I. scapularis*. A high percentage of ticks of three genera feeding on cattle became infected when exposed to entomopathogenic nematodes if exposed after six to nine days of feeding. Infection resulted in mortality ranging from 24% in *A. maculatum* to 96% in *D. variabilis* (Kocan et al. 1998). Ault and Elliott (1979) reviewed the literature on predation of *Ornithodoros* ticks.

The use of resistant breeds of cattle for the control of ticks has been extensively investigated (Anon. 1979, Wharton and Norris 1980). This strategy involves the use of African or Indian bulls in the breeding program. These breeds appear to have an innate resistance to ticks, especially *Boophilus* spp. Infestation density and biological fitness parameters of *A. americanum* on *Bos taurus, Bos indicus* and *Bos taurus* x *Bos indicus* crossbred cattle have been compared (Strother et al. 1975, Garris et al. 1979, Williams 1989). Williams' paper also covered *A. maculatum* on *Bos indicus* and *Bos taurus.* Other comparative studies have included African ticks on Nguni, Bonsmara, and Hereford cattle (Rechav et al. 1990). Brazilian workers compared *B. microplus* populations on Nellore, Ibage, and Nellore x European crossbreds (Gomes et. al 1989) and on Brahman and Simmentaler cattle (Rechav et. al 1990). Rechav (1987) studied differential serological factors involved in the comparative resistance of Brahman and Hereford cattle to African ticks. Differing susceptibilities of *Bos indicus, Bos taurus,* and their crosses to tick infestation and physiological effects on the cattle were demonstrated in Tanzania by Wambura (1998).

The promise of selective breeding for animal resistance to ectoparasites and disease has been documented (McDowell and Smith 1966, Drummond et. al 1988). Refined understanding of the physiology, immunology, and genetics of hosts as pertaining to parasites and disease should allow more rapid progress in breeding for resistance (van Dam 1981, Gavora and Spencer 1983). Steelman and co-workers (1993a) documented genetically-based differences in cattle to horn flies, not only between breeds but among individuals within breeds. They also reported significant interbreed differences in population densities of face flies on cattle (Steelman et al. 1993b). Thus, it seems that innate resistance of a given breed, whether to flies or ticks, might be further enhanced by within-breed selection.

Trager (1939a, 1939b) made important early observations on acquired host immunity to ticks. Behavioral, physiological, and immunological interactions between hosts and ectoparasites (including ticks) were reviewed and synthesized by Nelson et al. (1975, 1977). McGowan and Barker (1980) provided an important resource in publishing a selected bibliography on tick-host resistance. Wikel (1982) reviewed the literature on host immune responses to arthropods and their products.

In addition to *Bos indicus* having innate genetic resistance to ticks, they also acquire enhanced levels of resistance through infestation by ticks. Wikel and Whelen (1986) made comparative evaluations of various tick antigens involved in stimulating host immunosuppression (George et al. 1985). Tatchell (1987) summarized the nature of natural interaction between ticks and their hosts to gain insights that might be applicable to managing ticks on domestic livestock. Contemporary research on host immune response to ticks was reviewed by Brown (1986). Tick-host interactions are placed in perspective with those of other ectoparasites and their hosts by Brown (1989) and Nelson (1989).

One should not overlook such complicating factors in tick-induced immune response as inducement of cross-resistance to other ectoparasites (de Castro et al. 1989, Rechav et al. 1989), which may prove beneficial. Nor should we overlook the fact that while we seek ways to recognize, enhance, and utilize host resistance to ticks, the ticks may be evolving to counteract the challenge (Chiera et al. 1989). And, although we tend to think of immune response to ticks as a beneficial phenomenon that we may capitalize on, it has been shown that tick antigens injected into the host during feeding cause immunomodulation that facilitates both engorgement and pathogen transmission (Ramachandra and Wikel 1992).

Bolstered by modern cytological and molecular techniques, 50 years of research in acquired immunity to ticks has, during the last decade, yielded practical artificial immunization procedures. Some of the mileposts in this development are traced, more or less chronologically, as follows: Brown (1986) isolated a tick salivary gland protein, from *A. americanum,* capable of inducing immune resistance in laboratory rodents. Extracts from adult female *B. microplus* were used to immunize cattle against tick parasitism (Agbede and Kemp 1986, Johnston et al. 1986, Kemp et al. 1986). Wong and Opdebeeck (1989) reported on the protective efficacy of antigens from gut membranes of *B.*

microplus, and Wong et al.(1990) studied the bovine immune response of Hereford cattle vaccinated with *B. microplus* antigens. Oviposition by *O. moubata* was reduced after feeding on vitellin-immunized rabbits (Chinzei and Minoura 1988).

Cattle have been immunized against *Hyalomma anatolicum anatolicum* using salivary gland extract (Banerjee et al. 1990), and guinea pigs were immunized for protection against adult *R. appendiculatus* using homogenates from immature ticks from the same species (Varma et al. 1990). Host and tick response to artificial immunization against *A. hebraeum* and *A. marmoreum* were studied by Tembo and Rechav (1992) and Dharampaul et al. (1993). Field use and cost-effectiveness of vaccination against *B. microplus* were evaluated by de la Fuente et al. (1998).

Willadsen (1987) reviewed immunological approaches to tick control and compared their practical potentials. Immunological strategies for suppression of ticks and other ectoparasites were reviewed by Wikel et al. (1992). Yong (1992) edited a major reference work on the use of biotechnology to control both endoparasites and ectoparasites.

Labarta et al (1996) developed a simulation model to evaluate the impact of vaccination regimes on tick control and to help integrate vaccination programs with other control methods. And Brossard (1998) reviewed the use of vaccines and genetically resistant animals in tick control.

Barnard (1989) found behavioral differences in Zebu vs. British and British-Zebu crossbred cattle with regard to relative time spent in tick-infested habitat where a choice was available. Perhaps this is complementary to the Zebus' innate resistance and might be another trait to consider in selective breeding.

Elimination or reduction of deer was used to reduce *I. scapularis* abundance on an island (Wilson et al. 1984). Success was limited by several factors even on an island where deer repopulation was not a problem. Deer exclusion by fencing provided significant reduction of *I. scapularis* populations in Connecticut and New York (Stafford 1993, Daniels et al. 1993). Deer exclusion in Kentucky resulted in 98 percent fewer *A. americanum* larvae, but the impact on nymphs and adults was far less and adult *D. variabilis* became even more abundant (Bloemer et al. 1986). Interactions between deer exclusion fencing, small and medium-sized mammalian hosts of ticks, and tick populations were studied by Daniels and Fish (1995).

Integrating pest management strategies for tick suppression is complicated by the presence of alternative hosts, by the paucity of biocontrol options, by the scale on which interventions must operate, and by the limited chemical options available. Challenges in integrating tick control measures have been addressed in South Africa (Spickett 1987), Paraguay (Brizuela et al. 1996), and elsewhere. Mathematical models have been developed to help understand and evaluate components of integrated tick management strategies, e.g., of *Ixodes scapularis* (Mount et al. 1997) and *Boophilus microplus* (Beugnet et al. 1998).

As this review indicates, few of the insecticides investigated for on-animal tick control have been registered or tested for use on horses. On-animal equine products are limited primarily to pyrethrins (and synergists) and pyrethroids. Biological control would be compatible with direct application of acaricides to hosts but probably not with area treatment of habitat with acaricides.

Summary of Research and Extension Needs

1) The host-parasite relationships of pest flies and ectoparasites of horses need to be investigated. In particular, the populations of these arthropods should be determined in various regions, including seasonality, population fluctuations, weather effects, and premise environmental conditions.

2) Evaluate disease transmission potential to horses by vector species and by regions.

3) Sampling methods should be developed for each of these groups. Samples should be correlated with absolute populations on the animal.

4) Action thresholds, based on both economic considerations and aesthetic concerns, need to be developed.

5) Novel insecticides and biological control agents should be developed, along with innovative delivery systems for their application. Strategic placement should reduce environmental contamination and minimize human and animal exposure while targeting susceptible life stages.

6) Develop pest management strategies that are socially, environmentally and economically acceptable.

7) Natural and induced host resistance should be investigated.

8) The use of genetic manipulations should be investigated for ectoparasites and fly pests of horses.

9) Potential for eradication of each pest should be assessed.

10) Research needs to evaluate the correlation between area-wide pest populations and numbers of the pest found on animals.

11) Information on the biology, ecology and physiology of ectoparasites needs to be worked out for various regions of the country. Information needed includes seasonal locations on the animals, climate induced mortality factors, and the effect of host physiology, breed and diet on populations and on economic injury levels.

12) Host specificity of each group should be determined, along with the suitability of alternative hosts.

13) Species population dynamics in relationship to emergence, dispersal and host seeking by regions should be investigated.

14) Several of these pest species are shared with cattle, sheep, other livestock and wildlife. Coordinated efforts should be established with researchers on these other groups and truly integrated pest management strategies devised.

15) Because many of these pests are dealt with by veterinarians, collaborative efforts of entomologists and members of the veterinary profession should be undertaken.

REFERENCES CITED

Adkins, T. R. Jr., W. L. Sowell & F. S. Arant. 1955. Systemic effect of selected chemicals on the bed bug and lone star tick when administered to rabbits. J. Econ. Entomol. 48: 139-141.

Agbede, R.I.S. & D. H. Kemp. 1986. Immunization of cattle against *Boophilus microplus* using extracts derived from adult female ticks: Histopathology of ticks feeding on vaccinated cattle. Int. J. Parasitol. 16: 35-41.

Ahrens, E. H., R. B. Davey, J. E. George & L. M. Cooksey. 1988. Flumethrin applied as a pour-on and whole-body spray for controlling cattle tick (Acari: Ixodidae) on cattle. J. Econ. Entomol. 81: 1133-1136.

Ahrens, E.H., R.B. Davey, J.E. George and J.S. Hunter III. 1998. Microencapsulated permethrin for controlling *Boophilus microplus* on cattle in South Texas. Southwest. Entomol. 23(1): 25-30.

Allan, S.A. and L.A. Patrican. 1995. Reduction of immature *Ixodes scapularis* (Acari: Ixodidae) in woodlots by application of desiccant and insecticidal soap formulations. J. Med. Entomol. 32(1): 16-20

American Horse Council (AHC). 1979. AHC horse industry directory. AHC publications. WA.

American Horse Council. 1994. The Economic Impact of the United States Horse Industry. AHC publications. WA. 23pp.

American Horse Council. 1996. Economic Impact Study. AHC Publications. Washington D.C.

Anderson, B. E., J. E. Dawson, D. C. Jones & K. H. Wilson. 1991. *Ehrlichia chaffeensis,* a new species associated with human ehrlichiosis. J. Clin. Microbiol. 29: 2838-2842.

Anderson, B.E., K.G. Sims, J.G. Olson, J.E. Childs, J.F. Piesman, C.M. Happ, G.O. Maupin and B.J.B. Johnson. 1993. *Amblyomma americanum*: A potential vector of human ehrlichiosis. Amer. J. Trop. Med. Hyg. 49: 239-244.

Anonymous. 1979. Proceedings of a Workshop on Livestock Pest Management: To Assess National Research and Extension Needs for Integrated Pest Management of Insects, Ticks, and Mites Affecting Livestock and Poultry. March 5-7, Manhattan, Kansas.

Ault, S. K. and K. D. Elliott. 1979. Spider predation (Araneae: Salticidae) on *Ornithodoros coriaceus* (Acarina: Argasidae), with a survey of the predators of the genus *Ornithodoros*. J. Med. Entomol. 15: 570-571.

Baker, D. W. & W. S. Monlux. 1939. *Hypoderma* myiasis in the horse--summary of a series of cases studied during spring and summer, 1939. J. Parasitol. (Suppl.) 25: 16.

Banerjee, D. P., R. R. Momin & S. Samantaray. 1990. Immunization of cattle (*Bos indicus* x *Bos taurus*) against *Hyalomma anatolicum anatolicum* using antigens derived from tick salivary gland extracts. Int. J. Parasitol. 20: 969-972.

Barbour, A.G., G.O. Maupin, G.L. Teltow, C.J. Carter and J. Piesman. 1996. Identification of an uncultivable *Borrelia* species in the hard tick *Amblyomma americanum*: Possible agent of a Lyme disease-like illness. J. Infec. Dis. 173: 403-409.

Barnard, D. R. 1989. Habitat use by cattle affects host contact with lone star ticks (Acari: Ixodidae). J. Econ. Entomol. 82: 854-859.

Barnard, D. R. & B. G. Jones. 1981. Field efficacy of acaricides for control of the lone star tick on cattle in southeastern Oklahoma. J. Econ. Entomol. 74: 558-560.

Barnard, D. R., B. G. Jones, G. D. Rogers & G. A. Mount. 1981. Acaricide susceptibility in the lone star tick: assay techniques and baseline data. J. Econ. Entomol. 74: 466-469.

Barnard, D. R., G. A. Mount, D. G. Haile & E. Daniels. 1994. Integrated management strategies for *Amblyomma americanum* (Acari: Ixodidae) on pastured beef cattle. J. Med. Entomol. 31: 571-585.

Barnard, D. R., G. D. Rogers & B. G. Jones. 1983. Strategic treatment of pastured beef cattle with acaricides: effect on populations of the lone star tick (Acari: Ixodidae). J. Econ. Entomol. 76: 99-102.

Barton, L. L. & T. M. Foy. 1989. Erlichia canis infection in a child. Pediatrics 4(3): 580 582.

Bauer, C. & J. J. Burger. 1986. Efficacy of an oxibendazole-trichlorfon paste formulation against third stage larvae of *Gasterophilus intestinalis* and its safety in horses. Vet. Rec. 19: 294-296.

Baumhover, A. H. 1966. Eradication of the screwworm fly, an agent of myiasis. J. Amer. Med. Assoc. 196: 240-248.

Belding, D. L. 1942. Textbook of Clinical Parasitology. D. Appleton-Century Company, New York.

Bello, T. R. 1989. Efficacy of ivermectin against experimental and natural infections of *Gasterophilus* spp. in ponies. Am. J. Vet. Res. 50: 2120-2123.

Beugnet, F., K. Chalvet-Monfray and P. Sabatier. 1998. Use of a mathematical model to study the control measures of the cattle tick *Boophilus microplus* population in New Caledonia. Vet. Parasitol. 77: 277-288

Bishopp, F. C. 1926. Screw worms and other maggots affecting animals. Farmers' Bull. #857.

Bishopp, F. C. 1933. Mosquitoes kill livestock. Science 77: 115-116.

Blagburn, B. L., D. S. Lindsey, C. M. Hendrix & J. Schumacher. 1991. Pathogenesis, treatment, and control of gastric parasites in horses. Comp. Cont. Educ. Prac. Vet. 13: 850-853, 856-857.

Blanton, F. S. & W. W. Wirth. 1979. The Sand Flies (*Culicoides*) of Florida (Diptera: Ceratopogonidae). Florida Dept. of Agric. and Consumer Services, Contribution No. 424, Bureau of Entomology.

Bloemer, S. R., E. L. Snoddy, J. C. Cooney & K. Fairbanks. 1986. Influence of deer exclusion on populations of lone star ticks and American dog ticks (Acari: Ixodidae). J. Econ. Entomol. 79: 679-683.

Bolte, J. R. 1988. Control of external parasites of beef cattle, pp. 169-194 (ch. 7) *In* Oklahoma Beef Cattle Manual (2nd ed.) Coop. Ext. Serv., Okla. State Univ., Stillwater, Okla.

Bolte, J. & S. Coppock. 1988. Horse external parasite control series I. 1. Introduction. 2. Fly pests. OSU Ext. Stillwater, OK: The Service 7015. 6pp.

Bolte, J. & S. Coppock. 1988. Horse external parasite control series II. 1. Lice, ticks, mites and their control. 2. Diseases transmitted by external parasites. OSU Ext. Stillwater, OK: The Service 7016. 4pp.

Bowne, J. G., A. J. Luedke, N. M. Foster & M. M. Jochim. 1966. Current status of bluetongue in cattle. J. Amer. Vet. Med. Assoc. 148: 1177-1180.

Brizuela, C.M., C.A. Ortellado, T.I. Sanchez, O. Osorio and A.R. Walker. 1996. Formulation of integrated control of *Boophilus microplus* in Paraguay: analysis of natural infestations. Vet. Parasitol. 63: 95-108.

Broce, A. B. 1985. Myiasis-producing flies. Pp. 83-100. In: Williams, R. E., R. D. Hall, A. B. Broce & P. J. Scholl (Eds.), Livestock Entomology. John Wiley & Sons, New York, NY., 335pp.

Broce, A. B. 1993. Dispersal of house flies and stable flies. In: Thomas, G. D. & S. R. Skoda (eds.), Rural Flies in the Urban Environment? North Central Regional Research Pub. No. 335, University of Nebraska Agricultural Research Division, Institute of Agriculture and Natural Resources Research Bulletin No. 317, Lincoln, NE.

Brossard, M. 1998. The use of vaccines and genetically resistant animals in tick control. Rev. Sci. Tech. 17: 188-199.

Brown, S. J. 1986. *Amblyomma americanum*: Physiochemical isolation of a protein derived from the tick salivary gland that is capable of inducing immune resistance in guinea pigs. Exp. Parasitol. 62: 40-50.

Brown, S. J. 1988. Highlights of contemporary research on host immune responses to ticks. Vet. Parasitol. 28: 321-324.

Brown, S. J. 1989. Pathological consequences of feeding by hematophagous arthropods: comparison of feeding strategies. pp. 4-14 *In* Proc. Symp.: Physiological Interactions Between Hematophagous Arthropods and Their Vertebrate Hosts. edited by C.J. Jones and R.E. Williams. Entomol. Soc. Am. Misc. Publ. 31.

Burgdorfer, W. 1975. A review of Rocky Mountain spotted fever (tick-borne typhus), its agent, and its tick vectors in the United States. J. Med. Entomol. 12: 269-278.

Burgess, E. C. 1988. Borrelia burgdorferi infection in Wisconsin horses and cows. Ann. N.Y. Acad. Sci. 539: 235-243.

Burgess, E. C., D. Gillette & J. P. Pickett. 1986. Arthritis and panuveitis as manifestations of *Borrelia burgdorferi* infection on a Wisconsin pony. J. Am. Vet. Assoc. 189(10): 1340-1342.

Bushland, R. C. 1974. Letter: Screwworm eradication program. Science 184(140): 1010-1011.

Carpenter, T. L., M. C. McMeans & C. P. McHugh. 1990. Additional instances of human parasitism by the brown dog tick (Acari:Ixodidae). J. Med. Entomol. 27(6): 1065-1066.

Carroll, J. F. & E. T. Schmidtmann. 1986. American dog tick (Acari: Ixodidae), summer activity on equine premises enzootic for Potomac horse fever in south central Maryland. J. Econ. Entomol. 79(1): 62-66.

Carroll, J. F. & E. T. Schmidtmann. 1992. Tick sweep: modification of the tick drag flag method for sampling nymphs of the deer tick (Acari: Ixodidae). J. Med. Entomol. 29: 352-355.

Catts, E. P. 1979. Hilltop aggregation and mating behavior by *Gasterophilus intestinalis* (Diptera: Gasterophilidae). J. Med. Entomol. 16: 461-464.

Chiera, J. W., R. M. Newsom & G. R. Karuhize. 1989. Adaptation of field strains of *Rhipicephalus appendiculatus* Neumann (Acarina: Ixodidae) to host resistance. Parasitology P1: 149-155.

Chinzei, Y. & H. Minoura. 1988. Reduced oviposition in *Ornithodoros moubata* (Acari: Argasidae) fed on tick-sensitized and vitellin-immunized rabbits. J. Med. Entomol. 25: 26-31.

Clymer, B. C., D. E. Howell & J. A. Hair. 1970. Animal hosts of economically important ticks (Acarina) in east-central Oklahoma. Entomol. Soc. Amer. Ann. 63(2): 612 614.

Cogley, T. P. 1989. Effects of migrating *Gasterophilus intestinalis* larvae (Diptera: Gasterophilidae) on the mouth of the horse. Vet. Parasitol. 31: 317-331.

Cogley, T. P., J. R. Anderson & L. J. Cogley. 1982. Migration of *Gasterophilus intestinalis* larvae (Diptera: Gasterophilidae) in the equine oral cavity. Int. J. Parasitol. 12: 473-480.

Cole, M. M. 1965. Biological control of ticks by the use of hymenopterous insects. A review. World Health Organization (WHO/EBL/43.66) 43: 1-12.

Conrad, M. E. 1989. Erlichia canis: a tick-borne rickettsial-like infection in humans living in the southeastern United States. Am. J. Med. Sci. 297(1): 35-37.

Cooke, L. 1994. Ticked off! Agric. Res. 42(4): 4-6.

Cooley, R. A. & G. M. Kohls. 1945. A summary on tick parasites. Proc. Fifth Pacific Science Congress. B5(6): 3375-3381.

Cooley, R. A. & G. M. Kohls. 1945. The genus Ixodes in North America. Nat. Inst. Health Bull. 184: 246 pp.

Craig, T. M. & J. M. Kunde. 1981. Controlled evaluation of ivermectin in Shetland ponies. Am. J. Vet. Res. 42: 1422-1424.

Cully, J. F., Jr. 1999. Lone star tick abundance, fire, and bison grazing in tallgrass prairie. J. Range Manage. 52(2): 139-144.

Daniels, T.J. and D. Fish. 1995. Effect of deer exclusion on the abundance of immature *Ixodes scapularis* (Acari: Ixodidae) parasitizing small and medium sized mammals. J. Med. Entomol. 32(1): 5-11.

Daniels, T. J., D. Fish & I. Schwartz. 1993. Reduced abundance of *Ixodes scapularis* (Acari: Ixodidae) and Lyme disease risk by deer exclusion. J. Med. Entomol. 30: 1043-1049.

Dart, A. J., D. R. Hutchins & A. P. Begg. 1987. Suppurative splenitis and peritonitis in a horse after gastric ulceration caused by larvae of *Gasterophilus intestinalis*. Aust. Vet. J. 64: 155-158.

Davidson, W. R., D. A. Siefken & L. H. Creekmore. 1994. Influence of annual and biennial prescribed burning during March on the abundance of *Amblyomma americanum* (Acari: Ixodidae) in central Georgia. J. Med. Entomol. 31: 72-81.

Davey, R.B., E.H. Ahrens, J.E. George, J.S. Hunter III and P. Jeannin. 1998. Therapeutic and persistent efficacy of fipronil against *Boophilus microplus* (Acari: Ixodidae) on cattle. Vet. Parasitol. 74: 261-276.

Davidson, W. R., D. A. Siefken and L. H. Creekmore. 1994. Influence of annual and biennial prescribed burning during March on the abundance of *Amblyomma americanum* (Acari: Ixodidae) in central Georgia. J. Med. Entomol. 31: 72-81.

Deblinger, R. D. & D. W. Rimmer. 1991. Efficacy of a permethrin-based acaricide to reduce the abundance of *Ixodes dammini* (Acari: Ixodidae). J. Med. Entomol. 28: 708-711.

de Castro, J. J., R. M. Newson & I. V. Herbert. 1989. Resistance in cattle against *Rhipicephalus appendiculatus* with an assessment of cross-resistance to *R. pulchellus* (Acari: Ixodidae). Exp. Appl. Acarol. 6: 237-244.

de la Fuente, J., M. Rodriguez, M. Redondo, C. Montero, J.C. Garcia-Garcia, L. Mendez, E. Serrano, M. Valdez, A. Enriquez, M. Canales, E. Ramos, O. Boue, H. Machado, R. Lleonart, C.A. de Armas, S. Rey, J.L. Rodriguez, M. Artiles and L. Garcia. 1998. Field studies and cost-effectiveness analysis of vaccination with Gavac against the cattle tick *Boophilus microplus*. Vaccine 16: 366-373.

DeVaney, J. A & J. J. Garcia. 1975. Longevity, oviposition, and fertility of several strains of the screwworm, *Cochliomyia hominivorax* (Diptera: Calliphoridae). J. Med. Entomol. 12(5): 511-513.

Dharampaul, S., W. R. Kaufman & M. Belosevic. 1993. Differential recognition of saliva antigens from the ixodid tick *Amblyomma hebraeum* (Acari: Ixodidae) by sera from infested and immunized rabbits. J. Med. Entomol. 30: 262-266.

Dimmitt, D. C., D. B. Fishbein & J. E. Dawson. 1989. Human ehrlichiosis associated with cerebrospinal fluid pleocytosis--a case report. Am. J. Med. 87: 677-678.

do Carmo Barcelos Correia, A., A.C. Fiorin, A.C. Monteiro and C.J. Verissimo. 1998. Effects of *Metarhizium anisopliae* on the tick *Boophilus microplus* (Acari: Ixodidae) in stabled cattle. J. Invert. Pathol. 71: 189-191.

Donahue, W.A., P.D. Teel, O.F. Strey and R.W. Meola. 1997. Pyriproxyfen effects on newly engorged larvae and nymphs of the lone star tick (Acari: Ixodidae). J. Med. Entomol. 34(2): 206-211.

Dove, W. E. 1937. Myiasis of man. Symposium: Insects affecting man. J. Econ. Entomol. 30(1): 29-39.

Drudge, J. H., E. T. Lyons, Z. N. Wyant & S. C. Tolliver. 1975. Occurrence of second and third instars of *Gasterophilus intestinalis* and *Gasterophilus nasalis* in stomachs of horses in Kentucky. Am. J. of Vet. Res. 36: 1585-1588.

Drummond, R. O. 1963. Tests with systemic insecticides for the control of *Gasterophilus* larvae in horses. J. Econ. Entomol. 56: 50-52.

Drummond, R. O. 1985. Effectiveness of ivermectin for control of arthropod pests of livestock. Southwest. Entomol. (Suppl.) 7: 34-42.

Drummond, R. O. 1986. Screening of acaricides for control of one-host ticks, 1985. Insecticide & Acaricide Tests 11: 455-456.

Drummond, R. O. 1987. Screening of acaricides for control of one-host ticks, 1986. Insecticide & Acaricide Tests 12: 362.

Drummond, R. O. 1988. Susceptibility of the Gulf Coast tick (Acari: Ixodidae) to acaricides. J. Econ. Entomol. 81: 1140-1142.

Drummond, R. O. & J. M. Ossorio. 1966. Additional tests with insecticides for the control of the tropical horse tick on horses in Florida. J. Econ. Entomol. 59: 107 110.

Drummond, R. O., S. E. Ernst, J. L. Trevino, W. J. Gladney & O. H. Graham. 1973. *Boophilus annulatus* and *B. microplus*: laboratory tests of insecticides. J. Econ. Entomol. 66(1): 130-133.

Drummond, R. O., J. E. George & S. E. Kunz. 1988. Control of arthropod pests of livestock: a review of technology. CRC Press, Inc., Boca Raton, Fla.

Egri, B., P. Sarkozy & G. Banhidy. 1995. Prevalence of botfly larvae and lice in studs of North Caucasus (Stawropol County, Russia). Acta Veterinaria Hungarica 43(2-3): 287-289.

Falco, R. C. & D. Fish. 1991. Horizontal movement of adult *Ixodes dammini* (Acari: Ixodidae) attracted to C02-baited traps. J. Med. Entomol. 28: 726-729.

Fehrenbach, P. 1992. Studying Lyme disease in Carolina. Pest Cont. 60: 46-48.

Foil, L. & C. Foil. 1988. Dipteran parasites of horses. Equine Pract. 10: 21-38.

Foil, L. & C. Foil. 1990. Arthropod pests of horses. Comp. Cont. Educ. Pract. Vet. 12: 723-731.

Frahm, J. 1983. Gasterophilosis (horse bot infestation)--biology, diagnosis and therapy. Vet. Med. Rev. 1483: 204-217.

Fryxell, K. J. & T. A. Miller. 1995. Autocidal biological control: a general strategy for insect control based on genetic transformation with a highly conserved gene. J. Econ. Entomol. 88: 1221-1232.

Galloway, J. H. 1972. Farm Animal Health and Disease Control. Philadelphia: Leak Febiger. 373 pp.

Galun, R., M. Warburg & A. Avivi. 1967. Studies on the application of the sterility method in the tick, *Ornithodoros tholozani*. Entomol. Exp. Appl. 10(2): 143-152.

Garris, G. I., B. R. Stacey, J. A. Hair & R. W. McNew. 1979. A comparison of lone star ticks on Brahman and Hereford cattle. J. Econ. Entomol. 72: 869-872.

Gavora, J. S. & J. L. Spencer. 1983. Breeding for immune responsiveness and disease resistance. Anim. Blood Groups Biochem. Genetics 14: 159-180.

George, J. E., R. L. Osburn and S. K. Wikel. 1985. Acquisition and expression of resistance by *Bos indicus* and *Bos indicus - Bos taurus* calves to *Amblyomma americanum* infestation. J. Parasitol. 71: 174-182.

George, J., A. Miller & M. Pound. 1991. Lyme disease suppression utilizing acaricides and systemics for tick control. pp. 13-14 *In* Proc. Southeast. Lyme Dis. Wkshp. Knoxville, TN, July 29-31. Tenn. Val. Author., National Park Serv., and Tenn. Dept. Health.

George, J. E., R. L. Osburn & S. K. Wikel. 1985. Acquisition and expression of resistance by *Bos indicus* and *Bos indicus - Bos taurus* calves to *Amblyomma americanum* infestation. J. Parasitol. 71: 174-182.

Goddard, J. 1989. Focus of human parasitism by the brown dog tick, *Rhipicephalus sanguineus* (Acari: Ixodidae). J. Med. Entomol. 26(6): 628-629.

Goddard, J. 1993. Physician's Guide to Arthropods of Medical Importance. CRC Press; Boca Raton, Fla. 332 pp.

Goldfarb, L. G. 1986. Epidemiological models of tick-borne infections (Acari: Ixodidae and Argasidae). J. Med. Entomol. 23: 125-131.

Gomes, A., M. R. Honer, M.A.M. Schenk & J.B.E. Curvo. 1989. Populations of the cattle tick (*Boophilus microplus*) on purebred Nellore, Ibage and Nellore x European crossbreeds in the Brazilian Savanna. Trop. Anim. Health Production 21(1): 20 24.

Gothe, R., K. Kunze & H. Hoogstraal. 1979. The mechanisms of pathogenicity in the tick paralyses. J. Med. Entomol. 16: 357-369.

Graham, O. H. & J. L. Hourrigan. 1977. Eradication programs for the arthropod parasites of livestock. J. Med. Entomol. 13(6): 629-658.

Greenberg, B. 1971. Flies and Disease. Vol. 1. Ecology, classification and biotic associations. Princeton University Press. Princeton, NJ. 856pp.

Greer, N. I. & J. F. Butler. 1973. Comparisons of horn fly development in manure of five animal species. Fla. Entomol. 56: 179-199.

Hadlow, W. J., J. K. Ward & W. L. Krinsky. 1977. Intracranial myiasis by *Hypoderma bovis* (Linnaeus) in a horse. Corn. Vet. 67: 272-281.

Hall, M. C. 1917. Notes in regard to horse lice, *Trichodectes* and *Haematopinus*. J. Am. Vet. Med. Assoc. 51: 494.

Hall, R. D., K. A. Hewadikaram, G. A. McDonald & K. E. Doisy. 1992. Research on ticks and tickborne pathogens in Missouri--an interim report. Missouri Med. 89: 339-342.

Harris, R. L., J. A. Miller & E. D. Frazar. 1974. Horn flies and stable flies: Feeding activity. Ann. Entomol. Soc. Am. 67: 891-894.

Harwood, R. F. & M. T. James. 1979. Entomology in Human and Animal Health. Macmillan, New York.

Herms, W. B. 1911. The housefly in its relation to the public health. Univ. Calif. Agric. Exper. Sta. Bull. 215, pp. 513-548.

Hoch, A. L., R. W. Baker & J. A. Hair. 1971. Further observations on the control of lone star ticks (Acarina: Ixodidae) through integrated control procedures. J. Med. Entomol. 8(6): 731-734.

Hogsette, J. A. & J. P. Ruff. 1985. Stable fly (Diptera: Muscidae) migration in Northwest Florida. Environ. Entomol. 14: 170-175.

Hooker, W. A., F. C. Bishopp & H. P. Wood. 1912. The life history and bionomics of some North American ticks. U.S. Dept. Agric. Bur. Entomol. Bull. 106: 239.

Hu, R., K. E. Hyland & T. N. Mather. 1993. Occurrence and distribution in Rhode Island of *Hunterellus hookeri* (Hymenoptera: Encyrtidae), a wasp parasitoid of *Ixodes dammini*. J. Med. Entomol. 30(1): 277-280.

Hu, R. and K.E. Hyland. 1998. Effects of the feeding process of *Ixodes scapularis* (Acari: Ixodidae) on embryonic development of its parasitoid, *Ixodiphagus hookeri* (Hymenoptera: Encyrtidae). J. Med. Entomol. 35(6): 1050-1053.

Imes, M. 1926. Lice, manage, and ticks of horses, and methods of control and eradication. U.S. Dept. Agric. Farmers' Bull. 1493.

Jacobson, H. A. & G. A. Hurst. 1979. Prevalence of parasitism by *Amblyomma americanum* on wild turkey poults as influenced by prescribed burning. J. Wildl. Dis. 15: 43-47.

Jaffe, H., D. E. Sonenshine, D. K. Hayes, W. H. Dees, M. Beveridge & M. J. Thompson. 1986. Controlled-release reservoir systems for the delivery of insect steroid analogues against ticks (Acari: Ixodidae). J. Med. Entomol. 23: 685-691.

James, M. T. 1948. The flies that cause myiasis in man. U. S. Dept. Agric. Publ. 631. 175 pp.

Johnston, L.A.Y., D. H. Kemp and R. D. Pearson. 1986. Immunization of cattle against *Boophilus microplus* using extracts derived from adult female ticks: effects of induced immunity on tick populations. Intl. J. Parasit. 16: 27-34.

Jones, R. H., H. W. Potter, Jr. & H. A. Rhodes. 1972. Ceratopogonidae attacking horses in south Texas during the 1971 VEE epidemic. Mosq. News 32: 507-509.

Kaaya, G. P., E. N. Mwangi and E. A. Ouna. 1996. Prospects for biological control of livestock ticks, *Rhipicephalus appendiculatus* and *Amblyomma variegatum*, using entomogenous fungi *Beauveria bassiana* and *Metarhizium anisopliae*. J. Invert. Pathol. 67: 15-20.

Keirans, J. E., H. J. Hutcheson, L. A. Durden and J.S.H. Klompen. 1996. *Ixodes* (*Ixodes*) *scapularis* (Acari: Ixodidae): Redescription of all active stages, distribution, hosts, geographical variation, and medical and veterinary importance. J. Med. Entomol. 33(3): 297-318

Kemp, D. H., R.I.S. Agbede, L.A.Y. Johnston and J. M. Gough. 1986. Immunization of cattle against *Boophilus microplus* using extracts derived from adult female ticks: feeding and survival of the parasite on vaccinated cattle. Int. J. Parasitol. 16: 115 20.

Kissling, R. E. & H. Rubin. 1950. Pathology of eastern equine encephalomyelitis. Am. J. Vet. Res. 12(43): 100-105.

Kitaoka, S. & T. Morii. 1967. Effects of gamma radiation and chemosterilants on the cattle tick, Boophilus microplus. Jap. J. Sanit. Zool. 16(3): 244-248.

Knapp, F. W. 1985. Arthropod Pests of Horses. Pp. 297-313. In: Williams, R. E., R. D. Hall, A. B. Broce & P. J. Scholl (eds.). Livestock Entomology. John Wiley, New York. 335 pp.

Kocan, K. M., M. S. Pidherney, E. F. Blouin, P. L. Claypool, M. Samish and I. Glazer. 1998. Interaction of entomopathogenic nematodes (*Steinernematidae*) with selected species of ixodid ticks (Acari: Ixodidae). J. Med. Entomol. 35(4): 514 520.

Krinsky, W. L. 1976. Review article -- animal disease agents transmitted by horse flies and deer flies (Diptera: Tabanidae). J. Med. Entomol. 13: 225-275.

Laake, E. W. 1936. Economic studies of screwworm flies, *Cochliomyia* species (Diptera, Calliphorinae), with special reference to the prevention of myiasis of domestic animals. Iowa State Coll. J. Sci. 10: 345-359.

Labarta, V., M. Rodriguez, M. Penichet, R. Lleonart, L. L. Luaces and J. de la Fuente. 1996. Simulation of control strategies for the cattle tick *Boophilus microplus* employing vaccination with a recombinant Bm86 antigen preparation. Vet. Parasitol. 63: 131-160.

Lane, R. S., L. E. Casher, C. A. Peavey and J. Piesman. 1998. Modified bait tube controls disease-carrying ticks and fleas. Calif. Agric. 52(2): 43-48.

Leprince, D. J. and R. S. Lane. 1996. Evaluation of permethrin-impregnated cotton balls as potential nesting material to control ectoparasites of woodrats in California. J. Med. Entomol. 33(3): 355-360

Linley, J. R. 1985. Biting midges (Diptera: Ceratopogonidae) as vectors of nonviral animal pathogens. J. Med. Entomol. 22(6): 589-599.

Lockhart, J. M., W. R. Davidson, D. E. Stallknecht and J. E. Dawson. 1996. Site specific geographic association between *Amblyomma americanum* (Acari: Ixodidae) infestations and *Ehrlichia chaffeensis*-reactive (Rickettsiales: Ehrlichieae) antibodies in white-tailed deer. J. Med. Entomol. 33(1): 153-158.

Loomis, E. C., A. S. Deal, W. C. Fairbank & G. G. Gurtle. 1975. Fly (Diptera) control on the horse ranch. Univ. Calif. Berkeley Ext. Service #2335. 12pp.

Luckhart, S., G. R. Mullen & J. C. Wright. 1991. Etiologic agent of Lyme disease, Borrelia burgdorferi, detected in ticks (Acari: Ixodidae) collected at a focus in Alabama. J. Med. Entomol. 28(5): 652-657.

Luedke, A. J., R. H. Jones & M. M. Jochim. 1967. Transmission of bluetongue between cattle and sheep by Culicoides variipennis. Am. J. Vet. Res. 28(123): 457-460.

Lyon, S. M., R. Van Driesche and J. D. Edman. 1998. Ecology of *Hunterellus hookeri* (Hymenoptera: Encyrtidae) and evaluation of its impact on *Ixodes scapularis* (Acari: Ixodidae) on Nonamesset Island in Massachusetts. Environ. Entomol. 27(2): 463-468.

Martin, P. A. and E. T. Schmidtmann. 1998. Isolation of aerobic microbes from *Ixodes scapularis* (Acari: Ixodidae), the vector of Lyme disease in the eastern United States. J. Econ. Entomol. 91(4): 864-868.

Masters, E. 1990. Case histories in Missouri. In Lyme Disease in the south central United States (Symp.). Missouri Acad. Sci. Occas. Paper #8, St. Louis Univ. pp. 5-8.

Matthysse, J. G., G. van Vreden, A. Purnasiri, C. J. Jones, H. R. Netherton & D. S. McClain. 1975. Comparative susceptibility of the chorioptic mange mite, northern fowl mites and brown dog tick to acaricides. Search Agric. 4(11): 1-31.

Maupin, G. O. and J. Piesman. 1994. Acaracide susceptibility of immature *Ixodes scapularis* (Acari: Ixodidae) as determined by the disposable pipet method. J. Med. Entomol. 31(2): 319-321.

McDougall, A. S. 1895. The ox-bot or warble-fly (*Hypoderma bovis*): Does it ever attack the horse? Vet. J. Ann. Comp. Path. 41: 451-452.

McDowell, R. E. & J. W. Smith. 1966. The future of animal breeding for resistance to disease and pests, pp. 148-155 *In* Pest Control by Chemical, Biological, Genetic, and Physical Means (Symp.) Section O, Agric. Am. Assoc. Adv. Sci. Dec. 26-31, 1964. Montreal, Canada. ARS 33-110.

McGowan, M. J. & R. W. Barker. 1980. A selected bibliography of tick-host resistance and immunological relationships. Bull. Entomol. Soc. Am. 26: 17-25.

McMullan, W. C. 1993. Skin disease of horses caused and/or transmitted by ectoparasites. In: Proceedings of the Second International Symposium on Ectoparasites of Pets. University of Kentucky, Lexington. pp. 56-58.

Mejlon, H. A., T.G.T. Jaenson and T. N. Mather. 1995. Evaluation of host-targeted applications of permethrin for control of *Borrelia*-infected *Ixodes ricinus* (Acari: Ixodidae). Med. Vet. Entomol. 9: 207-210.

Meleney, W. P. 1981. Control of psoroptic scabies on calves with ivermectin. Am. J. Vet. Res. 43: 329-331.
Miller, J. A. & J. E. George. 1994.

Efficacy of a combination neckband and tailtag containing amitraz or cyhalothrin K against lone star ticks on pastured cattle. J. Agric. Entomol. 11: 165-176.

Miller, R. J., R. L. Byford, G. S. Smith, M. E. Craig and D. Vanleeuwen. 1995.Influence of snakeweed foliage on engorgement, fecundity and attachment of the lone star tick (Acari: Ixodidae). J. Agric. Entomol. 12(2-3): 137-143.

Miller, J. A., D. D. Oehler and J. M. Pound. 1998. Delivery of ivermectin by injectable microspheres. J. Econ. Entomol. 91(3): 655-659.

Mock, D. E., D. B. Brillhart & S. J. Upton. 1992. Field ecology of Lyme disease in Kansas. Kans. Med. 93: 246-249.

Monsen, S. E., L. R. Bronson, C. R. Smith and J. Tucker. 1996. Tick control--a California study. SOVE Newsl. 27(3): 12.

Moreby, C. 1978. The biting louse genus Werneckiella (Phthiraptera: Trichodectidae) ectoparasitic on the horse family Equidae (Mammalia: Perissodactyla). J. Nat. Hist. 12(4): 395-412.

Morel, P. C. & G. Vassiliades. 1962. Les Rhipicephalus du groupe sanguineus: especes africaines (Acariens: Ixodoidea). Rev. Elev. Med. Vet. Pays Trop. 15: 343-386.

Mount, G. A. 1981a. *Amblyomma americanum*: Area control of overwintered nymphs and adults in Oklahoma with acaricides. J. Econ. Entomol. 74(1): 24-26.

Mount, G. A. 1981b. *Amblyomma americanum*: Control in Oklahoma parks with air blast sprayer applications of acaricides. J. Econ. Entomol. 74(1): 27-29.

Mount, G. A. 1983. Area control of larvae of lone star tick (Acari: Ixodidae) with acaricides. J. Econ. Entomol. 76(1): 113-116.

Mount, G. A. 1984. Aerial application of diazinon granules for area control of the lone star tick (Acari: Ixodidae). J. Econ. Entomol. 77: 1211-1213.

Mount, G. A., D. G. Haile and E. Daniels. 1997. Simulation of management strategies for the blacklegged tick (Acari: Ixodidae) and the Lyme disease spirochete, *Borrelia burgdorferi*. J. Med. Entomol. 34(6): 672-683.

Mulla, M. S., H. A. Darwazeh, L. Ede, B. Kennedy & H. T. Dulmage. 1985. Efficacy and field evaluation of *Bacillus thuringiensis* (H-14) and *B. sphaericus* against floodwater mosquitoes in California. J. Am. Mosq. Control Assoc. 1(3): 310-315.

Mullens, B. A. 1989. A quantitative survey of *Culicoides variipennis* (Diptera: Ceratopogonidae) in dairy wastewater ponds in southern California. J. Med. Entomol. 26(6): 559-565.

Murray, M. D. 1957. The distribution of the eggs of mammalian lice on their hosts. IV. The distribution of the eggs of *Damalinia equi* (Denny) and *Haematopinus asini* (L.) on the horse. Aust. J. Zool. 5: 183-187.

Muylle, E., W. Oyaert, and M. Rogiers. 1979. Endoscope examination of the horse stomach in the assessment of the efficacy of mebendazole/trichlorfon paste against *Gasterophilus intestinalis* larvae. Vlaams Diergeneeskundig Tijdschrift 48: 279-282.

Nelson, W. A. 1952. A note on the presence of first-stage larvae of *Gasterophilus intestinalis* (DeGeer) (Diptera: Gasterophilidae) in the mouth of the horse. Can. Entomol. 84: 356.

Nelson, W. A. 1989. Metabolic responses of livestock to hematophagous arthropod invasion. pp. 15-21 *In* Proc. Symp.: Physiological Interactions Between Hematophagous Arthropods and Their Vertebrate Hosts. Edited by C.J. Jones and R.E. Williams. Entomol. Soc. Am. Misc. Publ. 31.

Nelson, W. A., J. F. Bell, C. M. Clifford & J. E. Keirans. 1977. Interaction of ectoparasites and their hosts. J. Med. Entomol. 13: 389-428.

Nelson, W. A., J. E. Keirans, J. F. Bell & C. M. Clifford. 1975. Host ectoparasite relationships. J. Med. Entomol. 12: 143-166.

Olander, H. J. 1967. The migration of *Hypoderma lineatum* in the brain of a horse. Pathol. Vet. 4: 477-483.

Oliver, J. H. Jr., M. R. Owsley, H. J. Hutcheson, A. M. James, C. Chen, W. S. Irby, E. M. Dotson & D. K. McLain. 1993. Conspecificity of the ticks Ixodes scapularis and I. dammini (Acari: Ixodidae). J. Med. Entomol. 30: 54-63.

Panella, N. A., J. Karchesy, G. O. Maupin, J.C.S. Malan and J. Piesman. 1997. Susceptibility of immature *Ixodes scapularis* (Acari: Ixodidae) to plant-derived acaricides. J. Med. Entomol. 34(3): 340-345.

Patil-Kulkarni, V. G., V. S. Narasaput & M. A. Gafoor. 1967. Successful treatment of psoroptic mange in a horse with malathion emulsion. Indian Vet. J. 44(1): 65-67.

Patrican, L. A. and S. A. Allan. 1995a. Laboratory evaluation of desiccants and insecticidal soap applied to various substrates to control the deer tick *Ixodes scapularis*. Med. Vet. Entomol. 9: 293-299.

Patrican, L. A. and S. A. Allan. 1995b. Application of desiccant and insecticidal soap treatments to control *Ixodes scapularis* (Acari: Ixodidae) nymphs and adults in a hyperendemic woodland site. J. Med. Entomol. 32(6): 859-863

Petersen, L. R, L. A. Sawyer, D. B. Fishbein & P. W. Kelley. 1989. An outbreak of ehrlichiosis in members of an Army Reserve unit exposed to ticks. J. Infec. Dis. 159: 562-568.

Phelps, G. 1971. VEE in Texas. An on-the-scene report (July 28, 1971). Vet. Med., Small Animal Clinician 66(9): 911-913.

Pickens, L. G. & R. W. Miller. 1980. Biology and control of the face fly, *Musca autumnalis* (Diptera: Muscidae). J. Med. Entomol. 17: 195-210.

Pillers, A.W.N. 1923. *Hypoderma bovis* (DeGeer) and *H. lineatum* (DeVilliers) causing warbles in horses. Vet. J. 79: 329-331.

Pound, J. M., J. A. Miller, J. E. George, D. D. Oehler and D. E. Harmel. 1996. Systemic treatment of white-tailed deer with ivermectin-treated bait to control free-living populations of lone star ticks (Acari: Ixodidae). J. Med. Entomol. 33(3): 385-394.

Purnell, R. E., J. D. Dargie, A. D. Irvin & M. A. Legner. 1973. Co60 irradiation of the tick *Rhipicephalus appendiculatus*. Proceedings: Isotopes and radiation in parasitology. FAO/IAEA Division of Atomic Energy in Food and Agriculture. Vienna, Austria. 139-144.

Rabalais, F. C. & C. L. Votava. 1974. Cutaneous distribution of microfilariae of Onchocerca cervicalis in horses. Am. J. Vet. Res. 35(10): 1369-1370.

Ramachandra, R. N. & S. K. Wikel. 1992. Modulation of host-immune responses by ticks (Acari: Ixodidae): effect of salivary gland extracts on host macrophages and lymphocyte cytokine production. J. Med. Entomol. 29: 818-826.

Rechav, Y. 1987. Resistance of Brahman and Hereford cattle to African ticks with serum gamma globulin levels and blood composition. Exp. Appl. Acarol. 3: 219 232.

Rechav, Y., J. Dauth & D. A. Els. 1990. Resistance of Brahman and Simmentaler cattle to southern African ticks. Onderstep. J. Vet. Res. 57: 7-12.

Rechav, Y., A. Heller-Haupt & M.G.R. Varma. 1989. Resistance and cross-resistance in guinea-pigs and rabbits to immature stages of ixodid ticks. Med. Vet. Entomol. 3: 333-336.

Richter, P. J., Jr., R. B. Kimsey, J. E. Madigan, J. E. Barlough, J. S. Dumler and D.L. Brooks. 1996. *Ixodes pacificus* (Acari: Ixodidae) as a vector of *Ehrlichia equi* (Rickettsiales: Ehrlichieae). J. Med. Entomol. 33(1): 1-5.

Riek, E. F. 1954. Studies on allergic dermatitis (Queensland itch) of the horse: the aetiology of the disease. Austr. J. Agric. Res. 5: 109-129.

Samsinakova, A., S. Kalalova, M. Daniel, F. Dusbabek, E. Honzakova & V. Cerny. 1974. Entomogenous fungi associated with the tick Ixodes ricinus (L.). Folia-Parasit. 21(1): 39-48.

Scharff, D. K. 1973. Control of cattle grubs in horses. Vet. Med./Sm. Anim. Clin. 68: 791-792.

Schmidtmann, E. T., J .F. Carroll and D. W. Watson. 1998. Attachment-site patterns of adult blacklegged ticks (Acari: Ixodidae) on white-tailed deer and horses. J. Med. Entomol. 35(1): 59-63

Scholl, P. J. 1990. A review of parasites, pathogens, and predators of cattle grubs. Southwest. Entomol. 15: 360-365.

Scholl, P. J. 1993. Biology and control of cattle grubs. Ann. Rev. Entomol. 39: 53-70.

Scholl, P. J. & C. C. Barrett. 1986. Technique to extract *Hypoderma* spp. (Diptera: Oestridae) larvae from the backs of cattle. J. Econ. Entomol. 79: 1125-1126.

Scholl, P. J., J. J. Petersen, D. A. Stage and J. A. Meyer. 1981. Open silage as an overwintering site for immature stable flies in eastern Nebraska. Southwest. Entomol. 6: 253-258.

Scholl, P. J., D. D. Colwell, J. Weintraub & S. E. Kunz. 1986. Area-wide systemic insecticide treatment for control of cattle grubs, *Hypoderma* spp. (Diptera: Oestridae): Two approaches. J. Econ. Entomol. 79: 1558-1563.

Schulze, T. L. and R. A. Jordan. 1995. Potential influence of leaf litter depth on effectiveness of granular carbaryl against subadult *Ixodes scapularis* (Acari: Ixodidae). J. Med. Entomol. 32(2): 205-208.

Schulze, T. L., G. S. Bowen, E. M. Bosler, M. F. Lakat & B. G. Ormiston. 1984. Amblyomma americanum: a potential vector of Lyme disease in New Jersey. Science. 224(4649): 601-603.

Schulze, T. L., G. C. Taylor, R. A. Jordan, E. M. Bosler & J. K. Shisler. 1991. Effectiveness of selected granular acaricide formulations in suppressing populations of *Ixodes dammini* (Acari: Ixodidae): short-term control of nymphs and larvae. J. Med. Entomol. 28: 624-629.

Schulze, T. L., G. C. Taylor, L. M. Vasvary, W. Simmons & R. A. Jordan. 1992. Effectiveness of an aerial application of carbaryl in controlling *Ixodes dammini* (Acari:

Ixodidae) adults in a high-use recreational area in New Jersey. J. Med. Entomol. 29: 544-547.

Schulze, T. L., R. A. Jordan, L. M. Vasvary, M. S. Chomsky, D. C. Shaw, M. A. Meddis, R. C. Taylor & J. Piesman. 1994. Suppression of *Ixodes scapularis* (Acari: Ixodidae) nymphs in a large residential community. J. Med. Entomol. 31: 206 211.

Schulze, T. L., R.A. Jordan and R.W. Hung. 1995. Suppression of subadult *Ixodes scapularis* (Acari: Ixodidae) following removal of leaf litter. J. Med. Entomol. 32(5): 730-733.

Seibert, B. P., K. M. Newcomb & B. F. Michael. 1986. Critical test and safety evaluations of an oral paste preparation of mebendazole and trichlorfon in horses. Am. J. Vet. Res. 47: 1347-1350.

Shahan, M. S. & L. T. Giltner. 1945. A review of the epizootiology of equine encephalomyelitis in the United States. J. Amer. Vet. Med. Assoc. 107(824): 279 288.

Shefstad, D. K. 1978. Scanning electron microscopy of *Gasterophilus intestinalis* lesions of the equine stomach. J. Am. Vet. Med. Assoc. 172: 310-313.

Skoda, S. R. & G. D. Thomas. 1992. The stable fly as a pest of humans. In: Thomas and Skoda (eds), The Stable Fly: A Pest of Humans and Domestic Animals. USDA-ARS and University of Nebraska IANR MP 64.

Smith, T. A. & D. D. Linsdale. 1967. First supplement to an annotated bibliography of the face fly, Musca autumnalis DeGeer, in North America. Calif. Vector Views 14: 74-76.

Smith, T. A., D. D. Linsdale & D. J. Burdick. 1966. An annotated bibliography of the face fly Musca autumnalis DeGeer, in North America. Calif. Vector Views 13: 43 54.

Snow, W. E., E. Pickard & C. M. Jones. 1958. Observations on the activity of Culicoides and other Diptera in Jasper County, South Carolina. Mosq. News 18: 18-21.

Solberg, V. B., K. Neidhardt, M. R. Sardelis, F. J. Hoffmann, R. Stevenson, L. R. Boobar & H. J. Harlan. 1992. Field evaluation of two formulations of cyfluthrin for control of *Ixodes dammini* and *Amblyomma americanum* (Acari: Ixodidae). J. Med. Entomol. 29: 643-638.

Sonenshine, D. E. & G. Haines. 1985. A convenient method for controlling populations of the American dog tick, *Dermacentor variabilis* (Acari: Ixodidae), in the natural environment. J. Med. Entomol. 22: 577-583.

Sonenshine, D. E., & G. F. Levy. 1971. The ecology of the lone star tick, *Amblyomma americanum* (L.), in two contrasting habitats in Virginia (Acarina: Ixodidae). J. Med.

Entomol. 8: 623-635. Sonenshine, D. E., S. A. Allan, R. Andrew, I. Norval and M. J. Burridge. 1996. A self medicating applicator for control of ticks on deer. Med. Vet. Entomol. 10: 149 154.

Spickett, A. M. 1987. The natural control of heartwater vectors. Onderstepoort J. Vet. Res. 54: 535-539.

Srivastava, P. S. & N. N. Sharma. 1976. Effects of cobalt-60 irradiation on the early developmental stages of an ixodid tick, Hyalomma anatolicum. Int. J. Radiat. Biol. 29(2): 159-168.

Stafford, K. C., III. 1991. Effectiveness of host-targeted permethrin in the control of *Ixodes dammini* (Acari: Ixodidae). J. Med. Entomol. 28: 611-617.

Stafford, K. C., III. 1992. Third-year evaluation of host-targeted permethrin for the control of *Ixodes dammini* (Acari: Ixodidae) in southeastern Connecticut. J. Med. Entomol. 29: 717-720.

Stafford, K. C., III. 1993. Reduced abundance of *Ixodes scapularis* (Acari: Ixodidae) with exclusion of deer by electric fencing. J. Med. Entomol. 30: 986-996.

Stafford, K. C. III, A. J. Denicola and L. A. Magnarelli. 1996. Presence of *Ixodiphagushookeri* (Hymenoptera: Encyrtidae) in two Connecticut populations of *Ixodes scapularis* (Acari: Ixodidae). J. Med. Entomol. 33(1): 183-188.

Steelman, C. D., E. E. Gbur, G. Tolley & A. H. Brown, Jr. 1993a. Individual variation within breeds of beef cattle in resistance to horn fly (Diptera: Muscidae). J. Med. Entomol. 30: 414-420.

Steelman, C. D., E. E. Gbur, G. Tolley & A. H. Brown, Jr. 1993b. Variation in population density of the face fly, *Musca autumnalis* DeGeer, among selected breeds of beef cattle. J. Agric. Entomol. 10: 97-106.

Steelman, C. D. & P. E. Schilling. 1977. Economics of protecting cattle from mosquito attack relative to injury thresholds. J. Econ. Entomol. 70: 15-17. Sternberg, S., B. A. Peleg & R. Galun. 1973. Effect of irradiation on mating competitiveness of the male tick Argas persicus (Oken). J. Med. Entomol. 10(2): 137-142.

Stoffolano, J. G. Jr. 1968.The effect of diapause and age on the tarsal acceptance threshold of the fly, *Musca autumnalis*. J. Insect Physiol. 14(9): 1205-1214.

Strickland, R. K., R. R. Gerrish, J. L. Hourrigan & G. O. Schubert (revision). 1976. Ticks of veterinary importance. Animal and Plant Health Inspection Service. USDA Ag. Hndbk. 485: 122.

Strother, G. R., E. C. Burns & L. I. Smart. 1975. Comparing purebred Brahman, Hereford and Brahman x Hereford crossbred cattle on inherited and acquired resistance to the lone star tick (*Amblyomma americanum*). La. Agric. Exp. Sta., Livestock Prod. Day, La. State Univ. Anim. Sci. Dep. 15: 271-275.

Sukhapesna, V., F. W. Knapp, E. T. Lyons & J. H. Drudge. 1975. Effect of temperature on embryonic development and egg hatchability of the horse bot, *Gasterophilus intestinalis* (Diptera: Gasterophilidae). J. Med. Entomol. 12: 391-392.

Sutherst, R. W. 1971. An experimental investigation into the effects of flooding on the ixodid tick Boophilus microplus (Canestrini). Oecologia 6(3): 208-222.

Tatchell, R.J. 1987. Interactions between ticks and their hosts. Int. J. Parasitol. 17: 597-606.

Teel, P. D. 1985. Ticks. In Livestock Entomology. R. E. Williams, R.D.Hall, A.B. Broce & P.J. Scholl eds. John Wiley and sons, New York. pp. 129-149.

Teel, P. D., W. A. Donahue, O. F. Strey and R. W. Meola. 1996. Effects of pyriproxyfen on engorged females and newly oviposited eggs of the lone star tick (Acari: Ixodidae). J. Med. Entomol. 33(5): 721-725.

Tembo, S. D. & Y. Rechav. 1992. Immunization of rabbits against nymphs of *Amblyomma hebraeum* and *A. marmoreum* (Acari: Ixodidae). J. Med. Entomol. 29: 757-760.

Tolliver, S. C., E. T. Lyons & J. H. Drudge. 1974. Observations on the specific location of *Gasterophilus* spp. larvae in the mouth of the horse. J. Parasitol. 60: 891-892.

Townsend, L. H., Jr. & E. C. Turner Jr. 1976. Field evaluation of several chemicals against ear feeding black fly pests of horses in Virginia. Mosq. News 36(2): 182 186.

Townsend, L. H., E. C. Turner, & W. A. Allen. 1977. An assessment of feeding damage threshold of *Simulium vittatum*, a black fly pest of horses in Virginia. Mosq. News 37(4): 742-744.

Trager, W. 1939a. Acquired immunity to ticks. J. Parasitol. 25: 57-81.

Trager, W. 1939b. Further observation on acquired immunity to the tick *Dermacentor variabilis* Say. J. Parasitol. 25: 137-139.

USDA. 1972. Venezuelan equine encephalomyelitis, a natural emergency. USDA, APHIS-81-1, 24pp.

USDA. 1973. The origin and spread of Venezuelan equine encephalomyelitis. USDA, APHIS-91-10, 51pp.

USDA-APHIS. 1997. NAHMS Equine '98 Needs Assessment Survey Results.

USDA. National Agricultural Statistics Service. 1999.
http://usda.mannlib.cornell.edu/reports/nassr/livestock/equine/eqinan99.txt

Valder, S. M., T. L. Hopkins & S. A. Valder. 1969. Diapause induction and changes in lipid composition in diapausing and reproducing face flies, Musca autumnalis. J. Insect Physiol. 15(7): 1199-1214.

van Dam, R.H. 1981. Definition and biological significance of the major histocompatability system (MHS) in man and animals. Vet. Immunopathol. 2: 517-539.

Varma, M.G.R., A. Hellerhaupt, P.K.E. Trinder & A. O. Langi. 1990. Immunization of guinea-pigs against *Rhipicephalus appendiculatus* adult ticks using homogenates from unfed immature ticks. Immunology 71: 133-138.

Wambura, P. N., P. S. Gwakisa, R. S. Silayo and E. A. Rugaimukumu. 1998. Breed associated resistance to tick infestation in *Bos indicus* and their crosses with *Bos taurus*. Vet. Parasitol. 77: 63-70.

Webb, J. D. & F. W. Knapp. 1993. Evaluation of citronella for repellency of biting arthropods on horses, 1992. Insecticide & Acaricide Tests 18: 358-59.

Wharton, R. H. & K. R. Norris. 1980. Control of parasitic arthropods. Vet. Parasitol. 6: 135-164.

Wikel, S. K. 1982. Immune responses to arthropods and their products. Annu. Rev. Entomol. 27: 21-48.

Wikel, S. K., R. N. Ramachandra & D. K. Bergman. 1992. Immunological strategies for suppression of vector arthropods: novel approaches in vector control. Bull. Soc. Vector Ecol. 17: 10-19.

Wikel, S. K., & A. C. Whelen. 1986. Ixodid-host immune interaction. Identification and characterization of relevant antigens and tick-induced host immunosuppression. Vet. Parasitol. 20: 149-174.

Wilkenson, P. R. 1977. Effect of herbicidal killing of shrubs on abundance of adult Dermacentor andersoni in British Columbia. J. Med. Entomol. 13: 713-718.

Willadsen, P. 1987. Immunological approaches to the control of ticks. Int. J. Parasitol. 17: 671-677.

Williams, R. E. 1989. Susceptibility of Hereford and Brahman cattle to *Amblyomma* (Acari: Ixodidae) tick infestations. pp. 33-36 *In* Proc. Symp.: Physiological Interactions

Between Hematophagous Arthropods and Their Vertebrate Hosts. Edited by C.J. Jones and R.E. Williams. Entomol. Soc. Am. Misc. Publ. 31.

Wilson, J. G., D. R. Kinzer, J. R. Sauer & J. A. Hair. 1972. Chemo-attraction in the lone star tick (Acarina: Ixodidae): I. Response of different developmental stages to carbon dioxide administered via traps. J. Med. Entomol. 9: 245-252.

Wilson, M. L. 1986. Reduced abundance of adult *Ixodes dammini* (Acari: Ixodidae) following destruction of vegetation. J. Econ. Entomol. 79: 693-696.

Wilson, M. L., J. F. Levine & A. Spielman. 1984. Effect of deer reduction on abundance of the deer tick (*Ixodes dammini*). Yale J. Biol. Med. 57: 697-705.

Wong, J.Y.M., & J. P. Opdebeeck. 1989. Protective efficacy of antigens solubilized from gut membranes of the cattle tick, *Boophilus microplus*. Immunology 66: 149-155.

Wong, J.Y.M., J. H. Duffy & J. P. Opdebeeck. 1990. The expression of bovine lymphocyte antigen and response of Hereford cattle to vaccination against *Boophilus microplus*. Int. J. Parasitol. 20: 677-680. Yong, W. K. (ed.) 1992. Animal Parasite Control Utilizing Biotechnology. CRC Press, Boca Raton, Fla. 496 pp.

Zehmer, R. B., P. B. Dean, W. D. Sudia, C. H. Calisher, G. E. Sather & R. L. Parker. 1971. Venezuelan equine encephalitis epidemic in Texas, 1971. Health Services Reports 89(3): 278-282.

Zhioua, E., M. Browning, P. W. Johnson, H. S. Ginsberg and R. A. LeBrun. 1997. Pathogenicity of the entomopathogenic fungus *Metarhizium anisopliae* (Deuteromycetes) to *Ixodes scapularis* (Acari: Ixodidae). J. Parasitol. 83(5): 815 818.

****
****

End of Horses section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****
****

DOGS AND CATS SUMMARY

1. In the U.S., 58% (54.8 million) of all households own a companion animal. The total dog population is approximately 53 million, while the total cat population is estimated at 57 million.

2. Each year, 82% of dog owners and 62% of cat owners obtain veterinary care for their animals. Dog owners average 2.6 veterinary visits per year while cat owners average 1.8 visits to their veterinarian. Annual veterinary expenditures total $4.6 billion for dogs and $2.3 billion for cats.

3. Fleas are both the most common and the most costly pest of dogs and cats. The second most costly pest is mosquitoes, due to their serving as vectors of dog heartworm.

4. Costs of control of arthropod pests of dogs and cats combined with prevention and treatment of their associated diseases is estimated at $11 billion per year.

COMPANION ANIMALS

Committee Members

Dr. Nancy C. Hinkle, Chair
Dr. Michael W. Dryden
Dr. Philip G. Koehler
Dr. Donald E. Mock
Dr. Richard S. Patterson
Dr. Philip J. Scholl
Mr. William B. Warner

Economic Significance of Companion Animals

Dog and cat ownership contributes over $80 billion to the American economy every year (Anthony & McManus 1991, Jaegerman 1992). Estimates of annual expenditures for flea control, including homeowner markets as well as veterinary suppliers and pest control companies, exceed $4.6 billion. The amount spent on prevention, diagnosis and treatment of arthropod-transmitted diseases varies by region, but probably totals over $6.2 billion for dogs and around $1 billion for cats, not counting the cost of pest control. There is the additional factor of diseases transmitted from pets to humans, including rare conditions like plague, tularemia, ehrlichiosis, and cat scratch fever (Koehler et al. 1994).

Arthropod Pests of Dogs and Cats

The primary pest of dogs and cats is the cat flea. Additional pests include lice, mites, ticks, mosquitoes and flies.

Fleas, *Ctenocephalides felis* Bouché, *C. canis* (Curtis), *Echidnophaga gallinacea* (Westwood), and *Pulex simulans* Baker

Description and Biology. Members of the order Siphonaptera are small (< one quarter inch), wingless, laterally-compressed insects which are obligate hematophagous ectoparasites of mammals and some birds. Most fleas have backwardly projecting spines that permit forward movement while counteracting grooming efforts of the host. The flea's hind legs are adapted for jumping, and hooked tarsi enable attachment to the host pelage or plumage.

The cat flea, *Ctenocephalides felis felis* Bouché, is the most common ectoparasite of dogs and cats in North America. In the southern United States it is a year-round pest, while its season varies in more northerly climes. Fleas are second only to ants in the number of inquiries generated for extension and research personnel (Olsen 1987). In seemingly intractable situations, pet-owners will try almost anything to alleviate the suffering of their pets and themselves, in some cases going so far as to euthanize the pet (Anderson 1969, Fenster 1985). Blood loss to flea feeding is such a problem that veterinarians occasionally see kittens and puppies near death from flea-produced anemia (Whiteley 1987, Yaphe et al. 1993).

Additionally, the cat flea serves as the obligate intermediate host of *Dipylidium caninum*, the most common tapeworm of dogs and cats. The causative agent of murine typhus, *Rickettsia typhi*, is transmitted from the rodent reservoir by several species of fleas, including *C. felis*. Fleas and their associated diseases can make up over half of the caseload of veterinary practices in some areas of the country. More energy and money are spent battling these insects than any other problem in veterinary medicine (Scheidt 1988).

The life cycle of the dog tapeworm, *D. caninum*, and its interaction with its obligate intermediate host, *C. felis*, are intricately entwined (Chen 1934). Flea larvae, in their indiscriminate feeding on organic debris in their environment, encounter and bite into tapeworm proglottids, in the process ingesting the eggs they contain. The tapeworm eggs hatch and the immature stages develop within the flea larva as it goes through its subsequent metamorphosis. In the adult flea, the metacestodes complete their development within the body cavity of the adult flea. In the course of its grooming, the host (cat or dog) ingests the adult flea, the tapeworm metacestode breaks out of the flea carcass, attaches to the intestinal wall of the mammal, and begins a new phase of its existence. As the tapeworm matures, terminal segments fill with eggs, break off and travel down the intestinal tract. The proglottids actively crawl out of the anus and quickly dry to the characteristic egg packets upon exposure to air.

Flea allergy dermatitis (miliary dermatitis, weeping dermatitis, etc.) is a severe condition of flea allergy in which the hyperallergenicity of the animal causes exaggerated responses to the flea allergen. Flea allergy dermatitis (FAD) is a severe condition found primarily in dogs, but also occasionally seen in cats. It is a hypersensitivity reaction to the allergens in flea salivary secretions, manifested as intense pruritus accompanied by uncontrollable scratching resulting in self-mutilation. An affected animal typically displays obsessive grooming behavior, with accompanying depilation. The extreme scratching, licking and biting usually leaves the skin with weeping sores, often resulting in secondary bacterial skin infection. In a flea allergic animal, the antigens in a single flea bite are sufficient to initiate the entire cascade of symptoms (Slacek & Opdebeeck 1993).

Treatment of FAD has traditionally consisted of breaking the flea life cycle in the environment, controlling flea infestations on the pet, controlling and treating symptoms, and attempting to hyposensitize the allergic animal. Antihistamines have been used alone or in combination with glucocorticosteroids to help control inflammation and pruritus (Scheidt 1988).

Hyposensitization consists of administering allergens to a hypersensitive animal on a regular basis in an attempt to obtain a state of clinical non-reactivity to the bite of fleas. The effectiveness of currently available whole flea extracts is controversial. While some studies have indicated success, doubleblind studies in both dogs and cats have shown little efficacy (Halliwell 1981, Kunkle & Milcarsky 1985).

The cat flea is a cosmopolitan, eclectic species, having been recorded from over two dozen species, including kangaroos, opossums, raccoons, and even birds (Hinkle 1992). Its wide host range explains the flea's ability to repopulate domestic animals following "eradication" efforts. Because of its lack of host specificity and tendency to feed on humans, the cat flea is a pest of both our companion animals and of the humans with whom they share their abode. Reactions of humans to flea bites vary from individual to individual, ranging from simple irritation due to the mechanical injury of the mouthparts piercing the skin to severe rash, itching, and prolonged allergic response.

Only the adult stage is parasitic. Eggs are laid on the host, but sift through the coat and collect in the environment. The eggs are approximately 1 mm in length, with little surface structure other than aeropyles (permitting gaseous exchange for the developing embryo) and micropyles (for entrance of the sperm during fertilization). From the eggs hatch small, white, legless, eyeless larvae with chewing mouthparts.

Adult cat fleas have been demonstrated to spend their entire lives on the host, once they have successfully procured one, unlike rodent fleas and other nest fleas (Dryden 1989). The adults feed, mate, and reproduce on the host. The females lay eggs which sift through the coat of the animal, and collect in the environment. Simultaneously, adult flea feces that have collected in the coat fall into the same environment and serve as provisions for the larvae once they hatch from the eggs. Typically the eggs hatch within 24-48 hours of being laid. The larvae go through three instars that, under favorable conditions, can be completed in as little as ten days. Cooler temperatures, lack of food, or other unsuitable environmental conditions may stretch the larval developmental time to several weeks or months (Kern 1993). The third instar larva voids its gut approximately 24 hours prior to initiation of cocoon construction. The white prepupa wanders until it locates an appropriate site for pupation and then begins to spin a silk cocoon. Frequently, debris from the surroundings are incorporated into the cocoon, being tied together by the sticky silk fibers, so that the cocoon may appear as a small dirt clod or lint ball.

Within the cocoon, the prepupa molts to the pupa and continues metamorphosis to the adult flea within about four days under good conditions. This pre-emerged adult stage is the most variable in the life cycle of the flea, ranging from less than a day to several months (or perhaps a year). The mechanisms are not understood, but it appears that some individuals are programmed to delay emergence. Likely this is an evolutionary strategy whereby the offspring emerge over an extended interval, ensuring that some successfully encounter hosts. Stimuli such as pressure, carbon dioxide, and warmth have been demonstrated to serve as releasers, causing the adult flea to emerge from the cocoon. These are triggers that would logically be associated with a mammalian host. If the flea emerges and does not locate a host immediately, it can survive for approximately a week to ten days, or even longer under conditions of high humidity and low temperatures (Silverman et al. 1981).

Once the adult flea has attached to a host, it begins to feed, mates, and the females begin egg-laying within a couple of days. If a flea is dislodged from its host after initiating feeding, its survival time is greatly reduced, unless it can regain a host. On the host, a female flea averages about one egg per hour and, as a female flea can live for several weeks, its potential production can be over two thousand eggs in its lifetime. However, host grooming is the most significant mortality factor for adult fleas, with most being removed by the host within a week, so the actual egg production per female lifetime is more like two hundred eggs (Hinkle 1992).

Other natural mortality factors include desiccation of eggs and larvae, which have very low tolerance for relative humidities below 50%. Generalized predators, such as ants, can have a significant impact on larval numbers.

Echidnophaga gallinacea, the sticktight flea, is usually found on poultry, but it is not uncommonly a pest on cats and dogs which associate with infested chickens (Harman et al. 1987). The male flea is a mobile ectoparasite throughout its adult life while the female attaches herself permanently to the host with her mouthparts and remains in that position for the rest of her life. Sticktight fleas typically attach to the face and ear margins of cats and dogs or between the toes of dogs. The presence of sticktight fleas may predispose the tissue to secondary infection.

Pulex simulans, the human flea of the New World, is primarily a flea of medium to small sized wild mammals. It is common on raccoons and opossums, may be found on swine, and occasionally transfers to dogs (Harman et al. 1987).

Economic Importance. Not only do flea bites produce itching and urticaria, leading to scratching, alopecia, and secondary infection, but sensitive animals develop flea allergy dermatitis (with symptoms of dermatitis, racing stripes, weeping patches, hot spots). Fleas serve as the obligate intermediate hosts of the dog tapeworm, *D. caninum*, of both cats and dogs. Fleas also transfer to humans and cause itching. The market for environmental flea control is estimated at $1 billion. On-animal flea treatment is estimated to run $3.6 billion per year. Testing and treatment of ancillary conditions (FAD) and flea-borne diseases (tapeworm) cost an additional $2 billion (Hinkle unpublished data).

Methods of Control. Dryden and Rust (1994) provide a comprehensive overview of flea suppression strategies. There appear to be few effective control options for fleas, other than chemical insecticides (Donahue & Stemme 1992). Insecticide groups available for on-animal use include botanicals (Hinkle 1995), pyrethrins, pyrethroids, carbamates and organophosphates (Dryden et al. 1989). Compounds such as the chloronicotinyls and phenylpyrazoles (represented by imidacloprid and fipronil, respectively) combine ease of use and longterm efficacy (Franc & Cadiergues 1998). Topical application of insect growth regulators, while not killing the adult fleas, can reduce the population over time by eliminating viable eggs. The chitin synthesis inhibitor lufenuron (Program®) is formulated for ingestion and is transferred to fleas as they feed on the host, resulting in non-viable eggs being produced by the females. Mechanical control may be effected using a flea comb to extract fleas from the coat of the host. Other than insecticides, the most significant on-host mortality factor is host grooming (Hinkle 1992).

Premise treatments include both adulticides (typically carbamate, organophosphate, or pyrethroid insecticides) and insect growth regulators such as methoprene and pyriproxyfen (Hinkle et al. 1995a). Carpet treatment with various borate products have been demonstrated efficacious in killing flea larvae (Hinkle et al. 1995b). Sanitation is helpful in removing flea eggs before they hatch and reducing the food available to developing larvae. Flea traps may be useful for population monitoring, but probably will not eliminate a population (Dryden & Broce 1993). Mechanical controls include keeping the animal isolated from a chance of infestation. This may work with cats that can be confined indoors all the time, but does not work well with dogs that must occasionally be allowed outdoors, even for brief intervals (Dryden & Prestwood 1993). The catholic taste in hosts exhibited by *C. felis* means that virtually any mammal may serve as a source of inoculum, so any wild animal passing through the property may provide flea contamination to be acquired by any passing pet.

Biological control possibilities have only begun to be investigated (Hinkle et al. 1997). Most researchers looking for potential parasites and pathogens have found only marginally harmful symbionts. These include the protozoa and bacteria identified by Beard et al. (1990). *Bacillus thuringiensis* was demonstrated to be larvicidal for fleas (Maciejewska et al. 1988). As with any on-host pest, there is little opportunity for establishing populations of beneficial arthropods to serve as either parasites, predators, or competitors of the ectoparasites. In general, any arthropod is objectionable on our pets. The only biocontrol option marketed for flea control is the nematode *Steinernema* *carpocapsae*, available for suppression of immature stages in outdoor habitats (Manweiler 1994, Henderson et al. 1995).

Vaccination as a means of flea control is being explored (Heath et al. 1994). The immunology of reactions to flea antigen is poorly understood. Flea bites typically produce severe urticaria, often leading to frenzied scratching and self-induced alopecia and excoriation. Attempts at inducing host immunity have required isolation of the allergenic fraction to prevent such severe side-effects.

Ear Mites, *Otodectes cynotis* (Hering)

Description and Biology. Otoacariasis in cats, dogs, ferrets, and foxes is caused by auricular mites, *Otodectes cynotis* (Hering). Tremendous numbers of these mites can develop in the outer ear canal, producing a dark oily exudate, itching and tenderness. In addition to their wastes, irritation from their presence causes catarrh, which is diagnostic for the condition. In efforts to relieve the discomfort, animals typically shake their heads, exhibiting behavior that has been described as "fits" (Schneck 1988). This frantic urge of an animal to scratch its ears is, in fact, diagnostic of ear mites.

Otitis externa is an inflammatory condition of the outer ear, primarily seen in cats (84% of cases) and sometimes in dogs (5-10% of cases). Most infestations occur at an early age with the development of an immune response that helps to prevent later infestations (August 1988). Mites spend their entire life in the ears, taking approximately 3 weeks to develop from eggs to adults. *Otodectes* sp. mites are rarely found on feet, face, neck and the tailhead (Foley 1991). If mites are dislodged from their host, they can survive off the host for several weeks (Kwochka 1987). Ear mite infestations classically result in pruritus and self-induced trauma, creating the ideal environment for secondary invasion by opportunistic bacteria or fungi.

Economic Importance. Annual U.S. expenditures for treatment of ear mites is estimated at $838 million (Hinkle, unpublished data).

Methods of Control. Standard treatment of ear mites consists of cleaning the exudate from the ears and regular, every other day (for 3 weeks) treatments of the ear with commercial miticidal otic preparations. While not FDA approved, ivermectin (300 µg/kg) applied orally or subcutaneously at 2 to 3 week intervals is widely used (Kwochka 1987, Jeneskog & Falk 1990, Fukase et al. 1991, Song 1992). Ivermectin is approved only for use in dogs at 6 µg/kg as a heartworm preventative, but is widely used to treat a variety of internal and external parasitic diseases in dogs, cats and exotic animals at doses often exceeding 200 µg/kg. Potential adverse reactions include coma and death. In addition, certain breeds of dogs have a history of adverse reactions at these high doses, including Collies, Old English Sheep dogs and Australian Shepherds. The standard treatment for ear mites was mineral oil, massaged deeply into the canal. Pyrethrins may be dissolved in the oil and frequently an antibiotic is included to counteract any accompanying secondary infection.

Sarcoptes scabiei (DeGeer) and *Notoedres cati* (Hering)

Description and Biology. Sarcoptic mange (common mange) of dogs is caused by the *mite Sarcoptes scabiei* (DeGeer) var. *canis*. The infestation appears first on areas with sparse hair such as the ventral abdomen, chest, ears, elbows and ventrum. The disease then gradually spreads over the entire body, with mites infesting the back, abdomen and other portions of the body in severe infestations (Parish & Schwartzman 1993). *Sarcoptes* mites are roughly circular, females 330-600 µm and males 200-240 µm. All legs of both sexes are short, not extending beyond the body. The dorsal surface of *Sarcoptes* is covered with triangular spines while *Notoedres* has a "thumbprint" pattern of folds and no spines.

Cats are rarely infested by scabies mites (Kershaw 1989) but instead have *Notoedres cati* (Hering) (Scheidt 1987; Kwochka 1987). Notoedric mange of cats begins at the tips of the ears and gradually spreads over the face and head. The animal's scratching leads to self-inflicted trauma, producing lichenified and wrinkled skin covered with crusts (Parish & Schwartzman 1993).

The female mite burrows into skin, forming a tunnel in which she lays 40-50 eggs during her life. The eggs hatch in 3-5 days, producing 6-legged larvae that may stay in the tunnel or wander on the skin (Kwochka 1987). If they remain in the tunnel, they molt to nymphs. The final molt to adult stage occurs 17-21 days after the eggs are laid. Infestations are spread primarily through direct contact. Mites do not live very long off the host. At 68-77šF mites can live 5 to 6 days if the humidity is above 75% (Arlian & Vyszenski-Moher 1988). *Notoedres* are commonly found in group "nests" in the tunnels; *Sarcoptes* adult females are found singly.

Mites live in the keratin layer of the epidermis and are transmitted primarily by direct contact. Larvae and nymphs die in about 24 hours off the host. It has been reported that adults can survive off the host up to 10 days (Kwochka 1987, Lewis 1989).

Female mites burrow deeply in the skin, while other stages feed on surface debris. Mite activity produces irritation. The primary lesion is a cutaneous inflammation, resulting in pruritus. The animal's scratching and biting result in self trauma, causing alopecia and epidermal/dermal disruption. This results in secondary bacterial infection, serum exudation, and crusting. In chronic mite infestations the skin becomes thin and wrinkled. The distribution of lesions is generally in thin-haired parts of the body such as the ventral abdomen, chest, legs, elbows and hocks. The inflammatory reaction is the result of a hypersensitivity (Foley 1991, Moriello 1991) most likely produced by mite by-products. In dogs there is usually a rapid onset of seborrhea with or without pruritus. Cats generally

have a gradual increase in miliary lesions or generalized dandruff. Erythematous papules occur on the head, neck and back.
Asymptomatic carriers are common, especially in cats.

Diagnosis is based on a history of rapid onset with dorsal seborrhea with or without pruritus, recent group contacts (dog or cat shows, recently acquired from kennel, etc.), dermatologic lesions on people living in the household, and recovery of mites.

Humans can be transiently infested but mites apparently will not reproduce (Webster & Uhler 1986). Pruritus may last 2 to 3 weeks (Merchant 1990, Ackerman 1991). Scabies acquired from dogs or cats is a self-limiting disease in humans, with symptoms resolving spontaneously, as the infested animal is treated (Burgess 1994). It is not necessary to treat the environment.

Economic Importance. Treatment of all mange conditions in dogs and cats is estimated at $282 million per year (Hinkle, unpublished data).

Methods of Control. *Sarcoptes* is generally cured quite easily. Due to the presence of crusts and burrowing nature of mites, hair should be clipped and the animal bathed in antiseborrheic shampoo prior to treatment with a dip. Parasiticidal dips are used once a week or every two weeks until a cure is effected, typically for 3 to 5 dips. Compounds commonly used include lindane, chlorpyrifos, phosmet, lime sulfur, and amitraz (Knapp 1978, Kwochka 1987, Yathiraj et al. 1990). Also, while not an approved treatment, oral or subcutaneous ivermectin (at 200 µg/kg twice at 2 to 3 week intervals) has been very effective (Scheidt et al. 1984, Campbell 1989). Ivermectin should not be used in Collies, Old English Sheep Dogs and Australian Shepherds.

Cats infested with *Notoedres* are generally treated with lime sulfur or phosmet dips (Kwochka 1987, Grant 1989). Secondary pyoderma must be treated along with elimination of the mites. Antibiotic selection should be based upon culture and sensitivity. While mites do not typically live long off the host (only 2-3 days), in commercial facilities (i.e. kennels and pet stores) it may be necessary to treat for environmental contamination. Under conditions of low temperatures and high relative humidities, mites may survive off the host for over a week (Arlian et al. 1989). Environmental application of chlorpyrifos may be effective.

Demodex canis Leydig and *Demodex cati* Megnin

Description and Biology. Generalized demodectic mange (red mange) is a severe non-contagious disease caused by the microscopic alligator shaped mite, *Demodex canis* Leydig, that lives in hair follicles. Typically the mite infestation is accompanied by secondary infection with *Staphylococcus pyogenes albus*. Sustained conscientious effort is required to achieve remission of demodectic mange. The lesions occur chiefly around the eyes and muzzle and on the front feet. Newborn pups do not harbor mites. Transmission occurs during the first 72 hours of life while the pup is nursing. Evidence is that all dogs harbor *Demodex* mites and that susceptibility is influenced by the individual immune response of the animal. Demodectic mange can terminate fatally, even with treatment. *Demodex cati* Megnin causes a mild mange in cats.

There are two forms of demodicosis, localized and generalized demodectic mange. Localized demodicosis is characterized by well-demarcated areas of alopecia, erythema and scaling, usually confined to areas around the lips, peri-orbital area, and forelimbs. Typically there is no pruritus or secondary pyoderma. It is generally self limiting and not related to immune defect or heredity.

Generalized demodicosis occurs principally in young purebred dogs. The initial lesions of generalized demodicosis are similar to localized demodicosis, but as the disease progresses large areas of the body may be affected. Pruritus is a frequent complaint, due primarily to secondary bacterial infection. The animal may have a rancid odor (bacterial action on skin lipids), generalized lymphadenopathy, fever, anorexia and general debilitation. Approximately 10% of localized cases will progress to generalized (Kwochka 1987).

Generalized demodicosis is considered to be the result of an inherited immunologic defect, which is a functional abnormality associated with the cell-mediated (T-cell) immune system (Miller 1980, Folz 1983, Kwochka 1987). Dogs with localized demodicosis have normal electrophoretic (globulin) patterns and normal T-cell function, whereas dogs with generalized demodicosis have severely depressed T-cell responses (Scott et al. 1974, Scott et al. 1976).

Economic Importance. Treatment of all mange conditions in dogs and cats is estimated at $282 million per year (Hinkle, unpublished data).

Methods of Control. Because most dogs with localized demodicosis have no immunologic defect, the prospect for self-cure is excellent. Only approximately 10% of localized cases will become generalized. If lesions are asymptomatic and owner understands lesions should resolve spontaneously, no treatment is necessary.

The prognosis for generalized demodicosis is always guarded and may be poor. Long-haired dogs should be clipped prior to dipping. Amitraz (Mitaban) is the treatment of choice. Various treatment protocols exist with dipping schedules weekly or bi-weekly, and concentrations ranging from 0.025 to 0.125% (Folz et al. 1985, Kwochka et al. 1985, Bussieras & Chermette 1986, Scott & Walton 1985, Foley 1991). Treatment should continue 4 weeks after scrapings show no mites. If lesions have not cleared after 20 weeks, alternative therapies should be considered (Kwochka 1987). The protracted daily oral administration of milbemycin (0.5 to 1.0 mg/kg) or ivermectin (0.6 mg/kg) has proven quite effective (Yathiraj et al. 1990, Garfield & Reedy 1992, Paradis & Laperriere 1992, Ristic 1993). Treatment protocols should include antibacterial treatment for secondary pyoderma.

Cheyletiella

D Description and Biology. Cheyletiellosis is a severe dandruff-like dermatitis caused by mites in the genus *Cheyletiella*. Several species can infect dogs and cats. *Cheyletiella yasguri* Smiley is the dog fur mite, *C. blakei* Smiley is the cat fur mite, and *C. parasitivorax* (Megnin) is the rabbit fur mite that also affects cats (Kwochka 1987, Alexander & Ihrke 1982, Lewis 1989). While these mites are primarily parasites of dogs and cats, humans are often afflicted with transient (2 to 4 week) infections resulting in pruritus and erythematous macules (Thoday 1988, Merchant 1990). Most cases of cheyletiellosis are seen in young animals typically in or recently removed from kennels, catteries, and pet stores. Cheyletiellid mites will bite people but, as with *Sarcoptes* mites from pets, human infestations cease with resolution of the pet infestation (Smiley 1970).

The mites are 300-500 µm long with large protruding hook-like mouth parts (palpal claws). Legs are long and extend beyond the body. Eggs, which measure 110 x 230 µm, are attached to hairs by fibrillar strands.

Economic Importance. There are no estimates for costs of treatment of cheyletiellosis.

Methods of Control. Due to the highly contagious nature of cheyletiellosis, all in contact animals should be treated. Dogs are treated with miticidal dips containing carbaryl, lime/sulfur, or pyrethroids. Treatment protocol is 2 to 4 dips every 3 to 4 weeks. Cats are treated by dipping or shampooing with carbaryl or pyrethrum at the same treatment intervals as dogs. Although not approved, ivermectin administered at 300 µg/kg subcutaneously or orally and repeated in 2 to 3 weeks has been shown to be highly effective experimentally (Paradis & Villeneuve 1988, Paradis et al. 1990). In refractory cases the house, furniture, bedding and clothes may be contaminated and should be treated with chlorpyrifos or permethrin.

Ticks, *Dermacentor, Rhipicephalus, Amblyomma, Ixodes,* and other genera

Description and Biology. Ticks found on dogs and cats often represent a sampling of tick species present where the dogs and cats wander. Thus tick species on pets vary geographically. *Dermacentor variabilis* (Say), the American dog tick, is probably the most widespread tick species in the United States and, thus, most commonly occurs on pets in this country. Lone star ticks, *Amblyomma americanum* (L.), are serious pests of dogs throughout the Southeast as far west as Abilene, Kansas, and as far north as Omaha, Nebraska, to New Jersey. *Ixodes scapularis* Say, the blacklegged tick, commonly occurs on dogs in southeastern states and is the primary species on dogs and cats in many localities in the northeastern states. *Dermacentor andersoni* (Stiles), the Rocky Mountain wood tick, is the species most frequently found on dogs in the Rocky Mountain and Great Basin states where the previously mentioned species do not occur. *Rhipicephalus sanguineus* (Latreille), the brown dog tick, occurs in every state but is much more abundant in southern climes where it overwinters outdoors as well as indoors. This species is prevalent even on outdoor dogs south of about 38 degrees north latitude. Tick species that are primarily associated with wildlife often parasitize dogs and cats that wander without restriction or that are exercised in field and forest. For example, the rabbit tick *Haemaphysalis leporispalustris* (Packard) has been taken from cats;

Amblyomma maculatum Koch, the Gulf Coast tick, *Ixodes cookei* Packard, and *Ixodes kingi* Bishopp
have all been taken from dogs (D.E. Mock, unpublished). *Ixodes texanus* Banks was collected from a dog in Iowa (Eddy & Joyce 1942). Many additional examples can be gleaned from the literature.

R. sanguineus is a three-host tick in which all parasitic stages (larva, nymph and adult) can develop feeding on the dog. Due to its ability to withstand arid conditions and to develop on a single species of host, it frequently infests human dwellings. Female brown dog ticks commonly lay eggs in cracks and crevices of kennels or other dwellings that tick infested dogs inhabit. Brown dog ticks do not overwinter outdoors in north temperate regions. In protected climates with abundant hosts it can complete several generations in a year. While it can complete development on dogs, larvae and nymphs also feed on rodents and rabbits.

Dogs acquire *Dermacentor* ticks in fields or woods. These are ornate, 3-host ticks, whose parasitic stages will attach and feed on many hosts including humans. Larvae feed almost exclusively on small rodents. Larvae do not feed on dogs or man, but nymphs and adults will. Females lay 4,000 to 6,500 eggs, usually outdoors. Adults can live 2 years without feeding and overwinter as adults. The life cycle usually takes 2 years, but may be completed in as little as 3 months in warm climates with abundant hosts.

The principal vector of Lyme disease in the Midwest and eastern U.S. is *Ixodes scapularis*, the black-legged tick. This is a small 3-host tick, with a two year life cycle. Eggs are laid in early summer, with larvae hatching in late fall. Larvae, which are the size of a pin head, feed on small mammals (mice, squirrels, voles, shrews, raccoons). Peak nymphal numbers occur in early summer. Nymphs feed on the same hosts as larvae, in addition to cats and humans. Adults feed in the late summer or early the following spring and are typically found on white tail deer, raccoons and dogs. In the western U.S., *I.pacificus* and *I. neotomae* are vectors of Lyme disease.

The bite of some ticks may produce tick paralysis in pets, humans and other animals (Lane et al. 1984, Malik & Farrow 1991). Goethe et al. (1979) reported that "the capacity to produce paralysis has been demonstrated, described, or suspected for about 43 tick species in 10 genera." *Dermacentor andersoni* and *D. variabilis* are the species most commonly implicated in tick paralysis in North America, but several other North American species of *Dermacentor*, *Amblyomma*, and *Ixodes*, as well as *Otobius megnini* (Duges), may induce paralysis in their hosts (Goethe et al. 1979).

Tick-borne diseases in cats and dogs include Lyme disease, ehrlichiosis, Rocky Mountain spotted fever, babesiosis (Hoskins 1991) and cytauxzoonosis (Hoover et al. 1994). In cats and dogs, infection with Borrelia burgdorferi manifests as lameness, limb/joint disorders, fever, anorexia, or fatigue (Magnarelli et al. 1987, 1990). Early infections are typified by lethargy, lymphadenopathy, and the acute onset of stiffness or lameness (Greene 1991). Ticks also transmit *Ehrlichia* (McCloskey 1989), with exposure in some locations being quite common (Rodgers et al. 1989). Ehrlichial infection typically manifests as fever, edema, conjunctivitis, joint pain, and vomiting. Dogs infected with *Rickettsia rickettsii*, the causative agent of Rocky Mountain spotted fever, may suffer from fever and lethargy as a result. Canine babesiosis is a febrile disease of wild and domestic canids caused by *Babesia canis* or *B. gibsoni*. Infection causes rapid development of anemia, with high rates of mortality in untreated animals. Young canids are particularly susceptible, with transplacental infection occurring in addition to tick transmission. Cats infected with *Cytauxzoon felis* exhibit severe lethargy and generalized paresis, usually accompanied by high fever, pallor, icterus and dyspnea. In domestic cats, the infection is almost invariably fatal, typically within 48 hours following onset of symptoms (Walker & Cowell 1995).

Ticks are found less commonly on cats. Probably because of their grooming fastidiousness, cats are less likely to harbor ticks, and are thus not as likely to acquire tick-vectored diseases (Lissman 1991). Nevertheless, cats are far from being entirely exempt from parasitism by ticks and the fact that cats incur tick paralysis indicates that some ticks bite cats and remain attached for several days. In one study in a northeastern state, 22 of 93 cats had one or more ticks attached (Magnarelli et al. 1990).

Economic Importance. As most products sold for tick control are combination flea and tick products, it is difficult to separate out the value of treatments strictly for ticks. However, the medical conditions transmitted by ticks (particularly Lyme disease and ehrlichiosis) are estimated to cost $1.1 billion per year (Hinkle, unpublished data).

Methods of Control. While there are a number of efficacious acaricides registered for tick control on dogs and cats, source reduction remains the preferred means of preventing tick attacks (Garris 1991). Clearing brush and mowing grasses help to reduce populations of free-living ticks. Elimination of wildlife cover and food sources reduces introduction of ticks into the pet's territory. Restriction of pet activities to areas in which tick populations have been reduced will decrease their opportunity to acquire ticks. Pesticides such as carbaryl, chlorpyrifos, diazinon and permethrin may be used for area treatments against ticks. When brown dog ticks infest kennels, it is important to treat under cages and around false ceilings. Female ticks crawl to secluded sites to lay eggs; larvae typically crawl up, so are found around door frames and ceilings. Daily hand-searching of pets for ticks, and destruction of any found, prevents female ticks from engorging and ovipositing in or near the home and thus reduces the number of ticks there in the next generation. For a more complete discussion of off host tick control methods, refer to Current Control Methods in the section on Horses in this proceedings.

On-animal tick products include chlorpyrifos, permethrin, and fipronil. Tick collars include products such as dichlorvos and amitraz. Tick treatments for cats contain pyrethrins, carbaryl, or fipronil, formulated as sprays, dusts, shampoos or spot-on products.

Mosquitoes, *Aedes, Anopheles, Culex,* and other genera

Description and Biology. Mosquitoes are small, delicate insects, the females of which are mainly hematophagous, thus serving as potential vectors for a variety of disease organisms. All immature stages are aquatic, defining the prospective habitat of this group.

Mosquitoes serve as the intermediate host of *Dirofilaria immitis*, the dog heartworm, which is a filarial worm that infests canids and rarely cats or humans. Adult worms may reach lengths up to 31 cm and lodge in the right ventricle of the heart and the pulmonary artery where they restrict circulation, leading to a loss of exercise tolerance, chronic cardiac insufficiency, and heart failure. Common symptoms include a persistent cough that is aggravated by exercise and exaggerated tiring upon exertion. Infection rates up to 50% or higher have been recorded in enzootic areas. Geographic location of dog exposure (not just residence; remember, some dogs travel) is also a consideration. Lok's review (1988) states that *D. immitis* is enzootic in the eastern half of the U.S. and in Hawaii. There is a lower frequency of occurrence in the western continental U.S. except for an emerging focus of infection in northern California.

Dogs with an active infestation have adult worms within the heart that produce large quantities of an embryonic form of the parasite known as microfilariae. Microfilariae circulate in the bloodstream but do not grow into adult heartworms within the same host. The immature stages of *D. immitis* develop in and are transmitted by over 70 mosquito species. Microfilariae are ingested by feeding mosquitoes and escape from the midgut into the hemocoele where they develop in the malpighian tubules. Infective larvae then migrate to the head and enter the labium from which they escape while the mosquito feeds. Mature worms reach the heart in three to four months, mate, and produce microfilariae in six to eight months.

Surveys of dogs from various parts of the United States have reported from 0 to 55% of the dogs are estimated to have heartworm (Lok 1988), based on circulating microfilariae. Typically only half of the animals harboring adult worms test positive for microfilariae (Streitel et al. 1977), so incidence is probably underestimated, even among animals that are tested. On the other hand, Scoles (1994) suggested that much of the previous literature on *D. immitis* may, in fact, pertain to other species of *Dirofilaria*. While the microfilariae and occasionally adults of other *Dirofilaria* species (and of the related *Dipetalonema reconditum*) do occur in dogs, they seldom reside in the heart and are considered nonpathogenic in dogs (Lok 1988). The general distribution of *D. immitis* in wild canids indicates that autochthonous transmission is occurring throughout the country (Otto 1977, Weinmann & Garcia 1980).

The filariae of *D. immitis* may reach maturity in humans, locating in the heart and adjacent vessels. While they cannot reproduce in the human host, over 70 cases of such complications have been reported from the U.S. and are probably more widespread than suspected in hyperenzootic areas, such as the southeastern U.S., where vector mosquitoes are numerous and reservoir host populations abundant.

Economic Importance. As it is estimated that over two thirds of the animals in the United States live in heartworm enzootic areas, the cost of prophylaxis alone could amount to over $2 billion, plus annual veterinary exams to test for microfilariae. Treatment of uncomplicated adult heartworm infestation runs approximately $250 per case; extrapolated to a potentially exposed canine population of 35 million, this could yield a national price tag of almost $9 billion (Hinkle, unpublished data).

Methods of Control. Source reduction is the most effective and preferred method of mosquito control. Treatment of the host with insecticides and repellents is minimally effective. Elimination of breeding sites and suppression of immature stages eliminates development of the pestiferous blood-sucking adult stage. Elimination or modification of potential larval habitats may be accomplished by draining standing water and removal of structures that hold water. Modification includes vegetation management, manipulation of biological control organisms (predators, parasites, pathogens, and competitors), water interface disruption using oils and monomolecular layers of various substances, and chemical control, including both traditional chemicals and alternatives such as insect growth regulators and larvicides such as *Bacillus thuringiensis israelensis*.

Adult mosquito control is more problematic, being typically an area-wide effort. Adulticide sprays are temporary, palliative measures useful for local mosquito suppression. For further discussion of mosquito abatement, see the section on Horses in this volume. Currently, vector suppression is ineffectual in preventing *D. immitis* transmission and so prophylactic treatment of the canine is the standard method of prevention of dog heartworm.

Prevention of dog heartworm involves initial veterinary examination and subsequent prophylaxis extending throughout the season of potential mosquito activity. Treatment of an infected animal includes testing for microfilariae, hospitalization of the animal during treatment, medications, supportive therapy for symptoms secondary to the adulticide, and follow-up examinations.

Adult heartworms are usually destroyed by a series of injections of a nematocidal compound (thiacetarsamide sodium or melarsomine dihydrochloride), but dead parasites can cause occlusion of pulmonary vessels, leading to death or serious complications (Calvert & Rawlings 1993). Death of the host is caused by blockage of the pulmonary artery with emboli (masses of debris) resulting from dead heartworms.

Stable Fly, *Stomoxys calcitrans* (L.)

Description and Biology. The stable fly is also called the dog fly in recognition of its habit of feeding on dogs, particularly around the periphery of the pinna, producing bloody wounds that serve as sites of secondary infection. The stable fly is similar to the house fly in size and color, but possesses bayonet-like mouthparts that differentiate it from the house fly. Both sexes are vicious biters. They are strong fliers and range many miles from the breeding sites.

Larval stages are found in soggy hay, grain or feed, piles of rotting grass cuttings, and similar decomposing materials. The female, when depositing eggs, crawls into loose material, placing the eggs in crevices. Each female may lay a total of 500-600 eggs in 4 separate batches. Eggs hatch in 2 to 5 days and the newly hatched larvae burrow, begin to feed, and mature in 14 to 26 days. While the average life cycle is 28 days, this period will vary from 22 to 58 days, depending on weather conditions. Adult flies are capable of dispersing long distances (80 miles or more) with frontal systems, but tend to be worst within a mile of developmental sites.

Economic Importance. There is no estimate of the economic importance of stable flies to companion animals. Because stable flies are also a nuisance to humans in the neighborhood, their significance as pests is not limited only to the annoyance they cause to dogs.

Methods of Control. As discussed in other commodity sections, source reduction remains the most effective solution for stable flies, but because their flight range is so extensive, it must be conducted on an area-wide scale. In particular, piles of grass clippings and other backyard sources of breeding must be eliminated. Stable flies spend little time on the host, limiting the efficacy of contact insecticides applied to the animal. Residual insecticides applied to resting sites may be effective. Alsynite sticky traps can also help to reduce stable fly numbers in the area.

Lice, *Linognathus setosus* (Olfers), *Trichodectes canis* (DeGeer), *Heterodoxus spiniger* (Enderlein) and *Felicola subrostratus* (Burmeister)

Description and Biology. Lice are obligatory ectoparasitic insects. They may be classified into two separate orders Anoplura (sucking mouthparts; hang onto hair with claws; head longer than broad) and Mallophaga (biting mouthparts; hang onto hair with mandibles; head as broad as long). Lice are very host specific, usually being restricted to one genus of host. Lice found infesting dogs and cats in North America are *Linognathus setosus* (Olfers), the dog sucking louse (Anoplura); *Trichodectes canis* (DeGeer), the dog chewing louse (Mallophaga); *Heterodoxus spiniger* (Enderlein), the dog biting louse (Mallophaga); and *Felicola subrostratus* (Burmeister) the cat louse (Mallophaga) (Thoday 1981, Grant 1989, Georgi 1990).

Adult lice live on the host with females laying eggs (nits) cemented to hair. Nymphs that hatch from eggs undergo three nymphal molts before maturing to adults. Complete development occurs on the host. The developmental cycle of most lice is similar with each female louse laying 200 to 300 operculated eggs during her life, eggs taking from 1 to 3 weeks to hatch, and newly emerging nymphs developing rapidly into sexually mature adults in 1 to 3 days. In most species the egg to adult span is about 1-2 months (Georgi 1990). Transmission is usually by direct contact because lice cannot survive more than 2 to 3 days off the host.

Trichodectes canis, the dog biting louse, may produce irritation in dogs, particularly in puppies. It is a broad, short species, measuring about 1 mm in length. *Heterodoxus spiniger* is a chewing louse of coyotes and wolves that is occasionally found on dogs. Cats may become infested with the cat louse, *Felicola subrostratus*.

Louse infestations are frequently worse in the winter due to huddling of animals, stress, and increase in animals' haircoat length. Predisposing factors for louse infestations are poor nutrition, overcrowding, poor sanitation, and cold temperatures (Harwood & James 1979). Pups and old dogs are more likely to present with disease.

Chewing lice cause disease by irritation, feeding on skin scales, debris, hair and feathers, often resulting in severe debilitation. They do not serve as disease vectors, but some are intermediate hosts for helminths (the chewing louse of dogs can transmit *D. caninum*). Anoplurans feed on blood and tissue fluids causing anemia, hypoproteinemia and occasionally death.

Clinical signs are primarily due to irritation, pruritus and self trauma (scratching and chewing). Long hair becomes matted and areas of short hair develop alopecia. Animals may become weak and emaciated; if they are infested by sucking lice, anemia may develop.

Economic Importance. Louse infestations are sufficiently rare on dogs and cats that their economic significance is probably minor, totaling around $4 million per year (Hinkle, unpublished data). While an individual infestation may be costly, there are relatively few cases identified in veterinary practice.

Information on disease incidence and control expenditures for louse infestations is lacking. This is primarily due to multiple pest species labels on insecticide products. The vast majority, if not all, of products used to treat for lice are also labeled for fleas. The flea market so dominates insecticide sales that records of products sold for louse control are not maintained. In addition there is no system for tracking the number of cases seen by veterinarians.

Methods of Control. Control of lice is primarily through the use of insecticide based dips and shampoos. Compounds used to treat lice on dogs include carbaryl, pyrethrum, permethrin, and chlorpyrifos (Georgi 1990). Cats are generally treated with carbaryl or pyrethrin shampoos. Animals are treated 2 to 3 times at 2 to 3 week intervals.

There are strains of *Bacillus thuringiensis* which have been demonstrated effective against lice (Payne & Hickle 1993), but none of these have been commercially developed.

Research and Extension Needs

(1) Injury level. There are no data available on the population of ectoparasites necessary to induce disease. Research needs to be conducted on the immunologic response of the host to ectoparasites and their by-products. If the clinical signs associated with infestations are the result of a hypersensitivity, then a single individual pest may elicit disease.

(2) Identification. The true extent or severity of ectoparasitic infestations of dogs and cats is unknown. A system needs to be developed for veterinarians to track cases, with records being reported yearly. Information gathered should include number of animals infested, breeds, pest species, any predisposing factors, treatment protocol, and effectiveness of treatment. Better methods are needed of distinguishing among species of microfilariae in blood samples from dogs. A rapid, simple method that could be performed by local veterinarians would be especially useful.

(3) Sampling. One of the problems with many ectoparasitic infestations is difficulty in diagnosis. Research needs to be conducted on improving diagnostic (sampling) techniques.

(4) Biological and ecological. Basic knowledge of pest biology is lacking for several of these ectoparasites. Host specificity should be determined, along with parameters determining host-parasite adaptations. Research needs to be conducted on the impact of nutrition, pet density, and sanitation on ectoparasitic infestations. Additionally research needs to be conducted on the ability of certain hosts to withstand infestations, including investigation into host resistance (both natural and induced). The importance of fomites in transmission in commercial facilities (kennels, catteries, and pet stores) needs to be investigated.

(5) Control. More unbiased efficacy assessments need to be conducted on products marketed for ectoparasite control on companion animals.

(6) Extension. Information about applied aspects of pet ectoparasites and their management should be transferred to both veterinary practitioners and pet owners.

REFERENCES CITED

Ackerman, L. 1991. Mange in dogs and cats. Part II. Pet Focus 3: 21-23.

Alexander, M. M. & P. J. Ihrke. 1982. *Cheyletiella dermatitis* in small animal practice: a review. Calif. Vet. 3: 9-12.

Anderson, L. G. 1969. Fleas. Vet. Rec. 85(16): 449.

Anthony, J. & K. McManus. 1991. Financing Fido. Changing Times 45(3): 67-70.

Arlian, L. G. & D. L. Vyszenski-Moher. 1988. Life cycle of *Sarcoptes scabiei* var. *canis*. J. Parasitol. 74: 427-430.

Arlian, L. G., D. L. Vyszenski-Moher & M. J. Pole. 1989. Survival of adults and development stages of *Sarcoptes scabiei* var. *canis* when off the host. Exp. Appl. Acarol. 6(3): 181-187.

August, J. R. 1988. Otitis externa: a disease of multifactorial etiology. Vet. Clin. North Am. (Small Anim. Pract.) 18: 732-737.

Beard, C. B., J. F. Butler & D. W. Hall. 1990. Prevalence and biology of endosymbionts of fleas (Siphonaptera: Pulicidae) from dogs and cats in Alachua County, Florida. J. Med. Entomol. 27(6): 1050-1061.

Burgess, I. 1994. *Sarcoptes scabiei* and scabies. Adv. Parasitol. 33: 235-292.

Bussieras, J. & R. Chermette. 1986. Amitraz and canine demodicosis. J. Amer. Anim. Hosp. Assoc. 22: 779-782.

Calvert, C. A. & C. A. Rawlings. 1993. Treatment of heartworm disease in dogs. Canine Pract. 18(2): 13-28.

Campbell, W. C. 1989. Use of ivermectin in dogs and cats. In: Ivermectin and Abamectin. W.C. Campbell, ed. New York, Springer-Verlag. pp 245-259.

Chen, H. T. 1934. Reactions of *Ctenocephalides felis* to *Dipylidium caninum*. Z. ParasitenKd. 6: 603-637.

Donahue, W. A. & K. A. Stemme. 1992. Integrated pest management for flea control. Vet. Tech. 13(4): 279-295.

Dryden, M. W. 1989. Host association, on-host longevity and egg production of *Ctenocephalides felis felis.* Vet. Parasitol. 34: 117-122.

Dryden, M. W. & A. B. Broce. 1993. Development of a trap for collecting newly emerged *Ctenocephalides felis* (Siphonaptera: Pulicidae) in homes. J. Med. Entomol. 30(5): 901-906.

Dryden, M. W., J. J. Neal & G. W. Bennett. 1989. Concepts of flea control. Comp. Anim. Prac. 19(4-5): 11-20.

Dryden, M. W. & A. K. Prestwood. 1993. Successful flea control. The Compendium (Small Anim. Pract.) 15(6): 821-831.

Dryden, M. W. & M. K. Rust. 1994. The cat flea: biology, ecology and control. Vet. Parasitol. 52(1-2): 1-19.

Eddy, G. W. & C. R. Joyce. 1942. Ticks collected on the Tama (Iowa) Indian Reservation with notes on other species. Iowa State Coll. J. Sci. 16: 539-543.

Fenster, P. 1985. Designing a long term flea control program. Vet. Med. 80: 45-47.

Foley, R. H. 1991. Parasitic mites of dogs and cats. Comp. Cont. Educ. Pract. Vet. 13: 793-801.

Folz, S. D. 1983. Demodicosis (Demodex canis). Comp. Contin. Educ. Pract. Vet. 5: 116-124.

Folz, S. D., D. D. Kratzer, R. D. Conklin Jr., L. H. Nowakowski, T. J. Kakuk & D. L. Rector. 1985. Chemotherapeutic treatment of naturally acquired generalized demodicosis. Vet. Parasitol. 13: 85-93.

Franc, M. & M.C. Cadiergues. 1998. Antifeeding effect of several insecticidal formulations against *Ctenocephalides felis* on cats. Parasite 5(1): 83-86.

Fukase, T., S. Hayashi, H. Sugano, R. Shikata, S. Chinone & H. Itagaki. 1991. Ivermectin treatment of *Otodectes cynotis* infestation of dogs and cats. J. Vet. Med. Japan 44: 160-165.

Garfield, R. A. & L. M. Reedy. 1992. The use of oral milbemycin oxime (Interceptor) in the treatment of chronic generalized demodicosis. Vet. Dermatol. 3: 231-235.

Garris, G. I. 1991. Control of ticks. Vet. Clin. North Am. (Small Anim. Pract.) 21(1): 173-183.

Georgi, J. R. 1990. Parasitology for Veterinarians. 5th Ed. W.B. Saunders Co., Philadelphia.

Goethe, R., K. Kunze & H. Hoogstraal. 1979. The mechanisms of pathogenicity in tick paralysis. J. Med. Entomol. 16(5): 357-369.

Grant, D. I. 1989. Parasitic skin diseases in cats. J. Small Anim. Pract. 30: 250-262.

Greene, R. T. 1991. Canine Lyme borreliosis. Vet. Clin. North Am. (Small Anim. Pract.) 21(1): 51-64.

Halliwell, R. 1981. Hyposensitization in the treatment of fleabite hypersensitivity. Results of a doubleblind study. J. Amer. Anim. Hosp. Assoc. 17: 249-253.

Harman, D. W., R. E. Halliwell & E. C. Greiner. 1987. Flea species from dogs and cats in north-central Florida. Vet. Parasitol. 23(1-2): 135-140.

Harwood, R. F. & M. T. James. 1979. The Lice, in Entomology in Human and Animal Health. 7th Ed. Macmillan Publishing Co., Inc. New York. pp 129 - 141.

Heath, A. W., A. Arfsten, M. Yamanaka, M. W. Dryden & B. Dale. 1994. Vaccination against the cat flea *Ctenocephalides felis felis.* Parasite Immunol. 16(4): 187-191.

Henderson, G., S. A. Manweiler, W. J. Lawrence, R. J. Tempelman & L. D. Foil. 1995. The effects of *Steinernema carpocapsae* (Weiser) application to different life stages on adult emergence of the cat flea *Ctenocephalides felis* (Bouché). Vet. Dermatol. 6(3): 159-163.

Hinkle, N. C. 1992. Biological factors and larval management strategies affecting cat flea (Ctenocephalides felis felis Bouché) populations. Ph.D. dissertation, University of Florida, Gainesvillle. 190 pp.

Hinkle, N. C. 1995. Natural born killers. Pest Control Tech. 23(7): 54, 56, 116.

Hinkle, N. C., P. G. Koehler & R. S. Patterson. 1995a. Residual effectiveness of insect growth regulators applied to carpet for control of cat flea (Siphonaptera: Pulicidae) larvae. J. Econ. Entomol. 88(4): 903-906.

Hinkle, N. C., P. G. Koehler & R. S. Patterson. 1995b. Larvicidal effects of boric acid and disodium octaborate tetrahydrate to cat fleas (Siphonaptera: Pulicidae). J. Med. Entomol. 32(4): 424-427.

Hinkle, N. C., M. K. Rust & D. A. Reierson. 1997. Biorational approaches to flea (Siphonaptera: Pulicidae) suppression: present and future. J. Agric. Entomol. 14(3): 309-321.

Hoover, J. P., D. B. Walker & J. D. Hedges. 1994. Cytauxzoonosis in cats: eight cases (1985-1992). J. Am. Vet. Med. Assoc. 205(3): 455-460.

Hoskins, J. D. 1991. Canine haemobartonellosis, canine hepatozoonosis, and felinecytauxzoonosis. Vet. Clin. North Am. (Small Anim. Pract.) 21(1): 129-140.

Jaegerman, M. 1992. The cost of that dog in the window. The New York Times 141: 48.

Jeneskog, T. & K. Falk. 1990. The effect of local ivermectin treatment on ear mite infestation in a cat breeding colony. Scand. J. Lab. An. Sci. 17: 17-19.

Kern, W. H. Jr. 1993. The autecology of the cat flea (*Ctenocephalides felis felis* Bouché) and the synecology of the cat flea and its domestic host (*Felis catus*). Ph.D. dissertation, University of Florida, Gainesvillle. 183 pp.

Kershaw, A. 1989. *Sarcoptes scabiei* infestation in a cat. Vet. Rec. 124(20): 537-538.

Knapp, F. W. 1978. Paramite dip for control of sarcoptic mange on dogs. Vet. Med. Small Anim. Clin. 73: 1161-1163.

Koehler, J. E., C. A. Glaser & J. W. Tappero. 1994. *Rochalimaea henselae* infection, a new zoonosis with the domestic cat as reservoir. J. Am. Vet. Med. Assoc. 271(7): 531-535.

Kunkle, G. A. & J. Milcarsky. 1985. Double-blind hyposensitization trial in cats. J. Am. Vet. Med. Assoc. 186: 677-680.

Kwochka, K. W. 1987. Mites and related disease. Vet. Clin. North Am. (Small Anim. Pract.) 17: 1263-1284.

Kwochka, K. W, G. A. Kunkle & C. O'Neill Foil. 1985. The efficacy of amitraz for generalized demodicosis in dogs: a study of two concentrations and frequencies of application. Contin. Educ. 1985: 8-17.

Lane, R. S., J. Peek & P. J. Donaghey. 1984. Tick (Acari: Ixodidae) paralysis in dogs from northern California: acarological and clinical findings. J. Med. Entomol. 21(3): 321-326.

Lewis, T. L., II. 1989. Mites of importance in small animal practice. Vet. Focus 1: 33 37.

Lissman, B. A. 1991. Lyme disease in small animals: identification and clinical description, pp. 50-52 In Proc. 1st Symposium on Ectoparasites of Pets, June 17 19, 1991. Edited by F.W. Knapp. Univ. Kentucky, Lexington.

Lok, J. B. 1988. Dirofilaria sp.: Taxonomy and distribution, in P.F.L. Boreham & R.B. Atwell (eds), Dirofilariasis. CRC Press, Boca Raton, Florida. pp. 2-28

Maciejewska, J., W. F. Chamberlain & K. B. Temeyer. 1988. Toxic and morphological effects of *Bacillus thuringiensis* preparations of larval stages of the Oriental rat flea (Siphonaptera: Pulicidae). J. Econ. Entomol. 81(6): 1656-1661.

Magnarelli, L. A., J. F. Anderson, H. R. Levine & S. A. Levy. 1990. Tick parasitism and antibodies to *Borrelia burgdorferi* in cats. J. Am. Vet. Med. Assoc. 197(1): 63-66.

Magnarelli, L. A., J. F. Anderson, A. B. Schreier & C.M. Ficke. 1987. Clinical and serologic studies of canine borreliosis. J. Am. Vet. Med. Assoc. 191(9): 1089-1094.

Malik, R. & B. R. Farrow. 1991. Tick paralysis in North America and Australia. Vet. Clin. North Am. (Small Anim. Pract.) 21(1): 157-171.

Manweiler, S. A. 1994. Development of the first cat flea biological control product employing the entomopathogenic nematode Steinernema carpocapsae. Proceedings of the Brighton Crop Protection Conference. pp. 1005-1012.

McCloskey, R. V. 1989. Ehrlichiosis--"spotless spotted fever". Del. Med. J. 61(7): 335-337.

Merchant, S. R. 1990. Zoonotic diseases with cutaneous manifestations - Part I. The Compendium 12: 371-378.

Miller, W. H. 1980. Canine demodicosis. Comp. Cont. Educ. 2: 334-344.

Moriello, K. A. 1991. Parasitic hypersensitivity. Semin. Vet. Med./Surg. 6: 286-289.

Olsen, A. 1987. Fleas. Danish Pest Infest. Lab. Ann. Report. pp. 74-79.

Otto, G. F. 1977. Proceedings of the heartworm symposium '77. Veterinary Medicine Publishing Co., Bonner Springs, KS, 189 pp.

Paradis, M. & E. Laperriere. 1992. Efficacy of daily ivermectin treatment in a dog with amitraz-resistant generalized demodicosis. Vet. Dermatol. 3: 85-88.

Paradis, M. & A. Villeneuve. 1988. Efficacy of ivermectin against *Cheyletiella yasguri* infestation in dogs. Can. Vet. J. 29: 633-635.

Paradis, M., D. Scott & A. Villeneuve. 1990. Efficacy of ivermectin against *Cheyletiella blakei* infestation in cats. J. Amer. Anim. Hosp. Assoc. 26: 125-128.

Parish, L. C. & R. M. Schwartzman. 1993. Zoonoses of dermatological interest. Semin. Dermatol. 12(1): 57-64.

Payne, J. M. & L. A. Hickle. 1993. *Bacillus thuringiensis* isolates active against Phthiraptera pests. United States Patent No. 5,273,746.

Ristic, Z. 1993. Ivermectin in the treatment of generalized demodicosis in the dog. Vet. Dermatol. 4: 40-41.

Rodgers, S. J., R. J. Morton & C. A. Baldwin. 1989. A serological survey of *Ehrlichia canis, Ehrlichia equi, Rickettsia rickettsii,* and *Borrelia burgdorferi* in dogs in Oklahoma. J. Vet. Diagn. Invest. 1(2): 154-159.

Scheidt, V. J. 1987. Common feline ectoparasites. Part 2: *Notoedres cati, Demodex cati, Cheyletiella* spp. and *Otodectes cynotis*. Feline Pract. 17: 13-23.

Scheidt, V. J. 1988. Flea allergy dermatitis. Vet. Clin. North Am. (Small Anim. Pract.) 18(5): 1023-1042.

Scheidt, V. J., L. Medleau, R. L. Seward & R. M. Schwartzman. 1984. An evaluation of ivermectin in the treatment of sarcoptic mange in dogs. Am. J. Vet. Res. 45: 1201 1202.

Schneck, G. 1988. Use of ivermectin against ear mites in cats. Vet. Rec. 123(23): 599.

Scoles, G. 1994. Surveying for vectors of dog heartworm. Vector Control Bull. North Central States 3(1): 59-67.

Scott, D. W., B.R.H. Farrow & R. D. Schultz. 1974. Studies on the therapeutic and immunologic aspects of generalized demodectic mange in the dog. J. Amer. Anim. Hosp. Assoc. 10: 233-244.

Scott, D. W., R. D. Schultz & E. Baker. 1976. Further studies on the therapeutic and immunologic aspects of generalized demodectic mange in the dog. J. Amer. Anim. Hosp. Assoc. 12: 203-213.

Scott, D. W. & D. K. Walton. 1985. Experiences with the use of amitraz and ivermectin for the treatment of generalized demodicosis in dogs. J. Amer. Anim. Hosp. Assoc. 21: 535-541.

Silverman, J., M. K. Rust & D. A. Reierson. 1981. Influence of temperature and humidity on survival and development of the cat flea, *Ctenocephalides felis* (Siphonaptera: Pulicidae). J. Med. Entomol. 18: 78-83.

Slacek, B. & J. P. Opdebeeck. 1993. Reactivity of dogs and cats to feeding fleas and to flea antigens injected intradermally. Aust. Vet. J. 70(8): 313-314.

Song, M. D. 1992. Using ivermectin to treat feline dermatoses caused by external parasites. Vet. Med. 86: 500-502.

Streitel, R. H, P. C. Stromberg & J. P. Dubey. 1977. Prevalence of *Dirofilaria immitis* infection in dogs from a humane shelter in Ohio. J. Am. Vet. Med. Assoc. 170: 720.

Thoday, K. 1981. Skin Diseases of the Cat. In Practice Nov.: 22-34.

Thoday, K. L. 1988. Skin diseases of dogs and cats transmissible to man. In: Small Animal Clinic, p. 5-13.

Walker, D. B. & R. L. Cowell. 1995. Survival of a domestic cat with naturally acquired cytauxzoonosis. J. Am. Vet. Med. Assoc. 206(9): 1363-1365.

Webster, S. B. & D. M. Uhler. 1986. Common skin conditions in humans and animals. Am. Fam. Phys. 33(4): 177-182.

Weinmann, C. J. & R. Garcia. 1980. Coyotes and canine heartworm in California. J. Wild. Dis. 16(2): 217-221.

Whiteley, H. E. 1987. Five flea-control programs for cats. Vet. Med. 10: 1022-1026.

Yaphe, W., S. Giovengo & N. S. Moise. 1993. Severe cardiomegaly secondary to anemia in a kitten. J. Am. Vet. Med. Assoc. 202(6): 961-964.

Yathiraj, S., P. M. Rao, N.R.J. Reddy & M. T. Raj. 1991. Treatment of scabies in canines with amitraz. Indian Vet. J. 67: 867-868.

****

End of Dogs and Cats section

Top	Poultry	Dairy Cattle	Range Beef Cattle	Confined Beef Cattle
Swine	Sheep & Goats	Horses	Dogs & Cats	Bottom

****

...