THE EVALUATION OF POULTRY PEST MANAGEMENT TECHNIQUES IN FLORIDA POULTRY HOUSES

BY

JEROME ADKINS HOGSETTE, JR.

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By

Jerome Adkins Hogsette, Jr.

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Chairman: Dr. J. F. Butler
Major Department: Entomology and Nematology

The house fly, *Musca domestica* (L.), and the northern fowl mite, *Ornithonyssus sylviarum* (C.& F.), are the two major arthropod pests associated with the poultry industry in Florida. Presented is a system whereby various control techniques for these pests have been evaluated. Techniques were divided into two main areas: house fly control, based mainly on manure management for control of immature fly populations, and northern fowl mite control, based on evaluation of acaricides for control of mite populations on chickens.

Rotovation, a method of tilling and aerating manure for house fly control, was evaluated as a technique for drying and composting manure *in situ*. Drying was enhanced by tilling wood chips and an insect growth regulator (CGA 72662™) into wet manure areas. When CGA 72662 was applied topically, fly larval control was seen for 35 days with a single application. Commercially labeled organophosphorus larvicides lasted only 2 weeks. LC_{50} bioassay of CGA 72662 for house fly larvae was 0.45 ppm.

Methoprene and dimilin were evaluated as oral larvicides, but gave poor field results. When 20 ppm of CGA 72662 were added to the

drinking water of hens, bioassayed manure produced 100% fly mortality. Methoprene sand granules topically applied to manure gave house fly larval control of greater than 90% for 3 days post-treatment in bio-assayed manure samples.

Ophyra aenescens (Wied.), the black dump fly, was reared in the laboratory and the larvae were proven to be predators of house fly larvae. In the field, *O. aenescens* adults were considered pestiferous, and their use as a biocontrol agent is not recommended at this time.

In laboratory competition studies, *Hermetia illucens* (L.), an assumed biocontrol agent, did not prevent house fly larvae from reaching maturity when the two species were reared in the same container. In the field, a situation occurred where larvae of *H. illucens* and *M. domestica* were living in the same manure pack, but in different strata.

Light traps, baits, and residual sprays were evaluated for their ability to effectively reduce adult house fly populations. Light traps were generally ineffective, but baits gave good results. In laboratory studies, a bait consisting of dichlorvos and ronnel had the fastest knockdown, killing all flies in 10 minutes. A methomyl bait had the longest residual and was killing at a rate higher than 25% after a 6-week testing period. A Bomyl bait with Lure'em II™ attractant killed a significantly greater number of flies than all baits tested. A permethrin bait was unattractive to flies in the field even though its fly killing ability was demonstrated in the laboratory. When ICI 143, BW 21Z, and SD 43775 synthetic pyrethroid compounds were applied as a residual treatment to wooden panels hung in poultry houses, all compounds produced 100% fly mortality 121 days post-treatment.

In laboratory bioassay, permethrin LD_{50} for house fly adults was 18.0 ppm.

Northern fowl mite acaricide bioassays gave LC_{50}'s for carbaryl, malathion, and permethrin of 0.41, 1.70, and 2.9 ppm respectively. When carbaryl, malathion, and Ravap™ were applied to hens at rates of 0.30, 0.44, and 0.36% respectively, northern fowl mite control approaching 100% was achieved with Ravap in 2 weeks. Adequate control was achieved after 4 to 6 weeks with reapplication of carbaryl and malathion. Two synthetic pyrethroids, BW 21Z and SD 43775, applied once to floor birds at rates of 0.05% and 0.10% respectively, gave 100% control of mites for 7 weeks. In production trials when 12 strains of hens were evaluated for the effects of northern fowl mites on egg production, no overall difference in egg production could be found due to mite control. However, one strain of hens showed a significant increase in egg production of 3.67% due to mite control with Ravap.

The use of the above techniques, individually or in combinations, will enable the poultry operator to more efficiently regulate house fly populations in and around poultry houses. These techniques will also enable him to effectively control northern fowl mites on hens and determine whether or not mite populations are affecting hen performance.

INTRODUCTION

In many areas of Florida, poultry farms are located in close proximity to large housing developments. House flies (*Musca domestica*, L.) must be effectively controlled for more than just economic reasons, since poultrymen are quickly blamed when flies are found in or around nearby homes. Under Florida law, Chapter 386, which regulates excessive fly breeding and odors, complaints by three responsible citizens can result in sanitary inspections of suspect poultry farms by Health and Rehabilitative Services (HRS). Inspected farms not meeting HRS standards for sanitation and fly control can eventually be closed if farm owners fail to rectify discrepancies to the satisfaction of HRS and the surrounding neighbors. Although not a problem for homeowners, northern fowl mites, *Ornithonyssus sylviarum* (C.& F.), can be annoying to farm laborers and egg processors. Mites have also been known to cause decreased egg production and increased mortality in poultry flocks.

Although many compounds are available for fly and mite control, a large number have been rendered ineffective due to resistance problems. Slowness and indecision by the EPA have prevented the labeling of new compounds and put the labels of approved compounds in jeopardy.

In this dissertation house fly control was evaluated by rototilling, an *in situ* method for drying poultry manure. Stabilizers were added to wet manure to enhance drying. Insect growth regulators (IGR's) were tested in poultry feed and water as oral larvicides. IGR's were also evaluated when applied topically to poultry manure.

Ophyra aenescens (Weid.) was tested as a biocontrol agent against house flies. Laboratory and anatomical evidence is presented to show that larvae of *O. aenescens* are predaceous. The ability of *Hermetia illucens* (L.) to preclude larvae of other flies from its growth media was investigated and results corroborated by field observations.

Light traps, granular baits and contact residual insecticides were evaluated for adult house fly control.

The efficacies of labeled and unlabeled acaricides were evaluated for northern fowl mite control. The effects of northern fowl mites on egg production were evaluated on 12 strains of laying hens.

LITERATURE REVIEW

The House Fly

History and Economic Importance

The house fly, *Musca domestica* (L.), is a major economic pest of livestock and poultry. In 1977 the poultry industry in Florida lost an estimated $5.6 million due to flies (Butler, 1979). The mere presence of house flies in great numbers indicates the need for improved sanitation measures (Scudder, 1949) and may trigger legal action.

From early Biblical times, when swarms of flies ravaged Egypt, through ancient history (Cloudsley-Thompson, 1976), and up to the present, flies have been noted as pests (Greenberg, 1973). Flies fulfill all the conditions required of a disease vector (Greenberg, 1971), and have been rated second only to man as the most important animal in the transfer of human disease (Scudder, 1949). A single fly may carry more than 1 million bacterial cells. Any particular fly may be contaminated with more than 100 species of pathogenic organisms capable of causing such diseases as dysentary, typhoid fever, cholera, salmonellosis, anthrax, poliomyelitis, and hepatitis. Flies may also be contaminated with eggs of nematodes and cestodes (James and Harwood, 1969). Books by Greenberg (1971, 1973), Hewitt (1914), Lindsay (1956), and West (1951) should be consulted for in-depth details of fly-borne diseases.

What makes the house fly so important in these disease transmission cycles is its close coexistance with man, its consumption of both contaminated and uncontaminated food, its great flight activity and

dispersal, and its constant alternation between feces and food (Greenberg, 1971). Besides the transmission of diseases and helminths, house flies also cause myiasis. Cuticular, ocular, and urinary myiasis seem to be the types most frequently reported (James, 1947), but other types are recorded in the literature (Leclercq, 1969b).

Bionomics

Distribution

The house fly (*Musca domestica*) was described by Linnaeus in 1758, and is known as an ubiquitous insect (West, 1951). Hewitt (1914) called the house fly a qualified ubiquitous insect because *M. domestica* is divided into subspecies groupings in some geographic areas of the globe. These subspecies are listed in Stone et al. (1965).

Life cycle

The house fly life cycle varies in length depending on the reference. Bishopp et al. (1915) stated that the entire cycle required from 7 days to 7 weeks. Other estimates are 3 weeks per generation (James, 1947), 10 days under usual conditions but 7 days in warm weather (James and Harwood, 1969), and less than 1 week in the tropics (Oldroyd, 1965). The following times are given for the length of the developmental stages by Bishopp et al. (1915) and James and Harwood (1969) respectively:

Eggs hatch in:	24 hours	10 to 12 hours
Larval stages:	3 days to 3 weeks	5 days
Pupal stage:	3 to 26 days	4 to 5 days
Adult life span:	---	30 to 60 days

Some differences in the reported lengths of the life cycle can be attributed to varience in environmental factors, such as temperature. Melvin (1934) studied the duration of incubation periods when house fly eggs were incubated at different temperatures. Incubation periods ranged from 51.45 hours at 15.0 C to 8.05 hours at 40.6 C. At 42.8 C, no eggs hatched.

In fresh poultry manure, a temperature of 27.0 C and a moisture level of 60 to 75% proved optimal for larval development (Miller et al., 1974). In horse manure, larvae showed no ill effects when the temperature reached 45.0 C, but as the temperature approached 48.9 C, they began to migrate out. At 54.4 C, larvae died within 1 min, and at 60.0 C, death was instantaneous (Allnut, 1926).

The pH of the larval medium may also change cycle length. Erofeeva (1967) determined the optimum pH for house fly larval media to be between 7 and 8. This is also the pH of day old poultry manure (Beard and Sands, 1973).

Another temperature dependent variable that changes the length of the life cycle is the fly's ability to overwinter. Early investigators were unable to determine in which stage *M. domestica* overwintered (Hewitt, 1914; Graham-Smith, 1916), but it is now known that the house fly can overwinter in all of its developmental stages (Greenberg, 1971). Breeding occurs throughout the year in warmer climates where temperatures are 18.0 C or above (James, 1947; Greenberg, 1971).

Fecundity

At 5 to 7 days of age, the female house fly has mated and is ready to begin laying eggs (James and Harwood, 1969). A female may lay up to

1000 eggs (LaBrecque et al., 1972), and produce 5 to 20 or more batches of eggs with each batch containing 120 to 150 eggs (James, 1947). Population increases of up to six-fold can rapidly occur in field populations of house flies if conditions are right (LaBrecque et al., 1972). House flies oviposit all day without regard to time, with no more eggs laid in the morning than in the afternoon (Meyer et al., 1978).

Larval habitats

Larvae of *M. domestica* will develop in any decaying and fermenting organic material (James, 1947), in kitchen refuse and decaying vegetables (Bishopp et al., 1915), and in manure of all types (Bishopp et al., 1915; James, 1947), especially horse manure (Hewett, 1914, James and Harwood, 1969).

House fly larvae developing in media such as corpses or garbage cannot be killed by burying the media less than 1.2 m deep. Larvae will climb to within 30.5 cm of the soil surface, pupate and about 90% will survive (Mellor, 1919). Larvae that develop in microbial contaminated media may produce adults free from contamination (Greenberg, 1973).

As in other families of diptera, males of *M. domestica* emerge from their puparia before the females (Mellor, 1919).

Adult dispersion patterns

In most experiments designed to study house fly dispersal patterns, marked flies were released and recaptured at different intervals from the release point. Results indicate that flies can disperse from 8.3 (Quarterman et al., 1954) to 20.0 km (Bishopp and Laake, 1921: Lindquist et al., 1951). Greenberg (1973) reported a dispersion of 2.3 to 11.8 km within 24 hr.

Fly movement is apparently random (Schoof and Siverly, 1954a), especially when winds are variable (Pickens et al., 1967). Flies tended to disperse upwind when a steady 3.3 to 11.7 kph wind was blowing and dispersal rate increased when temperatures were 11.7 C or above (Pickens et al., 1967). A house fly apparently spends most of its life going from site to site (Schoof and Siverly, 1954a). In the study by Pickens et al. (1967), flies traveled 0.8 km past a clean farm to reach a dirty farm.

Nocturnal resting places of adults

Since the advent of contact residual pesticides, detecting and then treating house fly nocturnal resting places had been advocated as a method of control (Scudder, 1949). Kilpatrick and Quarterman (1952) found that flies congregate at dusk in large numbers and stay in the same place all night. In hot weather they rest outside on vegetation, but in cooler weather they rest inside structures (Oldroyd, 1965). Nocturnal resting sites of house flies are usually within 6.1 m of a favored daytime feeding and breeding area and are usually above the ground, but rarely higher than 4.6 m (Scudder, 1949). Anderson and Poorbaugh (1964a) found that on a test poultry farm 85% of the house fly population rested inside the poultry houses at night.

Methods for Larval Control in Poultry Houses

Manure management in dry systems

An average size hen produces from 90.9 g (Hart, 1963) to 168.2 g of wet manure daily (Winter and Funk, 1941). Fresh manure is approximately 70% moisture (Card and Nesheim, 1975; Hart, 1963) and has a pH of about 6 that rises to between 7 and 8 after about 12 hr due to

bacterial action (Beard and Sands, 1973). Since house fly larvae prefer a moisture level between 60 and 75% (Miller et al., 1974) and a pH of between 7 and 8 (Erofeeva, 1967), keeping manure dry and thereby stabilizing the manure habitat is one of the most important goals in fly control (Hartman, 1953; Legner et al., 1975; Wilson and Card, 1956). If moisture levels in manure exceed 80%, manure becomes anaerobic (Miller et al., 1974), rendering it unsuitable for house fly development (Beard and Sands, 1973).

Some authors have advocated frequent manure removal, i.e. every 5 days, to achieve good fly control (Wilson and Card, 1956). Others found that monthly or bi-weekly manure removal favored fly populations (Peck and Anderson, 1970). Abstention from manure removal has allowed populations of predators of dipteran larvae to increase (Peck and Anderson, 1969; Peck, 1969). Axtell (1970) attained good fly control by removing manure early in the fly season and then using residual sprays to keep adults in check. Loomis et al. (1975) recommended infrequent manure removal where drying was enhanced by frequent mechanical stirring. Although mechanical stirring does not succeed in drying manure in all situations (McKeen and Rooney, 1976), mechanical stirring, or rotovation, has proven to be a successful method for controlling flies on poultry farms in the Tampa Bay area of Florida (Hinton, 1977). If manure must be removed from poultry houses, a dry base 12.7 to 15.2 cm deep should be left to help dry out fresh droppings (Hartman, 1953), and re-establish house fly predators (Peck and Anderson, 1970).

Other management practices that a poultry farm operator can use to implement a manure management program are to prevent water from reaching

the manure, increase the drying surface of the manure, improve the amount and speed of airflow over the manure, and reduce the amount of fresh manure per m^2 of floor space (Hartman, 1953).

One of the benefits of keeping manure dry is that it retains its value as a fertilizer (Hinton, 1977; Loomis et al., 1975). One of the drawbacks of using poultry manure as a fertilizer, however, is that manure can increase soil salinity when it is applied in high levels (Shortall and Liebhardt, 1975). This is not a problem on acid eastern soils.

Hammond (1942) may have been the first to formulate diets for growing chickens using cow manure as a vitamin supplement. Since then, many types of manures have been tested as feed supplements. Poultry manure, which has an average nitrogen, phosphorus, and potassium analysis of 4, 3, and 2%, respectively (Woods, 1975), has been tested in poultry diets (Lee and Blair, 1973; Lee et al., 1976) and advocated for use by some authors (Woods, 1975).

For other options in manure management, consult the book on agricultural waste treatment by Hobson and Robertson (1977).

House fly biocontrol agents

The predators and parasites of immature house flies are many. Since few were dealt with in this study, only a selected review is deemed necessary. Additional references pertaining to house fly predators and parasites can be found in the house fly bibliography of West and Peters (1973). Books by Askew (1971), Clausen (1940), and Thompson (1943) are also recommended.

Spalangia endius (Walker)

Due to the individual attention that has been given this pupal parasite, and since it contaminated several of our fly colonies, a brief review is warranted.

Spalangia endius, Walker (Hymenoptera: Pteromalidae) was found to be not only a fairly common pupal parasite of house flies in poultry manure, but one which could outcompete other species of microhymenopterans (Ables and Shepard, 1974; Legner, 1967; Legner and Brydon, 1966). It is noted for its ability to rapidly find hosts (Ables and Shepard, 1974) and parasitize more hosts per unit time than its competitors (Legner, 1967; Legner and Brydon, 1966). Best results were achieved with *S. endius* during hot, dry weather (Olton and Legner, 1975). Besides the pupae of *M. domestica, S. endius* also parasitizes pupae of *Fannia femoralis* and *Ophyra leucostoma* (Legner and Brydon, 1966).

Morgan et al. (1975) suppressed a population of house flies in 35 days on a north Florida poultry farm using continuous releases of *S. endius*, and Weidhaas et al. (1977) designed a model to simulate the parasite-fly system. Thornberry and Cole (1978) found *S. endius* to be effective only on isolated farms with dry manure. Morgan et al. (1976) performed a laboratory study of the host-parasite relationships of *S. endius* and *M. domestica* and then devised a method for mass rearing the pupal parasite in the laboratory (Morgan et al., 1978).

Ophyra aenescens (Wied.)

Ophyra aenescens (Wied.) is a shiny black muscid fly easily distinguished from other members of the genus by its rufous-yellow palpi. It was described in 1830 by Wiedemann, who placed it in the genus

Anthomyia; in 1897, Stein transferred it to *Ophyra* Robineau-Desvoidy 1830 (Johnson and Venard, 1957). Several subsequent descriptions of the genus have been published, all placing *Ophyra* in the family Anthomyiidae (Malloch, 1923; Seguy, 1923; Aldrich, 1928; Graham-Smith, 1916; and Bryan, 1934). After studying the male terminalia, Crampton (1944) decided that *Ophyra* was a typical muscid, and placed it in that family, where it remains. Sabrosky, in 1949, described the genus in the Pacific region.

Distribution. *Ophyra aenescens* occurs in the United States from Oregon to Arizona, and from Illinois to the East Coast and Florida (Greenberg, 1971). It is also found in the Neotropics, the Galapagos Islands, Hawaii, Nauru, the Ocean Islands, and possibly in Bermuda (Stone et al., 1965).

Biology and rearing. The biology and morphology of *Ophyra aenescens* were described by Johnson and Venard in 1957. They used a larval medium consisting mainly of C.S.M.A.™ standard preparation. Initially, adults were maintained on cane sugar dissolved in water, but no fertile eggs were produced until a source of animal protein was provided. Fish meal was used dry as a protein source and moistened as a site for oviposition. Eggs hatched in 12 to 16 hours at 28° C. The development periods for the three larval instars and the pupal stage averaged 9 and 4 days, respectively. The complete cycle required a minimum of 14 days at 27° C ± 1. Males lived an average of 15 days and females lived an average of 20 days.

Roddy (1955) used a bacto-agar medium for rearing larvae. Preparation was time consuming and laborious compared to the C.S.M.A. medium of Johnson and Venard (1957).

Predaceous nature. Hobby (1934) suggested that adults of *Ophyra* might be predaceous. He noted them apparently feeding on dead insects, but did not see them actually capture prey.

Séguy (1923) stated that the larvae of *Ophyra* are predaceous. This was supported by Keilin and Tate (1930) who described the larvae of *O. leucostoma* as having buccopharyngeal armature characteristic of larvae that are both saprophagous and carnivorous. Later experiments proved that *O. leucostoma* was predaceous (Peck and Anderson, 1969; Peck, 1969), but not cannibalistic (Anderson and Poorbaugh, 1964b).

Relationship with *Musca domestica* in poultry manure. *Ophyra leucostoma* (Wied.) occurs commonly in poultry houses in many parts of the world (Peck and Anderson, 1970; Legner and Olton, 1968; Fujito et al., 1966). *Ophyra capensis* (Wied.) has been reported from poultry houses in Britain (Conway, 1970 and 1973), and *O. aenescens* from poultry houses in Florida (P. G. Koehler, *personal communication*).

Hermetia illucens (L.)

Description. *Hermetia illucens*, the black soldier fly (Sutherland, 1978), is a rather large hemisynanthiopic stratiomyid fly that is easily recognized (Greenberg, 1971). The genus *Hermetia* can be distinguished from all other North American genera of Stratiomyidae by the length of the style of the flagellum, which is as long as or longer than the remaining segments of the flagellum (James, 1935).

Linneaus described *H. illucens* in 1738 (May, 1961). Malloch (1917), Ricardo (1929), and Borgmeier (1930) described the immatures and pupae. James (1935), Linder (1938), and Iide and Mileti (1976) described the adults, with Linder's description being the most detailed. May (1961) described both adults and immatures.

Bionomics of *Hermetia*. The eggs of *H. illucens* take between 5 and 14 days to hatch at room temperature (May, 1961). They are laid singly to form masses of 500 to 1000 eggs (Furman et al., 1959). As many as 1062 may be laid by one female (May, 1961).

Larvae have been reared by placing the eggs in either moistened C.S.M.A. standard larval fly medium (Furman et al., 1959; Tingle et al., 1975), or in a medium consisting of dried milk, yeast, water, and paper tissue (May, 1961).

Larval development at 27 to 28 C required a minimum of 31 days (May, 1961). There are six larval instars as determined by measurement of molted head capsules. The first four instars have a creamy appearance, but a day or two after molting occurs, the cuticle of the fifth-instar larvae becomes shagreened and darkens to greyish yellow. The cuticle darkens even more after the molt to the sixth instar.

Before pupation, the larvae arrange themselves in a vertical manner in the medium with the head protruding above the surface and the two posterior segments curved ventrally (May, 1961). Furman et al. (1959) reported a pupation period of about 2 weeks at 21 to 28 C, but several pupae eclosed after 2 to 5 months. The cycle from egg to adult required 38 days at about 29.3 C in greenhouse conditions (Tingle et al., 1975).

Furman et al. (1959) demonstrated that the larvae of *H. illucens* are not paedogenic. Larvae fed on dead larvae and adults, but were not predaceous or cannibalistic.

Both Furman et al. (1959) and Tingle et al. (1975) found adults of *H. illucens* to be eurygamous. The adults reared by the former authors did not mate, but the females laid masses of sterile eggs. Tingle et al.

(1975) succeeded in getting *H. illucens* to mate by placing adults in large (76 x 114 x 137 cm) cages directly in the sun. Few matings occurred during cloudy weather or when the insects were shaded. Mating commenced during flight as stated by Copello (1926).

Due to the variable length of the larval and pupal stages, there are probably no more than two generations of *H. illucens* produced in a year (Copello, 1926), with overwintering occurring in the larval stage (May, 1961). Greenberg (1971) states that the adults readily enter houses while Furman et al. (1959) claim they do not.

Distribution. *Hermetia illucens* is rather widely distributed throughout the Western hemisphere, the Australian region from Samoa to Hawaii, and in some areas of the Palearctic region (Greenberg, 1971). Various authors report the presence of *H. illucens* in the Eastern hemisphere (Barbier, 1952; Peris, 1962; Adisoemarto, 1975). James (1935) states that *H. illucens* has been spread by commerce. Van Dyke (1939) believes *H. illucens* is a European species, but Leclercq (1966, 1969a) claims it is an American species transported to Europe and Asia.

Larval habitats. Immature stages of *H. illucens* are found in a variety of habitats. Copello (1926) found them living in beehives in Argentina where the larvae were destroying the weaker hives. Van Dyke (1939) found larvae of *H. illucens* in honey bees' nests in the U.S. Larvae have also been reported from nexts of Melponidae, a family of stingless bees (Borgmeier, 1930), from dead crabs (Ricardo, 1929), and from a human cadaver (Dunn, 1916). Other habitats include beeswax, catsup, decaying vegetables, potatoes (Malloch, 1917), and outdoor privies in the Southern U.S. (James, 1947).

Myiasis. Larvae of *H. illucens* may cause myiasis in man, particularly intestinal myiasis due to accidental ingestion of eggs or larvae (James, 1947; Greene, 1952; Werner, 1956).

Predators and parasites. Only one predator of *H. illucens* is noted in the literature. Bodkin (1917) found specimens of *H. illucens* in the nests of Bembecid wasps in British Guiana.

Wasps in the family Diapriidae are the only ones known to parasitize pupae of *H. illucens*. One species of Diapriid was found by Costa Lima and Guitton in 1962, and another, *Trichopria* n. sp., by Mitchell et al. in 1974. The latter parasite was reared (Tingle et al., 1975) and had an average life cycle of 26 days at 26.8 C. An average of 86 parasites emerged from each parasitized pupa. Twenty-three per cent of the field-collected pupae of *H. illucens* were parasitized (Tingle et al., 1975).

Relationship with *Musca domestica* (L.) in privies. The presence of larvae of *H. illucens* and *M. domestica* in privies is well documented in the literature (Howard, 1900; Hewitt, 1914; Parker, 1918; James, 1947; Quarterman et al., 1949; Schoof and Siverly, 1954b; Kilpatrick and Bogue, 1956). Further studies of the fly-breeding conditions in privies revealed an apparent antagonistic relationship between the larvae of these two species. When extremely high numbers of *H. illucens* larvae were found in privies, no larvae of *M. domestica* were present (Fletcher et al., 1956). Hypothesizing that the larvae of *H. illucens* may interfere with the development of *M. domestica*, a laboratory test was performed where various numbers of larvae of both species were grown together and separately in C.S.M.A. standard larval media.

Musca domestica adults emerged in approximately the same numbers from all jars and it was concluded that no antagonistic relationship existed (Fletcher et al., 1956).

Kilpatrick and Schoof (1959) noted that larvae of *M. domestica* were absent from privies where excretia was semiliquid and infestations of *H. illucens* were heavy. Attempts to dry the excretia with sawdust or by water manipulation caused excretia to crust over and resulted in an increase of house fly breeding and a decrease in soldier fly breeding.

Relationship with *Musca domestica* (L.) in poultry manure. The presence of larvae of *M. domestica* and *H. illucens* in poultry manure is also well documented in the literature (Cunningham et al., 1955; Tingle et al., 1975). The latter authors found them in Florida and claimed that the house fly population at one farm was being controlled by the soldier fly population. Few details were given to support that claim.

The hypothesis that larvae of *H. illucens* and *M. domestica* are antagonistic was again tested in the lab for Furman et al. (1959). This time, larval house fly populations did not develop in culture medium containing soldier fly larvae. Neither this experiment nor the previous one (Fletcher et al., 1956) had treatment repetitions and discrepancies do exist.

In the field, it was shown that *H. illucens* larvae will replace *M. domestica* larvae in poultry manure if the manure is moistened (Furman et al., 1959). It was also demonstrated that larval populations of *H. illucens* will develop successfully when the larvae are introduced underneath the crust of dry manure.

The outlook for *H. illucens* as a biological control agent in Mexico is considered good (Vazquez-Gonzalez et al., 1962). These authors advocate keeping poultry manure wet, especially in the dry season, and destroying manure cones to augment *H. illucens* populations.

Chemical control. In the past, most of the chemicals used for fly control in privies were shown to cause resurgence of house fly populations and damage soldier fly populations (Kilpatrick and Schoof, 1959). Under normal circumstances, privies produced few house flies. This was attributed to water content of the excretia and the presence of *H. illucens*. When privies were sprayed with dieldrin, BHC, or chlordane, house fly production greatly increased. DDT, malathion, and diazinon had little or no effect on house fly production.

Axtell and Edwards (1970) field-tested various larvicides against larval populations of *H. illucens* in poultry manure. The best control was achieved with a 0.5% solution of Ravap™. After eliminating the soldier fly populations, retreatments were necessary to control resurgent house fly populations.

House fly pathogens

Bacillus thurengiensis has been fed to caged layers for fly control, but when fed at levels providing the best control, decreases in feed consumption, body weight, and egg production resulted (Burns et al., 1961). When sprayed on manure as a larvicide, *B. thurengiensis* was effective against fly larvae and did not damage populations of predaceous mites (Wicht, Jr. and Rodriguez, 1970). Records of other types of pathogens affecting house flies are abundant in the literature (Briggs and Milligan, 1977; Burges and Hussey, 1971; Kramer, 1964; Beard and Walton, 1965).

Insect growth regulators (IGR's)

The first juvenile hormone was extracted from the abdomen of a male cecropia moth over 20 years ago (Williams, 1956). Researchers have since been trying to develop compounds showing juvenile hormone activity for use as pesticides that would be specific for limited species of target insects but would not be detrimental to the environment (Novak, 1975). House flies were sensitive to the early IGR's (Herzog and Monroe, 1972) as were mosquitoes (Spielman and Williams, 1966). Several books are available giving the history, chemistry and mode of action of IGR's (Novak, 1975; Gilbert, 1976; Menn and Beroza, 1972), and the types of compounds exhibiting juvenile activity on insects (Slama, 1971).

Methoprene

Methoprene, or ZR-515, has been widely tested for the control of mosquitoes, house flies, and other diptera. Treatment residuals are rapidly degraded by sunlight and the half life is only 2 to 24 hours depending on the type of formulation (Schaefer and Dupras, 1973). Methoprene does not leach out of treated media into the environment (Wright and Jones, 1976) and is not active against nontarget insects in bovine fecal pats (Pickens and Miller, 1975).

As a feed additive, methoprene gave significant fly control when fed to cows at 2.5 mg/kg (Miller and Uebel, 1967). Breeden et al. (1968) fed methoprene to chickens in 86.9% technical and 7% encapsulated formulations. The technical formulation at 50 and 100 ppm gave good fly control 3 days and 1 day post-treatment respectively. The encapsulated formulation at 5 and 10 ppm gave good control 8 and 2 days post-treatment respectively. Adams et al. (1976) fed methoprene to hens for 42 days at

10 g/ton of feed. Good larval control was achieved, but inability to produce total control was blamed on migration of adult flies. Morgan et al. (1975) found that methoprene in chicken feed at 0.0005 and 0.01% produced mortalities of 70.9 and 99.3%, respectively, and had no effects on the hens' weight. Methoprene was not effective, however, when poultry manure was treated topically in the field.

Dimilin

Dimilin, also known as TH-6040 and diflubenzuron, has been classified as an inhibitor of chitin synthesis. Many analogues of dimilin have been synthesized and tested, but none are as effective as diflubenzuron itself (DeMilo et al., 1978). In the larval stages, dimilin causes rupture of larval cuticle during or shortly before the next molt (Jacob, 1973). Topical application to pupae can affect emergence of adults (Cerf and Georghiou, 1974). Application of dimilin to house fly adults can result in the suppressed hatchability of eggs laid long after the application date (Wright and Spates, 1976).

Even though dimilin was active against all major nontarget insects in bovine fecal pats (Pickens and Miller, 1975), poultry farms treated topically with dimilin had greater parasitoid populations and species variety than did farms treated with dimethoate (Ables et al., 1975). When dimilin was fed to chickens at 6.2 to 12.5 ppm, fly control of 100% was achieved, but residues were found in all eggs sampled (Miller et al., 1975).

Resistance to IGR's

House fly resistance has been demonstrated for both methoprene and dimilin (Plapp and Vinson, 1973; Oppenoorth and Van Der Pas, 1977; Georghiou et al., 1978).

Chemical larvicides

The use of chemical pesticides started about the same time the poultry industry began keeping chickens in cages (Hartman, 1953). The following is a brief review of chemicals that have been used as larvicides in poultry manure and their efficacy at the time they were tested. For a more complete review of larvicides, see Miller (1970).

The idea of oral larvicides evolved in the late 1920's. Cows were fed tannic acid, linseed oil, Mg_2SO_4, and NaCl as possible controls for horn flies (Miller, 1970).

Wolfenbarger and Hoffmann (1944) may have been the first to advocate the use of DDT as a house fly larvicide on poultry farms. An emulsion of 0.25% DDT applied to manure at 1.9 l/9.3 m^2 gave good house fly control, but soldier flies, *Hermetia illucens*, were fairly tolerant (Tanada et al., 1950).

A 1% solution of malathion EC applied at 3.8 l/9.3 m^2 controlled fly larvae after two applications 5 days apart. Adults resting on manure were also killed (Mayeux, 1954a). Malathion was more toxic to predatory mites than to house fly larvae (Axtell, 1966).

Diazinon applied as a liquid and as a dust controlled fly larvae for 1.5 to 2 weeks, but fly resurgence occurred after 2 weeks (Wilson and Gahan, 1957). Wicht, Jr. and Rodruguez (1970) achieved good control with diazinon and claimed little damage was done to predatory mite populations. Axtell (1966), however, reported that diazinon is just as toxic to mites as it is to flies.

Dichlorvos, 20% Shell Vapona™ resin strips ground up, gave good control of house fly larvae and adults for about 7 weeks with three

treatments (Bailey et al., 1971b). Dichlorvos is also toxic to predaceous mites (Axtell, 1966).

Rabon™, when applied to poultry manure as a larvicide, controlled flies for 1 (Bailey et al., 1968) to 2 weeks (Matthyssee and McClain, 1973). Rabon was also fed to dairy cows as an oral larvicide (Miller et al., 1970), but it proved to be ineffective in commercial operations (Miller and Pickens, 1975).

Thiocarbamide or thiourea, when applied weekly to manure at a rate of 0.26 g per bird in 152.0 l of water, achieved between 68 and 94% control of fly larvae (Jaynes and Vandepopuliere, 1978). Thiourea, as a larvicide, affects first-instar larvae more than second-instar larvae, and second-instar larvae more than third-instar larvae. Fly eggs and pupae are not affected (Hall et al., 1979).

Methods for Adult House Fly Control in Poultry Houses

Light traps

Ultraviolet light between 3300 and 3700 angstroms is effective for attracting flies (Tarry et al., 1971). Claims of good control of flies with light traps, however, are sometimes the results of tests performed with small fly populations (Tarry, 1968), or in ideal situations (Tarry et al., 1971). Schreck et al. (1975) limited light trap catches to *Stomoxys calcitrans* by using CO_2 as an additional attractant. Traps tested by Morgan et al. (1970) averaged 439.1 house flies per day over a 22-day period. Pickens et al. (1975) increased the house fly catch 2.4 times by placing a heated fly bait in the trap.

Trap height influences fly catches. Pickens et al. (1975) found that lowering traps from ceiling level to 0.5 m above the ground

increased the house fly catch 1.8- to 4.6-fold. Driggers (1971) caught 10.24 times as many house flies with traps at ground level than with traps 1.5 m above the ground. Prime trapping time for house flies at a north Florida poultry farm was from 5 min before sunset to 5 min after sunset (Driggers, 1971). Driggers (1971) reduced house flies at the farm by 52.8 and 73.1% in 1 and 4 weeks respectively, by using four light traps placed at ground level in a 121.9 m poultry house. Thimijan et al. (1972) estimated that 52 light traps would be needed in a screened dairy barn to capture 0.5% of the 2500 to 5000 flies that were being released in the barn daily during the test period.

Catches of flies by light traps have been found to be highly variable. As a result, light traps are recommended for survey work, but they are not considered consistent enough to accurately estimate fly populations (Pickens et al., 1972). A more complete summary of light trap evaluations has been prepared by Hienton (1974).

Baits

An early account of killing flies by attracting them with baits was published by Morrill (1914). He gives a full account of all items tested and their efficacy. The best combination was overripe banana on sticky fly paper.

Most baits used today are granulated sugar baits with or without attractants. Baits in other forms have been tested with some success. Mayeux (1954a) made a 1% solution of malathion in honey. Burlap was painted with this solution and hung in poultry houses to kill flies. Good control was attained and the bait was active for 1 to 24 days.

Wicht, Jr. and Rodriguez (1970) mixed LC_{95} concentrations of naled and ronnel with one-to-one mixtures of malt and water. These solutions were painted onto squares of waxed paper which were attached to bait stations made of plywood squares. Paper was replaced weekly. The naled bait attracted more flies and had a quicker knockdown than ronnel.

Granular baits are convenient to store and use, and have been tested more extensively than other types of baits. Mayeux (1954b) reduced house fly populations by 90% or more within 1 hour with a 1% malathion bait applied at 85.2 to 113.6 g/9.3 m^2. If kept dry, the bait killed at this level for 3 to 7 days. Sampson (1956) ranked the efficacy of granular test baits in the following order: endrin, heptachlor, lindane, and parathion (all at 0.125%) more effective than diazinon, dieldrin, DDT, and phenthiazine (all at 0.125%) more effective than aldrin and thiourea (both at 1.0%). Bailey et al. (1970) tested 1% sugar baits of dimethoate, fenthion, formothion, naled, ronnel, and trichlorfon. All gave better than 75% control for 18 days.

In 1971, resistance to trichlorfon (from 2.5 to 135.0 times) and dichlorvos baits (from 2.3 to 16.6 times) was reported from Florida (Bailey et al., 1971a).

Rogoff et al. (1964) demonstrated the presence of a house fly sex pheromone which Carlson et al. (1971) later identified as (Z)-9-tricosene, or Muscalure™. Muscalure and its homologs were then synthesized in the laboratory by Richter and Mangold (1973). The addition of Muscalure to sugar baits increased house fly catches (Carlson and Beroza, 1973). Only males were caught in laboratory studies, but equal numbers of males and females were caught in the field. Mulla et al.

(1977) tested compounds attractive to house flies and found that trimethylamine and indole were the main house fly attractants. Baits consisting of trimethylamine, indole, NH_4Cl, and linoleic acid were significantly superior to commercial preparations containing (Z)-9-tricosene.

Location of bait stations in and around poultry houses was found to influence the size and sex ratio of the catches. Baits located in the sunlight-shade border areas collected the greatest number of flies (Willson and Mulla, 1973). In bait stations near the center aisles, females outnumbered males, but a one-to-one ratio was approached in catches from the perimeters of poultry houses (Willson and Mulla, 1975). Bait stations dominated by one sex had catches significantly lower than those of stations conducive to both sexes.

Space sprays

This brief review is limited to use of synthetic pyrethroids as space sprays. In a study by Willis and Thomas (1975), pyrethroids gave better results than the ronnel standard, and resmethrin gave better results than allethrin. In another study, ronnel was more effective than resmethrin (Wilson et al., 1975). Other trials have shown that resmethrin is effective as a space spray against house flies (Mathis et al., 1972) and mosquitoes (Haskins et al., 1974). Permethrin was shown to have a knockdown 8 to 16 times faster than that of allethrin, and an LD_{50} three and four times higher than those of mesrethrin and synergized mesrethrin respectively (Lhoste and Rauch, 1976).

Kissam and Query (1976) tested an automatic piped-aerosol system that used a 0.7% synergized pyrethrin solution for fly control in

poultry houses. The system provided effective fly control and cost in the range of other fly control systems.

Contact residuals and resistance

The ability of insects to develop resistance was questioned by Melander (1914). The question was answered when DDT resistance was reported from several countries in Western Europe in 1947 (West, 1951). One year later, DDT resistance was reported in the U.S. (Hansens et al., 1948). DDT had only recently been advocated for use on poultry farms despite its slow knockdown and kill (Wolfenbarger and Hoffmann, 1944). A survey in Canada showed that house flies were still highly resistant to DDT (Batth and Stalker, 1970).

Sequential formation of resistance to contact residuals

In 1953, Hansens reported that lindane, methoxychlor, chlordane, and dieldrin applied as residual sprays failed to give control of house flies. The residual action of diazinon extended 10 weeks against susceptible flies and 4 weeks against resistant flies (Hansens and Bartley, 1953). Resistance to diazinon was noted soon afterward (Hansens, 1958) and in Florida it was reported to be 5- to 38-fold (LaBrecque et al., 1958). By 1970, diazinon resistance was 8- to 62-fold in New Jersey and a 1% solution failed to give satisfactory control (Hansens and Anderson, 1970). Flies showing resistance to diazinon also showed resistance to stirofos (Pickens et al., 1972), DDT, methoxychlor, chlordane, dieldrin. lindane, parathion, malathion, dicapthon, ronnel, trichlorfon, and conmaphos (Hansens, 1958).

Malathion resistance in Florida was about 4-fold in 1956 (LaBrecque and Wilson, 1961), 133-fold in 1958 (LaBrecque et al., 1958), and 275-fold in 1960 (LaBrecque and Wilson, 1961).

Hansens and Anderson (1970) found that a 1% solution of the following insecticides failed to give satisfactory fly control when applied as contact residuals: dimethoate, ronnel, stirofos, and bromophos.

Ficam™ gave good results as a contact residual against house flies. Sucrose was added to the solution to improve the knockdown. No resistance data are available (Lemon and Bromilow, 1977).

Synthetic pyrethroids

The first synthetic pyrethroid to be synthesized was allethrin (Schechter et al., 1949) followed by resmethrin (Elliot et al., 1965). Although natural pyrethrins are known for their quick knockdown (O'Brien, 1967), resmethrin proved to be 55 times more toxic to adult females of *M. domestica* than mixed esters of natural pyrethrins (Elliot et al., 1967). Haskins et al. (1974) claim resmethrin to be effective as a contact residual, but Mathis et al. (1972) claim the opposite. Synergised resmethrin had increased toxicity against resistant flies and the synergist prevented knockdown recovery (Schulze and Hansens, 1968).

Decamethrin is a highly toxic pyrethroid ester with an acute oral LD_{50} for female rats of 31 mg/kg. It can be rapidly absorbed by inhalation (Kavlock et al., 1979).

Permethrin is more effective at lower temperatures (Harris and Kinoshita, 1977). Half life of permethrin in soils with low and high organic content was 7 and 16 weeks respectively, with the loss of insecticide being attributed to microbial action (Williams and Brown, 1979). As in insects, the cis-permethrin isomer was more toxic to aquatic arthropods than the trans-isomer (Zitko et al., 1979).

The mode of action of pyrethroid poisoning is fairly complex. Initial signs in insects are usually incoordination and locomotor instability which are collectively termed knockdown. Details can be found in Wouters and van den Bercken (1978).

Resistance to pyrethroids can be detected in house flies after several months of strong selection pressure (Keiding, 1976). Permethrin resistance has been reported in culicids (Priester and Georghiou, 1978), and cross-resistance has been reported in DDT-resistant strains of culicids (Prasittisuk and Busvine, 1977) and cattle ticks, *Boophilus microplus* (Nolan et al., 1977).

Shono et al. (1978) reported that metabolic detoxification by ester hydrolysis and hydroxylation is a major factor limiting the insecticidal activity of the permethrin isomers.

Northern Fowl Mites

Description and Biology

Economic importance

The northern fowl mite is considered to be the most serious ectoparasite of poultry in the state of Florida (L. W. Kalch, *personal communication*), as well as the U.S. (Sulzberger and Kaminstein, 1936; Miller and Price, 1977; Smith, 1978). Since it was first recognized as a poultry pest by Wood in 1920, the northern fowl mite continued to spread across the country with increasing incidence (Linkfield and Ried, 1958).

Lyon (1975) stated that in 1970, the northern fowl mite could be costing the poultry industry $80 million annually. Smith (1978) quoted DeVaney as estimating an annual $66 million loss due to external

parasites causing decreases in egg production; parasite prevention might cost as much as $1.1 million. In Florida, Butler (1979) attributed a $3.7 million loss in poultry profits to the northern fowl mite in 1978.

Taxonomy

Although fowl mites were reported in the literature as early as 1824 (Toomey, 1921), the first accepted name, *Dermanyssus sylviarum* (Canestrini and Fanzago), was not seen until 1877 (Cameron, 1938). The inability of authors to properly identify the northern fowl mite resulted in the appearance of many synonyms. Several authors have followed this synonymy through the years until the accepted scientific name of the northern fowl mite was changed to *Ornithonyssus sylviarum* (C.& F.) in 1963 (Cameron, 1938; Furman, 1948; Furman and Radovsky, 1963; Laffoon, 1963).

The northern fowl mite was originally placed in the family Dermanyssidae, but was later separated to the Macronyssidae (James and Harwood, 1969). For years, *O. sylviarum* was confused with another poultry pest, the chicken mite, *Dermanyssus gallinae*. The two can be distinguished by the shapes of the anal plates and by the shapes of the dorsal shields (Lapage, 1956; Baker et al., 1956; Weisbroth, 1960). *Ornithonyssus sylviarum* has a teardrop-shaped anal plate and the dorsal shield tapers posteriorly; *D. gallinae* has a truncate anal plate and the dorsal shield is more rounded posteriorly. The complete morphology of the northern fowl mite is well documented (Allred, 1970; Georgi, 1974; Pound and Oliver, 1976; Krantz, 1978).

Bionomics

Wood (1920) and Cleveland (1923) published early works describing the biology and life cycle of the northern fowl mite. Cameron's research

(1938) was fairly complete at the time, but since he could not colonize the mite past the larval stage, he could not fully describe the life cycle. Colonization has since been accomplished (Chamberlain and Sikes, 1950; Cross, 1954; Cross and Wharton, 1964), and the entire life cycle has been described (Sikes and Chamberlain, 1954; Soulsby, 1968). According to Sikes and Chamberlain (1954), females lay an average of two to three eggs, each one within 48 hours after a blood meal. Eggs hatch in less than 1 day to six-legged non-feeding larvae which molt in less than 1 day to eight-legged protonymphs. Protonymphs take an average of 2 days in which to require the two blood meals necessary for full engorgement. Protonymphs molt to non-feeding deutonymphs that molt to adults in about a day and a half. Time from adult engorgement to second generation adult was about 5 to 7 days at 38 to 40 C with a relative humidity of 90 to 100%. Length of the cycle varies at least partly due to the intermittant feeding habits of the mites (Cameron, 1938).

The whole life cycle of *O. sylviarum* occurs on the host (Sikes and Chamberlain, 1954; Kirkwood, 1968; Loomis, 1978); however, oviposition may occur in the nest of the host (Cameron, 1938). Even though the northern fowl mite has long been considered a winter pest (Loomis, 1978), mites have been found on chickens all year round (Kirkwood, 1963 and 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), as compared to 34 weeks for *Dermanyssus gallinae* (Kirkwood, 1963).

The area on the host most preferred by the northern fowl mite is the vent region (Cameron, 1938), but in severe infestations, mites can be found over the entire body (Anonymous, 1959; Metcalf et al., 1962; Loomis et al., 1970). 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 (Cameron, 1938; Abasa, 1965) stated that roosters have more mites than hens, possibly due to differences in plummage. Males have more contour feathers near the vent, while females have more down near the vent. Feathers are preferred over down by *O. sylviarum* (Kirkwood, 1968).

Cameron (1938) described the erratic behavior of mite populations on poultry. Mites transfer from bird to bird and populations rapidly rise and decline, but some birds remain entirely free of mites. This phenomenon has been seen by other authors (Kirkwood, 1963; Loomis et al., 1970), who were also unable to explain its cause. Hall and Gross (1975) found that roosters with high levels of plasma corticosterone response to social stress 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 was also found that hens subjected to higher social stress had significantly lower mite populations than unstressed hens (Hall et al., 1978; Turner, 1978). Additional experiments indicated that although hens first coming into production are most susceptible to northern fowl mite infestation, estrogen alone is probably not responsible for the difference in mite susceptibility between hens and roosters (Hall et al., 1978).

Distribution

What were probably the first and the earliest samples of the northern fowl mite in the U.S. were described by Banks (1906) from specimens collected in 1895 in North Carolina. Since then, the northern fowl mite has been found in most of the warmer areas of the U.S. and Mexico (Benbrook, 1965; James and Harwood, 1969). Some claim northern fowl mites are found world-wide in the plummage of chickens (Baker et al., 1956). Citings from Great Britain (Taylor, 1930), Hawaii (Garrett and Haramoto, 1967), and New Zealand (Thomas and Watson, 1958) substantiate this claim.

Hosts and methods of dissemination

The northern fowl mite occurs on at least 22 species of birds and domestic poultry (Benbrook, 1965), and Avian hosts are considered to be the true hosts (Cameron, 1938). Many papers cite records of northern fowl mites found on species of native wild birds (Boyd et al., 1956; Hanson et al., 1957; Foulk and Matthysse, 1965; Phillis and Cromroy, 1972; Phillis et al., 1976) and exotic caged birds (Sulzberger and Kaminstein, 1936; Anonymous, 1951). Several host lists are available (Peters, 1933; Cameron, 1938; Strandtmann and Wharton, 1958).

Cameron (1938) lists rodents and man as accidental hosts. Other such hosts are rabbits (Sikes and Chamberlain, 1954), the house mouse, *Mus musculus* (Drummond, 1957), the big brown bat, *Eptesicus fuscus*, the cave bat, *Myotis velifer* (George and Strandtmann, 1960), and the norway rat, *Rattus norvegicus* (Hall and Turner, 1976; Miller and Price, 1977). The northern fowl mite could not be induced to feed on man in the lab (Sikes and Chamberlain, 1954).

Dissemination studies are few. Besides spreading from bird to bird (Cameron, 1938), Foulk (1964) found that four main methods of poultry flock infestation are by infested hatcheries and contract started-pullet farms, infested trucks and crates used to carry infested birds, infested personnel, equipment, or egg crates, and infested wild birds that enter poultry houses. While Hartman (1953) believed northern fowl mites to be carried by sparrows, Foulk (1964) was unable to infest chicks with northern fowl mites from sparrows. Mites have also been carried from farm to farm on filler flats that have not been fumigated after use (Anonymous, 1968).

Since the northern fowl mite has been found on the Norway rat and the house mouse in poultry houses (Hall and Turner, 1976; Miller and Price, 1977), it is assumed that these rodents may aid in mite dissemination.

Effects on the Host

Patent effects of northern fowl mite infestation

The most obvious sign of a northern fowl mite infestation is feathers in the vent area which have become matted and discolored (Yunker, 1973) from the eggs and excretion of the mites (Metcalf et al., 1962). Examination of birds reveals mites and usually evidence of skin irritation and feather plucking (Anonymous, 1967). In more severe cases, the skin becomes thickened and scabby (Anonymous, 1959; Metcalf et al., 1962; Yunker, 1973) due to secondary infection of the bites (Cameron, 1938).

While northern fowl mites seen crawling on freshly laid chicken eggs are an indication of a mite infestation, the number of mites

observed is not necessarily an indication of the severity of the infestation (J. F. Butler, *personal communication*). The way to determine the severity of infestation is to directly examine the suspected fowl and check for the symptomology described above. Restlessness at night due to irritation may be indicative of northern fowl mite infestation (Petrak, 1969), but again, positive determination of infestation can best be made by examination of birds.

<u>Latent effects of northern fowl mite infestation</u>

Death of the bird host is often associated with severe northern fowl mite infestations and could be termed the utmost patent effect. Death, however, is due to the results of certain latent effects. Cameron (1938) blamed loss of vitality and death on loss of blood. Although it is not known whether blood loss produced an anemia, death in severe cases has been attributed to anemia which resulted from exsanguination (Metcalf et al., 1962; Petrak, 1969; Koehler, 1977; Matthysse et al., 1974). Recent studies have shown that this is not necessarily the case. Loomis et al. (1970) worked with hens having mite populations from light to severe and anemia was not shown to be a symptom of heavy mite infestations. DeVaney et al. (1977) found no anemia in roosters due to mite populations.

Weight loss has also been attributed to severe northern fowl mite infestation (Anonymous, 1967; Koehler, 1977). DeVaney et al. (1977) found no significant differences in the weights of roosters due to mite populations. In another study weights of two groups of hens did not change significantly due to mite infestations (DeVaney, 1979).

One of the longstanding economic reasons for keeping flocks free of northern fowl mites has been that mites cause a drop in egg production (Cameron, 1938; Metcalf et al., 1962; Anonymous, 1967; Koehler, 1977; Rock, 1978; Smith, 1978). Combs et al. (1976) demonstrated that chemical removal of mites improved egg production. Other work done in the last 10 years also conflicts with studies attributing decreased egg production to northern fowl mites. Loomis et al. (1970) could find no significant difference in egg production due to mite populations. Bramhall (1972) discounts northern fowl mites as a reason for reduced egg production and suggests that poultrymen control mites only to prevent discomfort to workers. Eleazer (1978) found that uncontrolled northern fowl mite infestations did not cause reduced egg production and DeVaney (1979) reported 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.

Medical Importance of Northern Fowl Mites

Allergic reactions

Gamasoidosis, a poultry handlers' dermatitis caused by fowl mites, was reported in 1824 (Toomey, 1921). It has since been well established that northern fowl mites will attack man and produce transitory rashes on the skin (Riley and Johannsen, 1915; Van Der Hoeden, 1964; Frazier, 1969; James and Harwood, 1969; Georgi, 1974; Ebeling, 1975).

Riley and Johannsen (1915) called the mite-produced rash a pruritis and not a dermatitis since man does not present favorable conditions for mite viability. Both terms, pruritis and dermatitis, have been used by recent authors to describe the condition (Cahn and Shechter, 1958;

McGinnis, 1959; Genest, 1960). Papular, vesiculo-papular, urticarial, or a combination of these primary lesions will develop at the bite site, the extent and severity of which is thought to be due to an allergic mechanism (Frazier, 1969). Contact with living mites may not be necessary to produce symptoms as both body parts and excretory products of the mites have inherent toxic properties (Chandler, 1949).

Several non-poultry-related cases of northern fowl mite dermatitis have resulted from mites entering buildings via window air conditioners (Cahn and Shechter, 1958; McGinnis, 1959; Genest, 1960). In all of the cases, abandoned birds' nests were found in or near air conditioner air intakes. Affected persons were advised to remove mites by bathing after which all symptomology disappeared in 24 hours. Fumigation of the buildings and air conditioners, and removal of birds' nests from the air conditioners eliminated the mite populations.

Disease transmission

When it was found that the chicken mite, *Dermanyssus gallinae*, could transmit the virus of St. Louis encephalitis directly and trans-ovarily (Smith et al., 1945, 1946, 1947), the question arose as to whether or not the northern fowl mite possessed the same capability. Collections of northern fowl mites from wild birds yielded mixtures of viruses containing not only St. Louis encephalitis virus, but also the virus of western equine encephalitis (Reeves et al., 1947; Hammon et al., 1948; Bisseru, 1967). The importance of the mite as a vector or reser-voir for either virus later proved questionable (Reeves et al., 1955). Subsequent studies have shown the northern fowl mite to be a very poor transmittor of western equine encephalitis (Chamberlain and Sikes, 1955)

and eliminated it as a possible transmittor of St. Louis encephalitis (Chamberlain et al., 1957; Chamberlain, 1968).

The northern fowl mite has also been accused of transmitting fowl pox (Brody, 1936), Newcastle virus (Hofstad, 1949), *Lankesterella corvi*, a blood parasite of rooks (Baker et al., 1959), a *Bedsonia* species of Ornithosis virus (Meyer and Eddie, 1960), and a microtatobiote, order of Rickettsiales, of the family Bartonellaceae (Mettler, 1969). Proof of transmission could not be demonstrated for any of the organisms listed above.

Control of Northern Fowl Mites

Chemical control has been the method of choice for controlling northern fowl mites primarily because it is the only method available. No parasites or predators of the northern fowl mite are known at this time. Since the mites complete their entire life cycle on the host, biocontrol agents may not exist.

Many books are available that list various northern fowl mite controls (Hartman, 1953; Benbrook, 1965; Anonymous, 1967; Loomis, 1978). Benbrook (1965) gives the most comprehensive list of controls prior to 1940, some of which include dust baths containing road dust and wood ashes, ointments and powders containing mercury compounds, caraway oil and derris (rotenone), and fumigants such as SO_2 and HCN.

Some classes of compounds cannot be used around poultry due to their toxicity or their tendency to form residues in meat and eggs. Chlorinated hydrocarbons have been removed from use on or around poultry due to their formation of residues. Nicotine SO_4 should be used with caution since it can be toxic to birds and man. Many organophosphorus

compounds, such as parathion, diazinon, and fenthion (Baytex), have extremely high avian toxicities and are also excluded from use on or around poultry (Loomis, 1978).

Little or no research has been done on field application of miticides on poultry. Poultrymen report widespread mite resistance to labeled miticides, but many of the resistance problems are due to poor application methods (Eleazer, 1978).

Application methods have changed drastically with the advent of caged birds and increased flock size. Before 1940, treatment of each bird in a poultry flock with a dust, ointment, or dip was quite common. By 1950, the average size of a caged flock was 1500 to 2000 birds (Hartman, 1953), and the use of treatments that involved the handling of individual birds rapidly ceased.

A laboratory method was devised for *in vitro* evaluation of miticides (Foulk and Matthyssee, 1964). Disposable pipettes are dipped into miticides and northern fowl mites then drawn inside by use of a vacuum. The large end of the pipette is covered with fine mesh cloth and after mites are inside, the small end is plugged with clay. Next, the pipettes are placed in chambers with controlled temperature and humidity, and mortality is recorded in 24 hours. This method was also used by Hall et al. (1978) after slight modification.

Sulfur and nicotine sulfate

These two compounds have been recommended for treatment of northern fowl mites perhaps longer than others and were initially used because they had been used successfully for poultry louse control.

The use of sulfur in a dip was recommended by Payne (1929). The dip consisted of 57 g of sulfur and 28 g of soap for each liter of water. The dip was only for warm weather use. Emmel (1937) intermittantly fed chickens a diet that was 5% sulfur by weight and controlled not only mites, but also fleas and lice. Povar (1946) found that sulfur actually repelled mites *in vitro* and the mites continued living for 14 days.

Sulfur has been shown to be effective for nothern fowl mite control when added to poultry litter at the rate of 0.5 kg per 4.7 m^2 of litter (Foulk and Matthysse, 1963). Sulfur is added to poultry litter on the University of Florida Department of Poultry Science Research Farm and is routinely used to control northern fowl mites on floor birds (R. H. Harms, *personal communication*). A 1% sulfur spray proved ineffective for northern fowl mite control on caged birds (Furman, 1953).

Nicotine sulfate, or Black Leaf 40™, has been used as a roost paint (Payne, 1929; Hansens, 1951), a dust, a dip (Bishopp and Wagner, 1931), and a spray (Povar, 1946; Hartman, 1953). Dips consisted of 1 part 40% nicotine sulfate in 9 parts water with or without the addition of 28 g of soap per gal of solution (Bishopp and Wagner, 1931). Sprays contained 1 part nicotine sulfate and 13 parts water. Hartman (1953) recommended spraying at night and using three treatments at 3-day intervals.

Nicotine sulfate gave good northern fowl mite control for up to 1 month (Cutright, 1929) and was considered by Povar (1946) to be the best method of mite control as late as 1946. Furman et al. (1953) reported good, but temporary control with nicotine sulfate. Nicotine

sulfate kills by contact and fumigation. It may cause a 24-hour reduction in egg production and may kill birds if ventilation is inadequate (Bishopp and Wagner, 1931).

DDT and lindane

Before their ban due to residue formation, some chlorinated hydrocarbons were tested on poultry for northern fowl mite control. DDT was considered an ineffective control when a 10% dust would not control mites *in vivo* (Povar, 1946). Lindane (2% EC) gave good results when sprayed on the vent region of chickens (Hansens, 1951), and a 0.2% lindane powder is still recommended for northern fowl mite control on caged exotic birds (Dall et al., 1964).

Malathion

Malathion was at one time an effective compound for northern fowl mite control. Sprays of 0.25 and 0.5% gave good results at an application rate of 25 ml per bird (Hoffman, 1956, 1960). Litter treatments of 4% dust at a rate of 0.3 to 0.5 kg per 1.9 m^2 of litter gave good results on hens, but severe cases on roosters had to be dusted by hand (Harding, 1955). In a more recent test, 4 and 6% dusts, and 0.5 and 1.0% sprays of malathion were ineffective for mite control; a 25% dust gave control for only 3 weeks (Rodriguez and Riehl, 1963). Foulk and Matthysse (1963) found malathion to be ineffective and suggested that mites may be showing some resistance to the compound. Perhaps the first northern fowl mite resistance to malathion in the East was found in laboratory and field studies by Hall et al. (1978). Nelson and Bertun (1965) synergized malathion with triethyl trithiophosphate (ethyl DEF) and increased its toxicity 12.9 times.

In an effort to determine malathion toxicity to fowl, various fowl were dipped into solutions containing high concentrations of malathion. A 4% solution killed all birds dipped including one mature goose (Furman and Weinmann, 1956).

Carbaryl

In the laboratory, 25.0 and 12.5 ppm solutions of carbaryl killed 100% of the northern fowl mites tested (Harrison, 1961). In the field, a 0.1% solution provided control for only 1 week (Hoffman, 1960). Others got better results with sprays of 0.25 and 0.5% (Kraemer, 1959; Furman and Lee, 1969), and 2 to 4% (Foulk and Matthyssee, 1963). Loomis et al. (1970) got poor control on heavily infested hens with a 0.5% spray, but Hall et al. (1978) found carbaryl to be the most toxic of the compounds used in their study.

In tests involving carbaryl dust, Foulk and Matthysse (1963) achieved good results with a 3% dust but Rodriguez and Riehl (1963) got control for 22 weeks with a 1% dust.

In studies of the systemic effects of carbaryl on laying hens, single doses of carbaryl administered orally at 800 and 150 mg per kg of hen could be detected in the blood for 48 to 72, and 24 hours respectively. Five days after cessation of the smaller dosage, no residues were found in muscle, liver, fat, skin, or gizzard samples (Furman and Pieper, 1962). In another study, hens were fed 200 ppm of carbaryl for 7 days. At 3 to 7 days post-treatment, no residues could be found in muscle, liver, gizzard, skin, or eggs (McCay and Arthur, 1962).

Ronnel

Laboratory and field tests have shown that ronnel is more toxic to northern fowl mites than either barthrin (a botanical) or malathion

(Bigley et al., 1960). Sprays of 0.25 and 0.5% ronnel effectively controlled field populations of northern fowl mites (Kraemer, 1959; Khan, 1969). Good control was also provided with dusts of 1 and 5% ronnel (Knapp and Krause, 1960; Foulk and Matthysse, 1963).

Miscellaneous compounds

Other compounds, mostly organophosphorous compounds, giving good northern fowl mite control are coumaphos (Bay 21/199) sprays and dusts (Kraemer, 1959; Hoffman, 1960; Knapp and Krause, 1960; Foulk and Matthysse, 1963; Khan, 1969), stirofos (SD 8447) sprays and dusts (Furman and Lee, 1969; Nelson et al., 1969; Combs et al., 1976; Christensen et al., 1977), dichlorvos sprays and impregnated resin strands (Khan, 1969; Nelson et al., 1969), crotoxyphos (Ciodrin™ or SD 4294) mist sprays (Foulk and Matthysse, 1963), trichlorfon (Dylox™) sprays (Khan, 1969), naled sprays (Kraemer, 1959), chlordimeform sprays (Hall et al., 1975; Combs et al., 1976; Christensen et al., 1977), and neotran and sulphenone sprays and dusts (Furman, 1953; Furman et al., 1953).

Pyrethroids

Pyrethrum dust has been used with good results on poultry for northern fowl mite control (Cameron, 1938). Two synthetic pyrethroids, Ectiban™ and SD 43775, gave good results in laboratory studies. In the field, effective control was achieved for 57 days with concentrations of SD 43775 ranging from 0.0125 to 0.05% (Hall et al., 1978).

Mechanical controls

The two compounds briefly mentioned here are included only because they present alternate methods for mite control although the efficacy

or practical value of either one is questionable. Volck, which is commonly used on plant pests and is nontoxic to birds and mammals, was tested as an ectoparasite control agent for farm animals (Bruce, 1928; Caler, 1931). As a 5 to 10% dip, it gave 100% control of northern fowl mites in 24 hours. Silica aerogel was used to eliminate a northern fowl mite population that had infested a home via a bird's nest (Tarshis, 1964).

Systemics

Sulfaquinoxaline alone or with other sulfonamides acts as a systemic acaricide in birds infested with northern fowl mites (Beesley, 1973). When feed containing 0.033% sulfaquinoxaline was fed to layers, mite populations were reduced to near zero in approximately 4 weeks (Furman and Stratton, 1963). Feed with 0.05% sulfaquinoxaline was fed to layers for 24 hours once a week and an economic control level for northern fowl mites was reached in 4 weeks (Furman and Stratton, 1964). Out of 15 poultry flocks fed diets containing combinations of sulfaquinoxaline, sulfadimedine, sulfamerezine, and sulfathiazole for 1.5 to 6 weeks at concentrations of 0.0125 to 0.02% total sulfonimides, 14 flocks were free of northern fowl mites at the end of the test (Goldhaft, 1970).

METHODS AND MATERIALS

Laboratory Trials

Environmentally Controlled Rearing Conditions

Rearing of immature diptera was accomplished in a Percival™ forced-air, upright growth chamber. The temperature was maintained at 29.4 C, and continuous lighting was provided by two 40-watt fluorescent bulbs. The growth chamber was modified to include an external exhaust system with air supplied to the unit from within the laboratory. Cages of adult flies of various species were also kept in this growth chamber unless otherwise specified. Whenever a growth chamber is mentioned without further clarification in this paper, reference is being made to the Percival at 29.4 C, continuous lighting, and ambient humidity lowered by chamber temperature.

Some adult flies were kept in a walk-in growth chamber which had a relative humidity of 85% and a temperature of 26.7 C. Lighting, fluorescent and incandescent, was continuous. Since this chamber was used so infrequently, it will be referred to specifically throughout the text if applicable.

Colonization and Rearing of Flies

Musca domestica (L.) -- the house fly. The laboratory house fly colony was started with adults obtained from a poultry farm in Starke, Florida, in October of 1975. Wild flies were placed into 3.8-l plastic jars half-filled with moistened CSMA™ (Consumer Specialties Manufacturing Association) and allowed to oviposit. Jars were screened and

placed in the growth chamber. Pupae were removed from the jars, separated from the CSMA by flotation, and dried on paper towelling. Care was taken to be sure pupae were free of any mites that may have been attached to the field-collected adult flies.

Pupae were placed in a standard colony cage (51 X 25 X 25 cm) with four sides and one end covered with window screen. The remaining end, fitted with surgical stockinet, was used as an entryway. Water was provided in a pan ca. 5 cm deep. The water surface was covered with polyfoam chips to provide a resting area for the flies and reduce mortality from drowning. The adult diet, a commercially prepared naso-gastric mixture (Table 5), was thinly spread over a small (ca. 5 X 10 cm) piece of aluminum foil with the edges raised to resemble a shallow pan. Additional diet was added daily in thin layers. This method allowed for a larger feeding surface and reduced waste. Cages with adults were kept in the walk-in growth chamber.

Eggs were collected over 4-hr time periods in moist CSMA from cages where females were an average of 7 days old. CSMA was mixed with water at a ratio of 5:3 and placed loosely in plastic pans 36 cm in diameter and 14 cm deep. Eggs, about 1000 per pan, were placed 1 to 2 cm below the CSMA surface to simulate oviposition. Pans were screened and placed in the growth chamber.

When the colony was well established, a rearing schedule was set up to provide two cages of flies per week for testing purposes. The schedule was based on the average time from egg to pupae at 29.4 C being 10 days. Adults were discarded after 3 weeks.

Interesting contrasts to the above method of rearing house flies are presented by Grady (1928) and Monroe (1962).

Ophyra aenescens (Wied.) -- the black dump fly. The *Ophyra aenescens* laboratory colony originated from adults collected on a west Florida poultry farm in December of 1976. Eggs were set in pans 25 cm in diameter and 8 cm deep containing the fortified diet of CSMA, horn fly dry mix, and water as shown in Table 4 of the results section. Pupae were separated from the medium by flotation 7 days later, dried, and placed in a colony cage as for house flies. Besides the water and nasogastric mix put in the house fly cages (Table 5 of the results section), adult dump flies were supplied with cane sugar and dry fish meal.

Eggs were collected in moistened fish meal from 5- to 7-day-old females. Approximately 500 to 1000 eggs were set twice a week to maintain the colony.

Hermetia illucens (L.) -- the black soldier fly. This fly was reared in the laboratory on many occasions, but attempts to colonize it did not succeed. Females placed in jars readily laid eggs on moistened CSMA or the screened jar lids. Eggs were then set in moistened CSMA as for house flies. In the growth chamber, larval development required 25 days and pupation another 10 days. Eggs were set primarily to provide a source of early instar larvae for testing purposes. Besides CSMA, *H. illucens* was reared in chicken feed, fish meal, and mixtures of fish meal and CSMA, all moistened with water.

Phormia regina (Meigen) -- the black blow fly. This fly was attracted to the laboratory during the cooler months of the year and it was colonized for testing purposes. Females oviposited in moistened fish meal. Eggs were set in a mixture of 1 part fish meal, 1.5 parts CSMA, and 1.8 parts water. A medium of fish meal and water was

sufficient for larval development, but the addition of CSMA produced a lighter textured medium with increased moisture-holding capacity. In the growth chamber, larval development required 6 to 7 days and pupation another 5 to 6 days. Adults were maintained on cane sugar, fish meal, and nasogastric mix as for *Ophyra aenescens*.

Fannia canicularis (L.) -- the little house fly. *Fannia* was briefly colonized for a series of experiments. Females would readily oviposit on the surface of fish meal that was mixed with enough water to make a semiliquid paste. This mixture was preferred after it aged in the growth chamber for 24 hr. The surface of the fish meal, which becomes crusty, could be left in place as the larvae developed, or inverted with the adhering eggs onto a fresh cup of the fish meal paste. Eggs set weekly in 120-ml cups of medium provided an ample number of flies. At 29.4 C, the larval and pupal stages both required ca. 7 days. Adults were maintained on dry fish meal and cane sugar cubes.

Sarcophaga robusta (Aldrich) -- the flesh fly. While flies were being reared on fish meal in the growth chamber, sarcophaged flies, along with other dipteran and coleopteran species, began appearing inside the growth chamber. This activity ceased when the chamber's exhaust pipe was covered with a screen. These sarcophagid flies are also found in poultry manure, so attempts were made to colonize them.

Six females of different sizes were captured and screened inside 360-ml plastic cups with 180 ml of very moist fish meal and placed in the growth chamber. After females died, they were pinned and labeled for later comparison with their progeny. Third-instar larvae began migrating inside the upper halves of the cups 3 days after the females were screened in. After 2 days of migrating, pupation occurred, and

9 days later, adults began emerging. The size variance in the six groups of F_1 adults was greater than the size variance among the six original females. Microscopic comparisons of the flies, made with reference to Aldrich (1916) and James (1947), revealed that all specimens belonged to the same genus and species, *Sarcophaga robusta* (Aldrich), syn. *S. plinthopyga* (Wied.).

The colony was easily established. Females began mating and larvipositing when 5 and 11 days old respectively. Immatures were maintained as described above and adults were maintained on fish meal and cane sugar cubes.

Dissection and Mounting of Cephaloskeletons of Third-Instar Fly Larvae

Cephaloskeletons of two species of fly larvae were examined for morphological clues indicative of the modes of life of the larvae. Third-instar larvae were killed in boiling water and dried on paper towelling. Each was cut behind the cephaloskeleton so that only a narrow band of integument still joined the two parts. Next, larvae were placed in 10% KOH and boiled gently until the unsclerotized tissues surrounding the cephaloskeletons had dissolved. At the completion of the KOH treatment, larvae were dried for 1-hr periods, first in 70% and then in 90% ethanol. Larvae were stored in 100% ethanol. While submerged in 100% ethanol, as much larval integument and remaining soft tissues as possible were teased from the cephaloskeletons. The cephaloskeletons were stored overnight in phenol and the remaining portions of the larvae were discarded.

Cephaloskeletons were worked into a mixture of phenol-balsam and placed in desired positions on mounting slides. Care was taken that the specimens were completely covered with the phenol-balsam mixture.

The mixture was also used to position the glass chips necessary for coverslip support. At this point, slides were racked and racks placed in a dust-free enclosure to allow the phenol to evaporate. Three or four days were necessary for the evaporation step to be completed. This step can be hastened by placing slides in an oven at 50 C for 48 hr. If coverslips are added before phenol has completely evaporated, specimens may be damaged.

After the phenol had evaporated, coverslips were placed over the specimens using pure balsam. Slides were set aside until the balsam had dried.

Bioassay of Poultry Manure

Manure for laboratory bioassay was collected in 360-ml plastic cups. Samples were capped and then frozen for a minimum of 24 hr to kill fauna present in the manure. Prior to testing, samples were removed from the freezer and allowed to thaw completely. Twenty-four hours were usually required for samples to reach ambient temperature.

Unless otherwise specified, manure samples were seeded with first-instar larvae of the particular fly species to be tested. Eggs were used exclusively at first but their use was discontinued when first-instar larvae produced more precise results. Manure was never reconstituted with water.

After larvae were added, samples were covered with screen and placed in the growth chamber. Adults were allowed to emerge and die prior to examination of samples. Pupae and adults were separated from manure by flotation.

Addition of a Liquid Insect Growth Regulator (IGR) to Larval Media of Flies

In order to simulate conditions in the field, larval diets were moistened with water containing various levels of a liquid IGR. Control diets were prepared using plain water.

Diets were placed in 360-ml cups and first-instar house fly larvae were added instead of eggs. Cups were covered with screen and put in the growth chamber until pupae eclosed and adults died.

Laboratory Tests with Granular Baits

Knockdown tests. Test baits were sprinkled in brown paper bags, 21 by 13 by 6 cm, which had been stapled side by side to a piece of wood ca. 61 cm in length. This arrangement of baits was stored outside, under the eaves of the laboratory, to simulate actual weathering conditions.

On the morning that baits were placed in the bags, the knockdown test was conducted. Three- to five-day-old female colony house flies were transferred by means of a vacuum system to cylindrical window screen cages, 12 cm high by 7 cm in diameter. Cages were inverted over the baits with 10 flies per cage and four cages per bait. The cages had no bottom surface and allowed flies to come in direct contact with the baits. Mortality was noted at 10-min intervals throughout the day until all flies had died. Criterion for death was total lack of movement. After the test, baits were stored as described above.

Residual tests. At some time interval after the knockdown test and at selected intervals thereafter, the residual activity of the same bait samples used above was tested until daily fly mortality was less than 50%. Flies were exposed to the baits as described above and mortality was recorded after a 6-hr exposure period.

Attractiveness tests. Baits were sprinkled into bait stations fashioned of 3.8-l milk jugs (R. C. Axtell, *unpublished data*). Bait stations were placed in a 1.8 by 1.8 by 3.7 m screened enclosure into which 200 five-day-old female house flies had been released. The enclosure was in full sun but baits were protected from sun and rain by a small structure inside the enclosure. Mortality was recorded after 24 hr.

Topical Application of Insecticides to House Fly Adults

Stock solutions were made by placing 1 g of the active ingredients (AI) of the insecticide in 100-ml volumetric flasks and adding enough acetone to bring the volumes up to 100 ml. Test concentrations were made in acetone from serial dilutions of the stock solutions.

Before use, all glassware was washed in a detergent and rinsed thoroughly with water. When dry, three final rinses of acetone were applied and glassware was baked in an oven at 176.7 C for 24 hr.

Laboratory colony house flies, 3 to 5 days old, were immobilized with a vacuum and males discarded. While immobilized, female flies were dosed with test concentrations using a 10-μl Hamilton™ syringe equipped with a Hamilton™ repeating dispenser. Flies were released into cylindrical cages, 12 cm high by 7 cm in diameter. Cotton balls saturated with a sucrose solution were placed on the tops of cages as a food source.

Tests were performed at 26.1 C and ambient humidity. Mortality was recorded after 24 hr. Criterion for death was total cessation of movement.

Laboratory Bioassay of Acaricides

Northern fowl mites were exposed to various dosage levels of acaricides to collect data necessary for dosage-mortality curves. The testing procedures were adapted from those of Hall et al. (1978). Tests were standardized as suggested by Peet and Grady (1928).

Squares of muslin cloth were secured with neoprene bands over the wide ends of 23-cm disposable Pasteur pipets. Acaricides were dissolved in acetone and serially diluted with acetone to the desired concentrations in final volumes of 100 ml. Pipets, with cloth squares in place, were immersed in the acaricide solutions for ca. 20 sec, then removed and rolled on paper towelling to dry the outsides. Control pipets were treated with 100 ml of acetone. Next, pipets were tapped on paper towelling for 2 min, tapered ends down, to dry the insides. More complete drying was achieved by using a Gast™ electric pump to force air through the pipets for 20 min. Pipets were removed from the pump and used within 1 hr.

After pipets were ready for use, mites were collected from caged chickens at the University of Florida Poultry Science Farm and transported to the laboratory. Mites were emptied into a porcelain emesis basin which was placed inside a larger pan half-filled with water to prevent mites from escaping. The vacuum side of the above-mentioned pump was fitted with a length of neoprene tubing and the pressure set at 106 g/cm^2. The large ends of the pipets, with cloth squares attached, were slipped into the open end of the neoprene tubing. When the pump was turned on, mites were pulled into the pipets. After the desired number of mites were inside, the pipets were removed from the tubing. The tapered ends were snipped with a hemostat to a length that would

allow the pipets to stand diagonally inside 1000-ml beakers and the tips were sealed with clay.

Desiccators were fashioned from 1000-ml beakers. Salt solutions of 4 g of NaCl and 10 ml of water were added to the beakers to maintain the relative humidity at approximately 80%. Dry 10-ml beakers were placed inside the 1000-ml beakers to receive the pipet tips and keep them out of the salt solutions. Once the sealed pipets were inside, desiccators were covered with two layers of saran secured with a rubber band. Desiccators were placed in the growth chamber at 29.4 C. Mortality was recorded 24 hr later with complete cessation of movement the criterion for death.

One pipet containing 15 gravid female mites served as a replication. Each treatment was replicated four times. Control mortality averaged 11.26% and was never higher than 16.39%.

Field Trials

Rotovation

Rotovation is a term coined by poultrymen in the Tampa, Florida, area to describe a method of mechanically stirring manure in poultry houses to keep it dry and unattractive to flies. Manure is composted in place and can be used for fertilizer without further treatment.

The tilling unit, made by Selpats Manufacturing, Inc., P. O. Box 149, Palatka, Florida, 32077, is officially called the Dryovator™. The tiller is operated by the power take-off of a modified Kuboda L175 diesel tractor. Tractor and tiller are shown in Figure 1.

The act of rotovating is termed rototilling or more frequently, just tilling. Tilling was accomplished by driving the tractor down the walkway of a poultry house and pulling the tiller through the

Figure 1. View of tractor and tiller.

manure on either side of the walkway. Houses were tilled by pulling the tiller down one walkway and up the other. The process was reversed each time a house was tilled in order to more thoroughly stir the manure. This became the standard procedure in all tilling trials, even when houses were tilled less frequently than once a day. Figure 2 shows the tiller in operation. Since the dimensions of poultry houses vary from farm to farm, tillers must be custom-made for each farm.

<u>Description of the Tilling Site</u>

Tilling trials were accomplished on a north Florida poultry farm near Starke, Bradford County. Prior to construction of the farm, earth was removed so that the foundations of the poultry houses were 0.9 m lower than the level of the ground immediately surrounding the farm. This complicated drainage problems, especially during periods of wet weather. The farm consisted of four California-style flat-deck houses (Figure 5) 90 m in length containing 5000 chickens each, and one double-wide stair-step house containing 15000 chickens. All birds were housed three to a cage. Only the California-style houses were used in the pest management studies. The layout of the farm and the designation of the houses are shown in Figure 3. The watering system consisted of one Hart™ cup per every two cages. This system worked well when properly maintained and cups were routinely cleaned. Water and feed were free choice.

<u>Monitoring Larval Fly Populations</u>

Techniques developed for field evaluations of larval fly populations included the use of pupal traps. Cylinders 31 cm tall by 10 cm in diameter made of 1-cm mesh hardware cloth were filled with moistened wood chips and inserted into the chicken manure pack in poultry houses.

Figure 2. Tiller in operation.

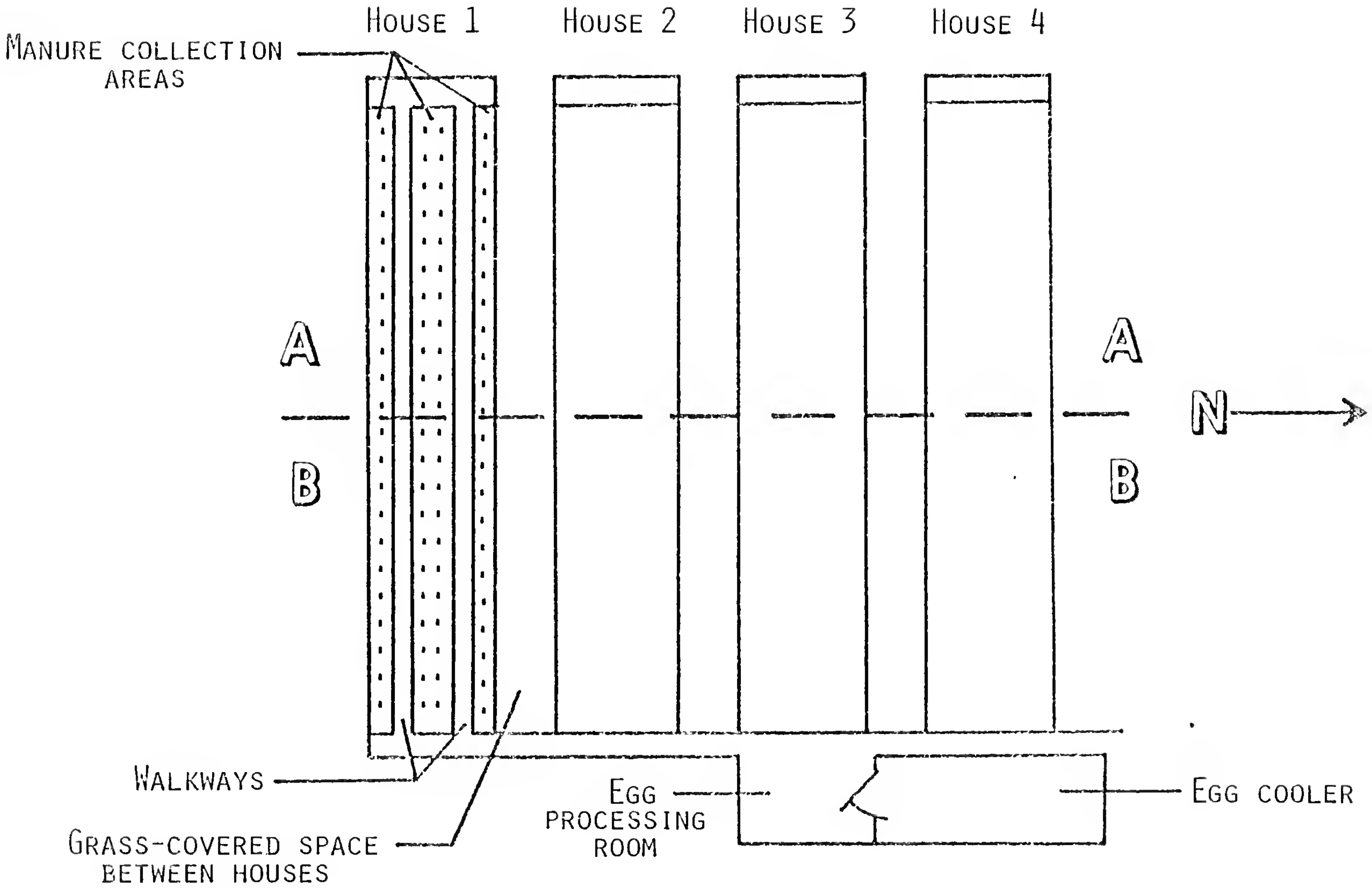


Figure 3. Layout and numeric designation of poultry houses at the tilling site.

A golf course turf plugger 10 cm in diameter was used to make the holes in which the cylinders were inserted. These cylinders, or pupal traps, were highly attractive to third-instar larvae as pupation sites and could therefore be used to collect fly pupae of known age.

In placement of pupal traps, areas in the manure pack were selected that appeared suitable for fly production. Once a site was chosen, the plugger was used to remove a horizontal plug of manure from the edge of the manure pack. The pupal trap was placed in the resulting hole and firmed into place. A tag with identifying data was tied to the bottom of the chicken cage directly above the trap to aid in locating the trap at a later date.

Collecting the traps was simple once they were located. Layers of manure made traps difficult to find at times even with the aid of the tags. Once found, traps were removed from the manure pack and tagged (Figure 4). Plastic bags are advisable for transport of traps after use.

Pupae were separated from the wood shavings by flotation. Trap contents were emptied into suitable containers and water was added. After ca. 30 min, wood shavings sink leaving only the pupae floating on the surface. If enough containers were available, all trap contents were floated simultaneously.

Poultry and Poultry Facilities Used When Evaluating IGR's as Oral Larvicides

When IGR's were tested as oral larvicides, the amount of manure needed for sampling and the frequency with which it was collected determined the number of hens used per treatment group. A hen voids ca. 92 ml of wet manure per day or 647 ml per week (Hart, 1963). Ten hens will produce 6468 ml of manure weekly, which is enough to provide a maximum

Figure 4. A tagged pupal trap after removal from manure pack.

of five 360-ml samples three times per week for laboratory bioassay. Therefore, the minimum number of hens used in a treatment group was 10.

To consolidate manure as much as possible, hens were housed two to a cage in cages measuring 20 by 45 by 43 cm. To prevent cross-contamination of manure, two cages were left empty between treatment groups and/or vertical tin dividers ca. 46 cm high were placed in the manure collection area between treatment groups. Manure collection areas were cleaned out before experiments began and covered with sheets of polyethylene or tin to catch the treated manure.

When IGR's were mixed with feed, vertical dividers were placed between feed troughs of treatment groups to prevent hens from sampling treatments other than their own. The continuous watering troughs used by all treatment groups were directly below the feed troughs and cross-contamination was possible via spilled feed. To minimize this problem, clay dividers were placed in the water troughs and water was independently piped into and drained out of the sections of trough that served each treatment group.

When IGR's were mixed with drinking water, water troughs were again divided between treatment groups. Water treatments were given to the chickens at 9:00 a.m., 12 noon, and 4:00 p.m. daily in the amounts of 50 ml per bird per treatment. Care was taken not to spill treated water into manure collection areas. Water troughs were lined with polyethylene to prevent them from being contaminated by unlabeled compounds.

Feed, treated or untreated, was always offered free choice and water was provided in either a continuous gravity flow system or on the schedule described above. Hens were used instead of roosters so that egg production could be monitored if desired. To maintain a maximum

rate of lay, hens were exposed to 14 hr of light by the use of supplemental incandescent lights in the morning and evening. All eggs produced by hens consuming unlabeled IGR's were destroyed and hens were destroyed after the experiments were terminated.

Calculation of Hen-Day Egg Production and Average Daily Feed Consumption

The term hen-day implies that during the time period over which the calculations for production or consumption have been made, daily hen mortality has been taken into consideration. In order to calculate on a hen-day basis, daily mortality records were kept.

Hen-days are calculated by multiplying the number of hens on hand by the number of days in the designated time period. This is simple if no mortality occurs. For example, 10 hens during a 7-day period constitute 70 hen-days. However, if one hen died on day 5, the number of hen-days becomes nine hens times 7 days plus one hen times 4 days for a total of 67 hen-days. Whether or not the day on which hen mortality occurs is to be counted in the calculations for hen-days should be decided in advance and adhered to.

Hen-day production is therefore the number of eggs laid during a time period divided by the number of hen-days calculated for the same time period. The quotient is multiplied by 100 since hen-day production is expressed as a per cent.

Consumption figures are usually expressed as the amount of feed consumed per bird per time period, e.g. 114 g per bird per day, and not as hen-day consumption. However, total hen-days as well as total feed consumption must be known in order to calculate the average daily consumption per bird. Total feed consumption is all the feed consumed

over a time period, which is found by weighing feed at the beginning and end of the designated period and subtracting the two figures.

Average daily consumption is therefore the total feed consumed during a time period divided by the hen-days for the time period.

Addition of IGR's to Poultry Feed

Insect growth regulators (IGR's) were added to the University of Florida Basal Layer Diet (Table 1). The basal diet was mixed in 136.4-kg lots in a Kelley-Duplex™ vertical mixer. When the basal diet was thoroughly mixed, ca. 4.5 kg were removed to a small paddle mixer where the appropriate amount of IGR was added while the mixer was in operation. This premixture was mixed for 10 min, returned to the vertical mixer, and added slowly to the mixing basal diet. After all of the premixture was added, mixing continued for 10 min. Diets were removed from the vertical mixer in six 22.7-kg batches and placed in aluminum cans for ease of handling. Feed cans were labeled and dated for identification purposes.

The vertical mixer was cleaned by swirling 11 kg of cracked corn inside the mixer for 10 min. The corn was removed and discarded. Next, fine feed particles were removed from the mixer with compressed air. The paddle mixer was cleaned with a whisk broom and compressed air. Mixers were always cleaned between mixes.

Topical Application of Granular IGR's to Poultry Manure

In the manure collection area of one poultry house at the tilling site, a granular IGR was applied to manure with a hand-held fertilizer spreader. Granules were preweighed at the laboratory and the amount for each treatment was individually placed in the spreader. The IGR was applied uniformly to manure treatment blocks until the spreader was empty.

Table 1. Composition of basal diet for poultry feed trials.

Ingredients	Per cent
Yellow corn	69.33
Soybean meal (50% protein)	19.00
Alfalfa meal (20% protein)	2.50
Ground limestone	6.17
Dicalcium phosphate	2.25
Iodized salt	.25
Micro-ingredient mix[a]	.50
Total	100.00
C.M.E./kg[b]	2890
Per cent protein[c]	16.2
Per cent calcium[c]	2.98
Per cent phosphorus[c]	.73

[a] Supplies per kg of diet; 6600 I.U. vitamin A; 2200 I.C.U. vitamin D_3; 11 I.U. vitamin E; 2.2 mg menadione dimethylpyrimidinal bisulfite (MPB); 4.4 mg riboflavin; 13.2 mg pantothenic acid; 59.6 mg niacin; 998.8 mg choline chloride; 22 mcg vitamin B_{12}; 110 mcg biotin; 125 mg ethoxyquin; 60 mg manganese; 50 mg iron, 6 mg copper; 0.198 mg cobalt; 1.1 mg iodine; 60 mg zinc.

[b] Calories metabolizable energy per kilogram.

[c] Calculated values.

On the University of Florida Poultry Science Farm, granules were uniformly applied to manure collection areas beneath treatment blocks of cages with a shaker fashioned from a 480-ml glass jar. Amounts were preweighed and applied to manure treatment blocks until the shaker was empty.

<u>Mixing and Application of Liquid IGR's and Organophosphorus Larvicides</u>

Liquid IGR's and commercial larvicides were applied to manure with a 7.7-l Sears™ pressure sprayer. The nozzle was adjusted so that the spray was emitted in a broad cone. Each treatment was applied to its block by computing the volume of larvicide to be applied, mixing the volume in the sprayer, and applying the volume uniformly to the particular treatment block until the sprayer was empty. The sprayer was cleaned thoroughly with water between applications of treatments.

Samples consisted of four 360-ml cups of manure collected from the center third of each treatment block. After a sample was collected, it was emptied into an aluminum pie pan. The pan was placed in the sun and the living larvae of selected fly species present in the sample were counted. Criterion for death was total cessation of movement.

<u>Addition of a Liquid IGR to the Drinking Water of Hens</u>

The facilities and water treatment application techniques used were described in the poultry facilities section. Test concentrations were prepared by serially diluting IGR stock solutions of 0.1 and 1.0%. Samples were bioassayed in the lab using first-instar house fly larvae.

<u>Placement of Light Traps</u>

Two blacklight electrocutor grid traps were evaluated at the tilling site. One trap was hung in the aisle between house 3 and the egg processing room, and the second was hung in the same aisle, but between

house 4 and the egg cooler (Figure 3). Traps were 1.8 m above the ground and 6.1 m apart. Both traps were similar in design but the one near the egg processing room, trap A, was yellow and the other one, trap B, was black (Figures 5 and 6). The manufacturer stated that the light sources for the two traps were producing light at different wave lengths, but exact values were not disclosed.

The traps were automatically turned on along with the farm's supplementary lighting system in the morning. They were in operation all day and were turned off at night along with the farm's supplemental lighting system. This reduced the collection of insects other than those associated with the poultry farm, e.g. nocturnal moths.

Traps were emptied weekly and the contents transported to the laboratory in plastic bags. Bags were labeled and frozen until contents could be analyzed. Catches were analyzed by counting selected species of flies in a representative sample of each catch. Samples consisted of a volume of each catch that weighed 10% of the total catch weight. The number of flies in a catch was assumed to be the number of flies counted in the sample multiplied by 10.

Field Tests with Granular Baits

Bait stations were fashioned from brown paper bags, 21 by 13 by 6 cm. The 6-cm lip helped keep baits and dead flies from being blown from the bait station by strong winds.

When testing was done at the tilling site, bait stations were placed at the sun-shade interface on the south sides of the poultry houses and secured by punching a 12-penny nail through the bag and into the ground. Baits were added to the bags after bags were secured.

Figure 5. Light trap opposite egg processing room. Note the flat-deck cage arrangement.

Figure 6. Light trap between egg cooler and house 4.

Following a 6-hr exposure period, each bait and station was placed into a plastic bag, returned to the laboratory, and the catch processed by sex.

When testing was done at the University of Florida Thoroughbred Unit at Ocala, Florida, bait stations were spaced along the edge of the concrete center aisle of a horse barn and each was secured with a rock.

After a 24-hr exposure period, baits and stations were collected and processed as above.

Application of Contact Residuals to Selected Surfaces

Templates of plywood, cement block, and galvinized tin were selected for use in residual tests because these are the types of surfaces most likely to be sprayed with a contact residual in poultry houses.

All templates were cleaned with soap and water and allowed to air dry prior to treatment. Pesticides were applied to run-off with a hand-held trigger-action sprayer. The first test began as soon as the templates had dried.

Another method for testing residuals by use of blotting paper templates is described by Batth (1974).

Application of Contact Residuals to Plywood Panels

Panels, 61 by 122 by 0.6 cm, were cut from 1.2 by 2.4 m plywood sheets and designed to hang with the long side in a horizontal position. Panels were hung by attaching two 46-cm lengths of light-weight chain to the upper corners. Aluminum rain gutters, for catching insects killed while on the panels, were placed horizontally along both sides of each panel so that the bottom of the guttering was even with the lower edge of the panel (Figure 7).

Figure 7. Panel with guttering suspended by chains at the tilling site.

Compounds were mixed using formulas as described in Neal (1974) and applied to run-off with hand-held trigger-action sprayers. Nozzles were adjusted to produce a cone 15 to 20 cm in diameter when the sprayers were held 31 cm from the panel surface. Sprayers were calibrated with graduated cylinders.

After insecticides were applied and allowed to dry, panels were hung in houses 1 through 4 at the tilling site, and the guttering was attached.

Evaluation of Northern Fowl Mite Populations

Field estimates. Field evaluation of mite populations on individual birds required two workers. The first worker, the handler, suspended the birds by their feet with the birds' breasts facing the second worker, the counter (Figures 8 and 9). The counter examined the birds, starting at the tip of the keel bone, working caudally to the vent area, and over the dorsal portion of the tail. In severe cases, mites were found on both legs down to the shanks, and more anteriorly than the tip of the keel bone.

Counts were designated as follows:

No mites seen	0
From 1 to 10 mites	Counted individually
From 10 to 100 mites	Counted by 5's, i.e. 15,20,25,etc.
From 100 to 200 mites	Counted by 50's
Over 200 mites	Counted by 100's

Counters and handlers never interchanged. Counters identified the birds and recorded the mite counts after birds were examined. Counters frequently double-checked each other to be sure that counts were uniform.

Calculation of a conversion factor. An attempt was made to correlate field-estimated mite populations with mite populations actually present on hens by extracting field-estimated mite populations from hens with a

Figure 8. A pair of workers examining a hen for mites. The counter is on the left. Note the stair-step cage arrangement.

Figure 9. A close-up view of Figure 8. The darkened area on the chicken is due to mites and mite debris.

soap and water solution. Ten birds with five different levels of field-estimated mite populations were washed and the mites counted in the laboratory. The field estimations, the laboratory estimations, the ratios between the two, and the mean of the ratios are shown below:

Field Estimation	Laboratory Estimation	Lab Est. / Field Est.
100	2710	27.10
500	2895	5.79
1000	5065	5.07
2500	4735	1.90
5000	4050	0.81
		$\bar{x}$ = 8.13

The mean of the ratios between the laboratory estimate and the field estimate was used as the correction factor. Field estimates were multiplied by 8.13 to arrive at a corrected field estimate. Unless otherwise stated, mite values referred to in the text are field-estimated values. Converted values are for reference only.

Field Application of Acaricides to Caged Hens

Acaricides were applied to caged hens with B.& G.™ cans and a Sears 7.7-l pressure sprayer. Nozzles on both types of sprayers were adjusted to emit cones ca. 15 cm wide when nozzles were held 31 to 46 cm below the cages. Acaricides were applied from beneath the cages in an effort to thoroughly soak the vent areas of the chickens. Whenever possible, applicators stood on the side of the cages opposite the feed trough to give then an unobstructed view of the hens while applying the acaricides.

Acaricides were mixed and applied according to directions found in the Insect Control Guide (FAES). Sprayers were cleaned thoroughly between treatments.

Field Application of Acaricides to Floor Birds

Birds were suspended by their feet and sprayed individually. The area between the keel bone and the tail was thoroughly saturated with the acaricide solution. Application was made with a B.& G. can. Litter was not treated.

Compounds Utilized for Fly or Mite Control

Common names, code letters and numbers and/or trademarks of the compounds utilized in this study are shown in Table 2. Names which are in accordance with the principles of Chemical Abstracts nomenclature are given if available (Kenaga and End, 1974). If the compound was supplied by a cooperator, the manufacturer's name is included. Compounds without a manufacturer's name were purchased locally.

Treatment of Data

Statistical Analysis System (SAS). Data were analyzed at the Northeast Regional Data Center (NERDL), University of Florida, Gainesville, Florida, using the Statistical Analysis System (SAS) of Barr et al. (1976, 1979).

Comparison of means. Methods for comparison of means, such as chi square and Tukey, were found in Snedecor (1961) and Freese (1963). Duncan's multiple range tests were performed by SAS.

Probit analysis. Probit analysis and the plotting of dosage-mortality curves were performed by SAS. An in-depth explanation of probit analysis and the calculation of a probit line was found in Finney (1964). Aid in interpretation of probit lines was given by Hoskins and Gordon (1956) and Tsukamoto (1963).

Correction of mortality. Where applicable, the results of pesticide trials were corrected by the methods of Abbott (1925).

Table 2. Compounds utilized for fly and/or mite control.

Common name	Code letters and numbers and/or trademarks	Chemical name	Cooperator
		Insect Growth Regulators (IGR's)	
methoprene	ZR-515, Altosid^{TM}	isopropyl (E,E)-11-methoxy-3,7,11-trimethyl-2,4-dodecadienoate	Zoecon^{TM}
dimilin	TH 6040	1-(4-chlorophenyl)-3-(2,6-difluorobenzoyl) urea	Thompson-Hayward
---	CGA 72662	---	Ciba-Geigy
		Larvicides	
dimethoate	Cygon^{TM}	0,0-dimethyl S-(N-methylcarbamoylmethyl) phosphorodithioate	---
dichlorvos	DDVP, Vapona^{TM}	2,2-dichlorovinyl dimethyl phosphate	---
---	Ravap^{TM}	Rabon^{TM} [2-chloro-1-(2,4,5 trichlorophenyl) vinyl dimethyl phosphate] + Vapona^{TM} (See dichlorvos above)	---

Table 2. Continued.

Common name	Code letters and numbers and/or trademark	Chemical name	Cooperators
		Baits	
---	Bomyl™	dimethyl 3-hydroxyglutaconate dimethyl phosphate	Farnam
dichlorvos	See Larvicides above		
ronnel	Korlan™	0,0-dimethyl 0-2,4,5-trichlorophenyl phosphorothioate	Farnam
methomyl	Lannate™	methyl N-[(methylcarbamoyl)oxy] thioacetimidate	Farnam
---	Golden Malrin™	dichlorvos (See Larvicides above) + ronnel (Above)	---
		Contact Residuals	
fenvalerate	SD 43775	---	Shell
permethrin	BW 21Z	(3-phenoxyphenyl)methyl($\pm$)-cis, trans-3-(2,2-dichloroethenyl)-2,2-dimethylcyclopropanecarboxylate	Burroughs Wellcome
permethrin	ICI 143, JFU 5819, JFU 5021A	---	ICI Americas

Table 2. Continued.

Common name	Code letters and numbers and/or trademark	Chemical name	Cooperators
		<u>Acaricides</u>	
fenvalerate	SD 43775	---	Shell
resmethrin	SBP-1382	---	Penick
permethrin	BW 21Z	(See Contact Residuals above)	Burroughs Wellcome
---	Ravap	(See Larvicides above)	---
permethrin	Ectiban^{TM}	(3-phenoxyphenyl)methyl(±)-cis, trans-3-(2,2-dichloroethenyl)-2,2-dimethylcyclopropanecarboxylate	ICI United States
carbaryl	Sevin^{TM}	1-naphthyl methylcarbamate	---
malathion	Cythion^{TM}	diethyl mercaptosuccinate, S-ester with 0,0-dimethyl phosphorodithioate	---

RESULTS

House Flies

Manure Management

Tilling wet manure. At the tilling site, fairly dry manure, 10 to 15 cm deep, had become wet from seasonal blowing rains and threatened to overflow onto the walkways. An attempt was made to dry the manure by tilling every house twice a day, 7 days a week. Houses 1 through 4 were tilled and then the process was repeated after a 30-min interval. Tilling was done during the noon hour when the temperature was high and workers were not in the houses.

Results. When tilling began, the manure pack was not uniformly wet, but too wet in most places for house flies to breed. The presence of house fly adults was hardly noticed and soldier flies, if present, were not evident. Manure was a shapeless mass with the consistency of a thick paste. Problems were compounded in some areas by leaking Hart cups.

Tilling tended to dry and texturize as well as push manure away from the walks and leave it in mounds towards the center of the manure collection areas (Figure 10). After 1 week of tilling, the manure pack began to hold its shape, but flowed back to the edges of the walkways after 24 hr. Although the moisture level had dropped by a noticeable amount, the manure had the consistency of mashed potatoes and was not yet breaking into individual pieces when tilled.

Figure 10. The appearance of fairly dry manure after tilling. Note how manure is pushed away from the walk and mounded in the center of the manure collection area.

As drying increased, pockets of house fly larvae began to show up in areas now suitable for their development. Tilling stirred the flies and caused them to reorient at the manure surface, but it is doubtful that tilling at this rate prevented them from completing their cycles. Pockets of maggots tilled one day had reformed by the next. Soldier flies were still not present in large numbers and the manure was now becoming drier than they preferred.

By the end of the second week, manure began to break up into chunks ranging from 3 to 10 cm in size (Figure 11). Although this was a sign that the moisture level of the manure was decreasing, numerous pockets of house fly larvae were proof that the manure was still not dry enough to retard their development. The manure pack now held its shape overnight and no longer threatened to overflow onto the walkways.

By the end of the third week, manure was becoming more friable. In most areas, manure had broken into 3-to 5-cm chunks which were crusty on the outside and wet on the inside. Drying continued and pockets of house fly larvae became fewer in number. The manure pack was gradually losing volume due to the drying process. This was evident from the increased space in the manure collection area, i.e. the space, after tilling, between the walk and the manure pack, and by the decrease in the amount of manure thrown onto the walks while the tiller was in operation.

At this time, the farm owner decided that the manure was dry enough to be removed from the houses. Despite my suggestions that he wait until a later date, the manure was removed and the tilling program terminated.

Figure 11. Manure which has dried enough to form particles of various sizes when tilled.

No rain fell during this tilling experiment. Temperatures were between 29.4 and 32.2 C during the day and a stiff breeze was blowing at ground level.

Results recorded during this and other tilling experiments were mostly subjective due to the difficulty in utilizing objective sampling methods. Pupal traps could not be employed because of the tilling schedules and facilities for drying manure were not available when all tilling experiments were performed.

Tilling after the addition of wood chips and sand to manure. When manure had completely liquified due to blowing rains, and tilling was ineffective, builder's sand and wood chips were added to manure in houses 1, 2, and 4 to improve the consistency. The experimental design is shown in Figure 12. The manure collection areas between the walks were treated and evaluated. The treatment blocks in houses 2 and 4 were 7.44 m^2. House 1 was divided in half, and chips and sand were put in the back(A) and front(B) halves respectively (see Figure 12). Chips were added until they were an average of 5 cm higher than the walk after spreading. Equivalent amounts of sand were added to the assigned blocks (Figure 12). House 3 was tilled, but no chips or sand was added. After spreading chips and sand with rakes, all four houses were tilled. Figures 13 and 14 show the appearance of the chips before spreading and after the initial tilling. Tilling continued on a daily basis for only 11 days, at which time the poultryman decided to clean out the houses.

Results. Sand was found to be ineffective for improving the consistency of liquified manure. It was heavy and difficult to distribute in the houses. When moistened by the manure, the sand became even

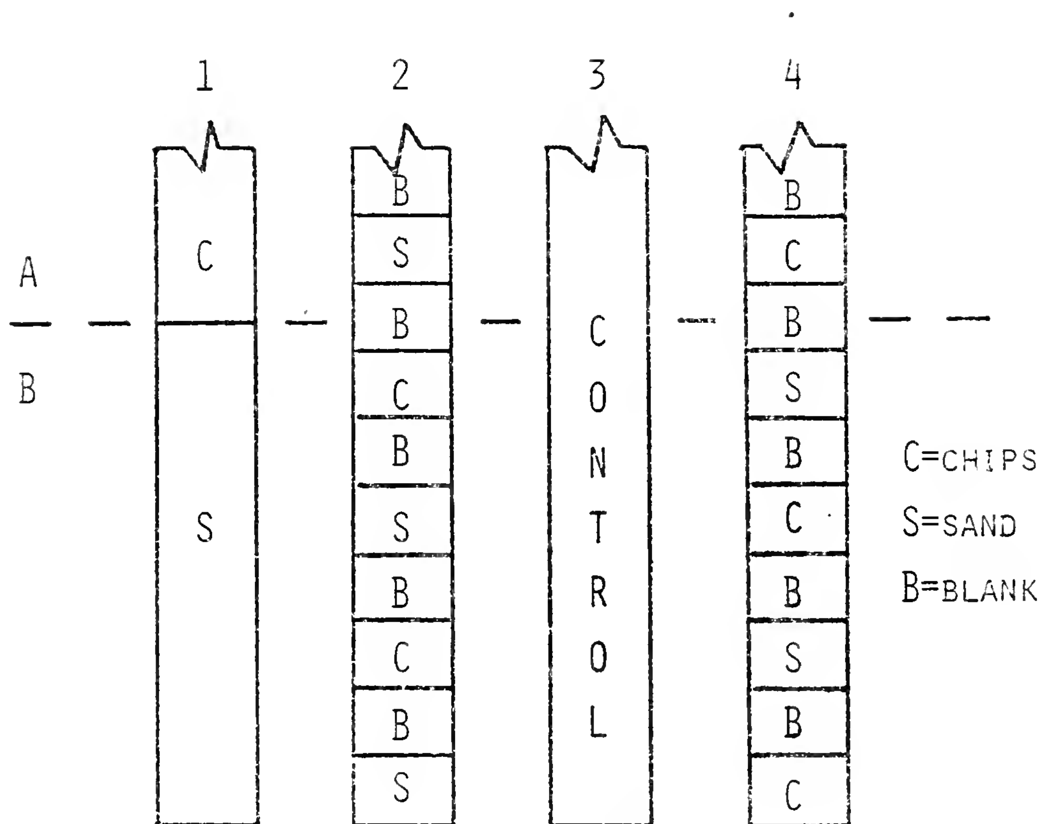


Figure 12. Experimental design for adding builder's sand and wocd chips to houses 1 through 4 at the tilling site. Only the front two-thirds of the houses are shown.

Figure 13. The appearance of chips before spreading.

Figure 14. The appearance of chips after the initial tilling.

heavier and made the manure difficult to remove when poultry houses were cleaned out. The extra weight of the sand could not be tolerated by the manure spreader owned by the poultryman, and he voiced his dissatisfaction after having to make several minor repairs as a result.

Instead of aerating manure, sand packed it down tight. Sufficient amounts could not be added to wet manure to provide the consistency needed for tilling without causing the manure to be too heavy.

Wood chips proved to be an excellent additive to liquified poultry manure. Chips were light and easy to handle. They aerated and aided in drying manure, and facilitated manure removal. Wood chips were also 50% cheaper in price than sand and more readily available.

After chips had been added and manure was tilled once a day for 2 days, the manure had a consistency that was still wet, but friable. Fresh manure had a relatively dry bed to fall upon before being tilled. Chips did not pack like sand, but remained light and enhanced drying by providing increased surface area.

By the 11th day, the areas where chips had been added were still wet but in much better condition for removal from the poultry houses than was the manure in other treatment groups. The control house was unchanged and the manure had the consistency of thick soup. The houses where sand had been added were essentially the same as the control, but some areas now had a thicker, heavier consistency.

No rain fell while the experiment was performed, but skies remained overcast. Temperatures averaged 27 C and the air was calm.

Tilling with and without the addition of wood chips to manure. On one occasion when the poultryman had his houses cleaned out, the manure and the sand beneath it, both of which were dry, were removed to a level

ca. 4 cm below the lower surface of the concrete walks. Subsequent heavy rains flooded the farm and the manure collection areas (Figure 15). It was decided to add wood chips to all manure collection areas to help dry and texturize the manure. The treatment schedule is shown below:

House	Chips
1	not added
2	added to rear of house only
3	added to entire house
4	not added

The level of the chips before tilling is shown in Figure 16. After chips were added, all houses were tilled twice to stir in the chips and make moisture levels more uniform when the pretreatment manure samples were taken. House 1 was tilled on Mondays, house 3 on Mondays and Fridays, and house 4 on Mondays, Wednesdays, and Fridays. House 2 was not tilled.

On Fridays, one 360-ml manure sample was collected from the center of each quarter of each house. At the laboratory, 30 g of each sample were weighed on an electric pan balance and dried for 36 hr on Berlese funnels. Samples were reweighed and the dry weights subtracted from 30 g. These differences were converted to per cents to arrive at the moisture contents of the samples.

Results. The results from the manure samples dried for moisture content are shown in Table 3 and Figure 17.

Moisture levels were reduced in all treatments except 2-B, where no chips were added to the manure and the manure was not tilled. The moisture level in this treatment increased significantly from 49.35 to 64.32% and constituted a net moisture increase of 14.97%. All other changes in moisture levels were nonsignificant.

Figure 15. The appearance of manure collection areas at the tilling site after manure removal and subsequent flooding. Note that the walk has been exposed from improper manure removal procedures.

Figure 16. Addition of wood chips to flooded manure collection areas.

Table 3. Moisture levels(%) of manure samples from the tilling site and net change in moisture content(%).

Treatment Period	Poultry Houses 1 A	1 B	2 A	2 B	3 A	3 B	4 A	4 B
Pre-Treatment	64.70	67.93	63.57	49.35[a]	53.30	63.10	72.83	65.13
1	68.40	57.47	67.80	64.37	66.67	61.03	70.07	66.30
2	69.47	62.50	63.93	66.03	43.33	51.27	66.47	69.77
3	63.20	66.20	62.77	61.40	57.37	52.07	65.75	70.00
4	54.32	63.90	57.85	48.25	53.08	61.65	69.03	66.98
5	55.83	60.67	61.33	50.92	66.57	64.42	71.57	63.25
6	59.13	61.37	58.85	53.62	54.23	59.68	63.93	67.85
7	58.50	55.55	64.47	49.02	54.93	53.80	69.63	66.78
8	47.38	51.85	44.82	55.42	41.33	49.63	55.27	46.32
9	55.20	62.58	56.45	64.32[a]	44.70	57.37	61.82	60.72
Net Change	-9.50	-5.35	-7.12	+14.97	-8.60	-5.73	-11.01	-4.41

[a] Indicates significance at $p < .05\%$ between the pretreatment sample and the sample from period 9.

Note: The duration of each treatment period was 7 days. Treatments are as follows:
1A - no chips, tilled 1X/wk
1B - no chips, tilled 1X/wk
2A - chips added, no tilling
2B - no chips, no tilling
3A - chips added, tilled 2X/wk
3B - chips added, tilled 2X/2k
4A - no chips, tilled 3X/wk
4B - no chips, tilled 3X/wk

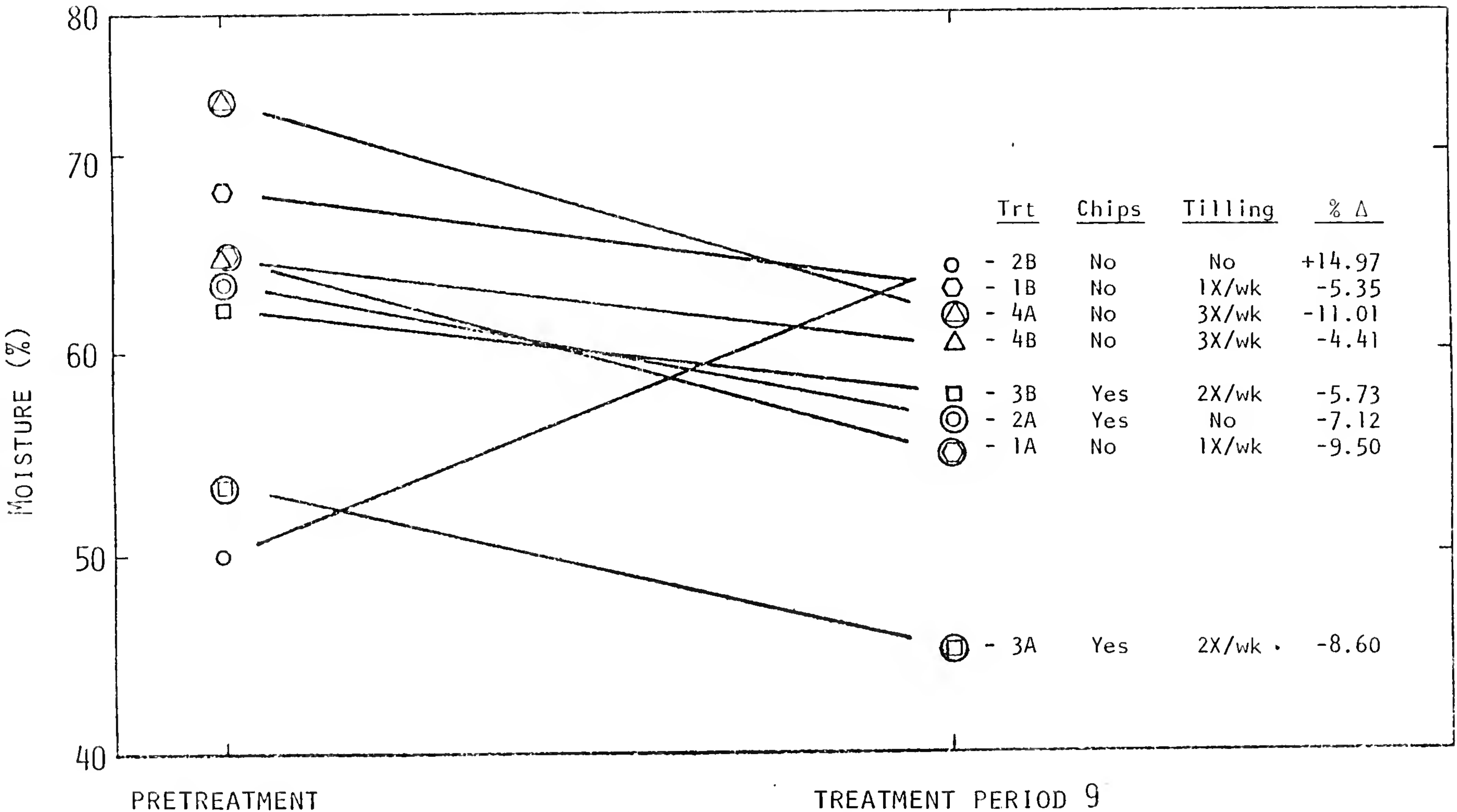


Figure 17. Net results of the manure drying experiment with wood chips and tilling 9 weeks post-treatment.

The greatest reduction in moisture level, 11.01%, was in treatment block 4-A, where no chips were added and manure was tilled 3 times per week. This treatment area also had the highest moisture level when the experiment began and third highest when the experiment ended. The final moisture level of treatment block 4-A was approaching 60% which is the minimum level that flies prefer to use for breeding purposes (Miller et al., 1974).

Moisture levels of treatments 1-B (without chips), 4-B (without chips), 1-A (without chips), and 3-B (with chips) were all reduced to near 60% or below 60% by the use of different tilling schedules (Figure 17). The moisture level of treatment 2-A (with chips) dropped to below the 60% level during the treatment period with no tilling at all. The moisture level of 53.30% in treatment 3-A (with chips) was well below the 60% level when the experiment began and dropped to 44.70% when the experiment ended. Figure 18 shows manure in 3-B at the end of the 9-week experiment.

When the experiment was terminated, the manure in all treatment areas except 4-A, 4-B, and 1-B maintained good form after tilling. The manure in the three above-mentioned areas appeared quite wet even though the moisture levels were just above 60%.

In treatment areas that were not tilled, a crust had formed on the manure in most places. In wet areas, this kept moisture in and prevented further drying. Pockets of house fly larvae were not noted in these areas during the course of the experiment.

Arriving at more accurate manure moisture levels was limited by the number of samples that could be processed by our laboratory. Only two samples could be taken from each treatment area every week. Eight

Figure 18. The manure in 3-B at the end of the experiment.

samples would have been much more satisfactory since each treatment area was 45.7 m in length. Overall limiting factors were drying facilities, driving distance to the tilling site, and labor.

Ophyra aenescens Basic Biology Studies

Larval diet and adult longevity studies. Four preliminary studies were used to develop larval and adult diets for *O. aenescens.* The ingredients in the two larval diets are shown in Table 4, and the diet combinations for larvae and adults are summarized in Table 5. Since these were only preliminary studies and the resulting fly numbers were relatively large (> 30), there were no treatment replications.

Pans containing the experimental larval diets were seeded with 500 and 1000 eggs. Eggs were covered loosely with approximately 2 to 3 cm of medium to simulate oviposition. Pans were covered with fine nylon screen and placed in the growth chamber (29.4 C).

After 10 days, pupae were separated from the media by flotation, dried, and placed in standard colony cages. Each cage contained one of the adult diets shown in Table 5, and a water source with polyfoam chips on the surface to reduce drowning mortality. Cages of adults were kept in the walk-in growth chamber (26.7 C).

Adults were allowed to emerge for 24 hr, after which the remaining pupae were removed from the cages. Beginning 24 hr later, dead flies were removed from the cages on a daily basis and daily mortality records were kept by sex until all the flies in each cage were dead.

Results. The results of the four *O. aenescens* adult longevity studies plus the average adult life span and length of the life cycle in each study are shown in Table 6. Raw data are shown in Appendix 1. Fortification of the larval diet did not shorten the 12- to 14-day

Table 4. Fortified and unfortified diets used during the preliminary colonization studies with *Ophyra aenescens* larvae.

	Diet	
Constituent	Unfortified	Fortified
CSMA™[a]	500 ml (151g)	400 ml (113g)
Horn fly dry mix[b]	---	100 ml (72g)
Water	375 ml	275 ml
Yield	~700 ml	~800 ml

[a] Consumer's Specialties Manufacturing Association (Ralston-Purina).

[b] Horn fly dry mix (Greer, 1975)--wheat flour-960g, fish meal-720g, Na_2CO_3-120g, alfalfa meal-400g.

Table 5. Combinations of larval and adult diets used during the four preliminary colonization studies with *Ophyra aenescens*.

Diet	1	2	3	4
Larval	Unfortified[a]	Fortified[b]	Unfortified	Fortified
Adult	Nasogastric mix[c] and fish meal	Nasogastric mix, fish meal, and cane sugar	Nasogastric mix, fish meal, and cane sugar	Fish meal and cane sugar

[a] CSMA™ (Ralston-Purina) plus water.

[b] CSMA™, horn fly dry mix (Greer, 1975)--960g wheat flour, 720g fish meal, 120g Na_2CO_3. 400g alfalfa meal; plus water.

[c] Challenge Cream and Butter Ass., Los Angeles, Ca. Each 270g furnishes: 7.5g moisture, 2.7g milk fat, 73.0g protein, 16.6g ash, 168.8g carbohydrate, 1.0g sodium, 3.2g potassium, 1mg thiamine hydrochloride, 2mg riboflavin, 10mg nicotinamide, 0.5mg pyridoxine hydrochloride, 10mg calcium pantothenate, 0.25mg folic acid, 150mg ascorbic acid.

Table 6. Summary of the four *Ophyra aenescens* adult longevity studies including average adult life span and the length of the life cycle from each study.

Diet	1		2		3		4	
Larval	Unfortified[a]		Fortified[b]		Unfortified		Fortified	
Adult	Nasogastric mix[c] and fish meal		Nasogastric mix, fish meal, and cane sugar		Nasogastric mix, fish meal, and cane sugar		fish meal and cane sugar	
	Males	Females	Males	Females	Males	Females	Males	Females
Average adult life span(days)	2	2	18	21	18	22	14	17
Time from egg to adult(days)	13		14		12		12	

[a] CSMA™ (Ralston-Purina) plus water

[b] CSMA™, horn fly dry mix (Greer, 1975)--960g wheat flour, 720g fish meal, 120g Na_2CO_3, 400g alfalfa meal; plus water.

[c] Challenge Cream and Butter Ass., Los Angeles, Ca. Each 270g furnishes: 7.5g moisture, 2.7g milk fat, 73.0g protein, 16.6g ash, 168.8g carbohydrate, 1.0g sodium, 3.2g potassium, 0.5mg pyridoxine hydrochloride, 10mg calcium pantothenate, 0.25mg folic acid, 150mg ascorbic acid, 1mg thiamine hydrochloride, 2mg riboflavin, 10mg nicotinamide.

period required for larval development, but sugar increased the average adult life span 9 times. The average life span for adults when sugar was not included in the diet was 2 days compared to 18 days for adults having access to sugar. By the time adults were 2 weeks old, mortality averaged 20% in groups consuming sugar and 90% in the group not consuming sugar (Figure 19).

Females lived for an average of 20 days and outlived males by an average of 3 to 4 days. Individual males and females were kept alive in the laboratory for 36 and 39 days respectively. Sex ratios were 50:50 when the totals of both sexes from all four tests were combined and subjected to a chi square analysis.

Larval viability study. To evaluate the effects of fortified and unfortified larval diets on larval development, 700 ml of each diet were loosely placed in four 1000-ml pyrex beakers. Sixty first-instar larvae of *O. aenescens* were added to each beaker. Beakers were covered with fine nylon screen and placed in the growth chamber. When adults began to emerge, remaining pupae were separated from the media by flotation, dried, weighed, and set aside for emergence of remaining adults. After adults emerged, empty pupal cases were reweighed.

Results. The number of pupae, the per cent pupation, the numerical and per cent larval emergence, and the larval viability of *O. aenescens* reared in fortified and unfortified larval diets are shown in Table 7. Fortification of the larval diet significantly increased the number of pupae that formed and the number of adults that emerged. The per cent viabilities of the larvae grown in the fortified and unfortified diets were 52.5 and 3.8% respectively. Development time from larvae to adults was 14 days for both diets. There was no significant difference in pupal

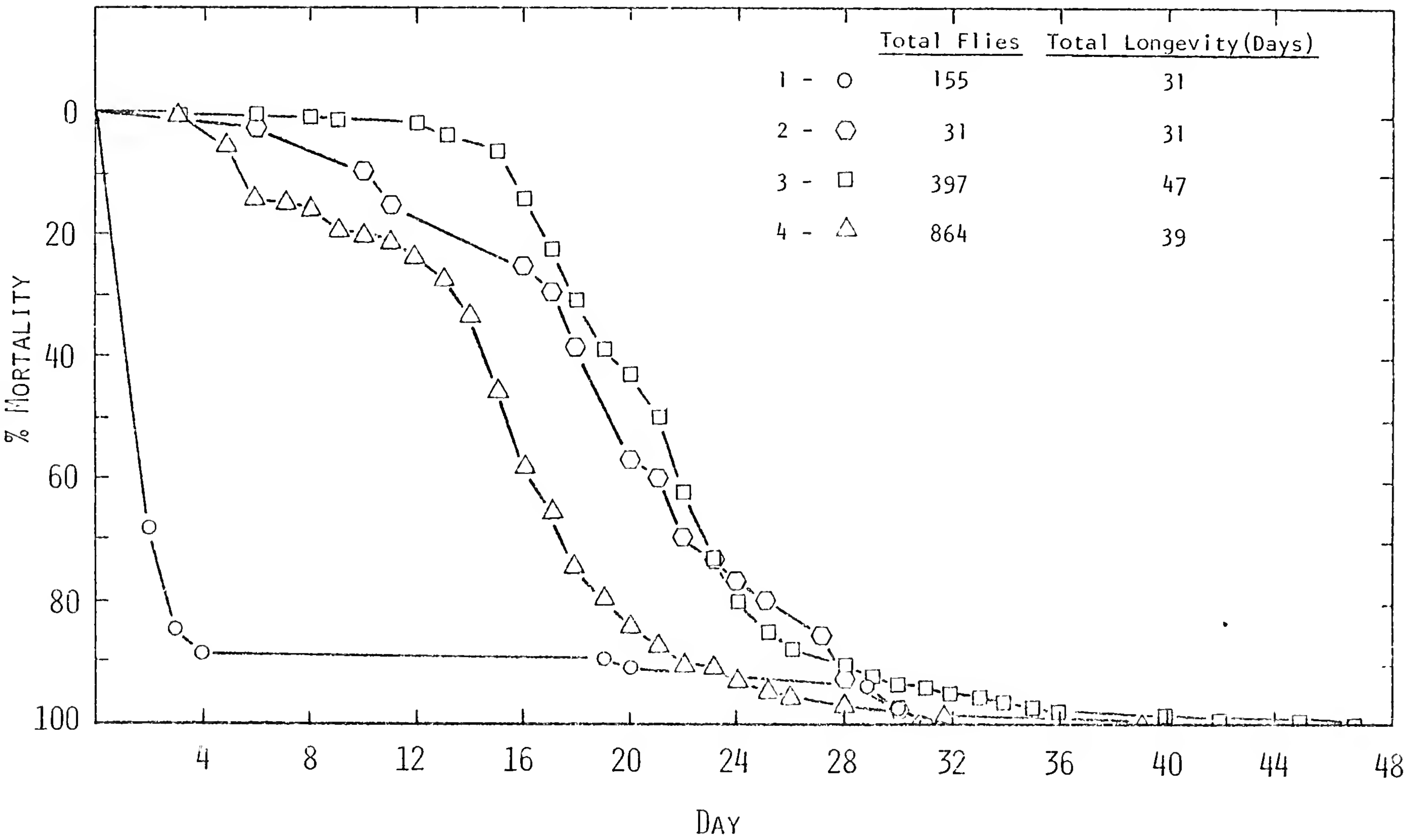


Figure 19. Graphic representation of the four *Ophyra aenescens* adult longevity studies.

Table 7. The number of pupae, the per cent pupation, the numerical and per cent emergence, and the larval viability of *Ophyra aenescens* reared in fortified and unfortified larval diets.

Rep	Number of Pupae	% Pupation	Numerical Emergence	% Emergence	Larval Viability(%)[a]
			Fortified Diet[b]		
1	41/60[c]	68.3	38/41[d]	92.7	63.3
2	34/60	56.1	30/34	88.2	60.0
3	33/60	55.0	31/33	93.9	51.7
4	29/60	48.3	27/29	93.1	45.0
	137/240	57.1	126/137	92.0	52.5
			Unfortified Diet[e]		
1	2/60	3.3	1/2	50.0	1.7
2	5/60	8.3	3/5	60.0	5.0
3	3/60	5.0	3/3	100.0	5.0
4	4/60	6.7	2/4	50.0	3.3
	14/240	5.8	9/14	65.7	3.8

[a] Equals $\frac{\text{number of adults}}{\text{number of pupae}} \times 100$

[b] CSMA™, horn fly dry mix (Greer, 1975)--960g wheat flour, 720g fish meal, 120g Na_2CO_3, 400g alfalfa meal; plus water.

[c] Represents the number of pupae that formed from 60 first-instar *O. aenescens* larvae.

[d] Represents the number of adults that eclosed from the respective number of pupae shown in column 2.

[e] CSMA™ (Ralston-Purina) plus water.

weights due to the diet (Table 8), although pupae from the unfortified diet were numerically lighter by an average of 0.3 mg. This indicated that adults from both treatments were the same size.

Larval density study. This study was designed to determine whether a minimum number of *O. aenescens* larvae must be present per unit area in order to have maximum development in a minimum amount of time. The fortified diet (Table 4) was placed loosely in 360-ml plastic cups to within 5 cm of the rim. Four population levels were used, each replicated four times. One cup with media served as a replication. To each cup in the respective treatment group, 5, 25, 50, and 100 first-instar *O. aenescens* larvae were added. Larvae were 12 to 18 hr old with an average length of 1.0 mm. After addition of larvae, cups were covered with screen and placed in the growth chamber. Pupal cases were separated from the medium after the emergence and death of the adults.

Results. The number of adults that emerged when the larval density averaged five larvae per 360 ml of medium was significantly lower than the number of adults that emerged from treatments with higher larval densities (Table 9). The treatment with the lowest larval density averaged 40.0% viability while treatments with 25, 50, and 100 larvae per 360 ml of medium averaged 91.0, 89.5, and 86.3% respectively. Results indicate the existance of an *O. aenescens* minimum population density threshold in between 5 and 25 larvae per 360 ml of medium. Additional research is needed to more accurately define the threshold.

Temperature of larval medium. Two aluminum pans, 25 cm in diameter and 8 cm deep, were filled to within 2 cm of the rims with the fortified larval diet (Table 4). One pan was seeded with ca. 500 eggs of *O. aenescens* and the other was used as a control. Pans were screen-

Table 8. Uneclosed pupal weights of *Ophyra aenescens* reared in fortified and unfortified diets.

Fortified Diet[a]				Unfortified Diet[b]			
Reps[c]				Reps[d]			
1	2	3	4	1	2	3	4
0.0114[e]	0.0100	0.0116	0.0156	0.0096	0.0130	0.0119	0.0134
0.0127	0.0135	0.0114	0.0140	0.0148	0.0121	0.0106	0.0128
0.0134	0.0123	0.0128	0.0111		0.0110	0.0131	0.0135
0.0102	0.0110	0.0105	0.0111				0.0106
0.0146	0.0098	0.0131	0.0133				
0.0126	0.0123	0.0123	0.0115				
0.0136	0.0123	0.0122	0.0130				
0.0143	0.0123	0.0125	0.0125				
0.0144	0.0100	0.0111	0.0175				
0.0125	0.0117	0.0144	0.0127				
0.1297	0.1152	0.1219	0.1323	0.0244	0.0361	0.0356	0.0503
			$\bar{x}$ = 0.0125				$\bar{x}$ = 0.0122

[a] CSMA™ (Ralston-Purina), horn fly dry mix (Greer, 1975)--960g wheat flour, 720g fish meal, 120g Na_2CO_3, 400g alfalfa meal; plus water.

[b] CSMA™ plus water

[c] Ten pupae from each beaker were chosen at random for weighing.

[d] All available pupae from each beaker were weighed.

[e] All pupal weights are in g.

Table 9. Emergence of adults of *Ophyra aenescens* when various numbers of first-instar larvae were reared in the same volume of growth medium.

Rep	5		25		50.		100	
	♂	♀	♂	♀	♂	♀	♂	♀
1	0	0	14	11	22	24	41	38
2	1	0	11	9	23	19	48	48
3	2	2	10	12	20	25	39	46
4	1	2	18	6	18	28	34	51
Σ by sex	4	4	53	38	83	96	162	183
Σ by trt	8		91		179		345	
$\bar{x}$	2.0[a]		22.8		44.8		86.3	
% viability	40.0		91.0		89.5		86.3	

[a] Indicates that this value is significantly different from others in the same row at $p < .05\%$.

covered and placed in the growth chamber. After 24 hr, the temperature of the medium in each pan was measured by use of a Taylor™ dial thermometer. Pans were divided into quarters and temperature measurements taken from the centers of the pans and from the centers of the quarters. The temperatures of the media were recorded every 24 hr from day 1, i.e. 24 hr after eggs were set, through day 7 of the larval development period. On day 7, pupae in the treated pan were floated and the experiment was terminated.

Results. The daily temperatures of larval media as influenced by *O. aenescens* larvae over the 7-day larval development period are shown in Table 10. The medium with larvae showed a daily increase in temperature, reaching 42.4 C on day 2 and then declining to 30.0 C on day 7. The medium without the larvae also peaked on day 2 at 33.7 C and declined to 28.7 C on day 7. Elevated temperatures in the medium with the larvae were attributed to larval interaction with the medium. Daily differences in temperature between the two pans were all significant ($p < .01$).

Competition study between *Ophyra aenescens* and *Musca domestica*. In this trial, 180-ml cups containing 90 ml of diet were used with four replications per treatment. First-instar larvae of both fly species were used to seed the diets. Experimental design and diet used are shown in Table 11. Treatments consisting of only one fly species were used as controls. Cups with larvae were covered with screen and placed in the growth chamber. Adults were allowed to eclose and die before they were counted.

Results. The results of the competition study between *O. aenescens* and *M. domestica* are shown in Table 12. In all treatments where house flies and dump flies were developing together, house fly mortality was

Table 10. Daily temperatures of larval media as influenced by *Ophyra aenescens* larvae over the 7-day larval development period.

Rep	Day 1	2	3	4	5	6	7
				Medium with Larvae			
1	45.6[a]	45.0	41.1	42.2	32.2	32.2	30.0
2	43.3	40.0	41.1	34.4	32.2	31.1	30.0
3	40.0	40.0	34.4	34.4	34.4	32.2	30.0
4	40.0	42.2	38.9	36.7	34.4	32.2	30.0
5	42.2	45.0	40.0	37.8	31.1	31.1	30.0
$\bar{x}$	42.2A	42.4A	39.1A	37.1A	32.9A	31.8A	30.0A
				Medium without Larvae			
1	33.3	31.7	30.0	32.2	28.9	27.8	30.0
2	28.9	35.6	32.2	28.9	31.1	30.0	28.9
3	30.0	33.3	31.1	26.7	28.9	27.8	28.9
4	32.2	34.4	30.0	26.7	27.8	27.8	27.8
5	31.1	33.3	31.1	27.8	28.9	27.8	27.8
$\bar{x}$	31.1B	33.7B	30.1B	28.5B	29.1B	28.2B	28.7B

[a] All values are in degrees centigrade.

Note: Means in the same column with uncommon letters are significantly different ($p < .01$).

Table 11. Experimental design of and the larval diet used in the competition study involving larvae of *Ophyra aenescens* and larvae of *Musca domestica*.

Treatment No.	Treatment
1	15[a] *O. aenescens* larvae[b]
2	15 *M. domestica* larvae
3	15 *O. aenescens* larvae + 15 *M. domestica* larvae
4	15 *O. aenescens* larvae + 30 *M. domestica* larvae
5	30 *M. domestica* larvae
6	15 *O. aenescens* larvae + 45 *M. domestica* larvae
7	45 *M. domestica* larvae

Larval Diet: 400 ml CSMA[TM]
100 ml horn fly dry mix[c]
275 ml water
ca. 700 ml medium

[a] Represents the number of larvae per replication with four replications per treatment.

[b] First-instar larvae of both species were used in the trial.

[c] 960g wheat flour, 720g fish meal, 120g Na_2CO_3, 400g alfalfa meal (Greer, 1975).

Table 12. Results of the competition study between *Ophyra aenescens* and *Musca domestica*.

Reps	1	2	3		4		5	6		7
	15 *O. aenescens*[a]	15 *M. domestica*[b]	15 *O. aenescens* +	15 *M. domestica*	15 *O. aenescens* +	30 *M. domestica*	30 *M. domestica*	15 *O. aenescens* +	45 *M. domestica*	45 *M. domestica*
1	15	15	15	0	15	0	30	15	4	42
2	15	15	15	0	13	0	30	15	3	45
3	15	15	15	0	15	0	28	15	5	43
4	14	14	15	0	15	2	30	15	5	45
$\bar{x}$	14.8[c]	14.8X	15.0	0.0X	14.5	0.5Y	29.5Y	15.0	4.3Z	43.8Z
% Mortality	1.7	1.7	0.0	100.0	3.3	98.3	1.7	0.0	90.6	2.8

[a] Indicates number of *O. aenescens* larvae used in each rep.

[b] Indicates number of *M. domestica* larvae used in each rep.

[c] Represents the mean number of viable individuals at the end of the experiment.

Note: Pairs of means having common letters are significantly different ($p < .05$).

significantly higher than in the control groups, while dump fly mortality remained unaffected. Fifteen *O. aenescens* larvae prevented the development of an equal number of *M. domestica* larvae, and produced mortality rates of 98.3 and 90.6% when reared with 30 and 45 house fly larvae respectively. House fly larvae that pupated in treatments 3, 4, and 6 reached the adult stage (Table 12). The remaining house fly larvae were prevented from reaching the pupal stage and died as larvae, but no dead larvae could be found in the growth media. This strongly suggested that the larvae of *O. aenescens* were preying upon the larvae of *M. domestica*.

Predation study with *Ophyra aenescens*. In this experiment, adapted after Peck (1969), the competition studies described above were modified and the availability of the larval medium as a food source was removed. Replications were 180-ml clear plastic cups containing 90 ml of vermiculite. Each treatment was replicated four times. Prior to addition of larvae, enough water was added to the vermiculite to make it damp but not soggy. Too much water forced larvae to the surface, not enough water caused them to stick to the sides of the cups. Water was added to all cups daily throughout the experiment to maintain the proper moisture level. The experimental design is shown in Table 13. In treatments 1, 2, and 3, 10 first-instar larvae of *O. aenescens* were placed in each cup of vermiculite. Then 25, 100, and 200 first-instar house fly larvae were added daily to each cup in the respective treatment group until pupae were noted in the cups. Treatments 4 through 8 were used as controls. Cups were covered with screen and kept in the growth chamber except when larvae were being added. At the end of the experiment, flies were allowed to eclose and die before they were counted.

Table 13. Experimental design of *Ophyra aenescens* predation study.

Treatment No.	Treatment	
	Larvae added initially	Larvae added daily
1	10[a] *O. aenescens*[b]	25[a] *M. domestica*
2	10 *O. aenescens*	100 *M. domestica*
3	10 *O. aenescens*	200 *M. domestica*
4	25 *M. domestica*	---
5	100 *M. domestica*	---
6	200 *M. domestica*	---
7	100 *O. aenescens*	---

[a] Represents the number of larvae per replication with four replications per treatment.

[b] Larvae used were first instars of both fly species.

Results. The results of the predation study are shown in Table 14. The mortality rate of *O. aenescens* larvae was reduced significantly in all cases by the daily addition of house fly larvae. Control mortalities were 100% in all cases, indicating that neither fly species is cannibalistic and that in order to survive, larvae of *O. aenescens* were preying upon larvae of *M. domestica*. This was, in fact, the case and it was observed on numerous occasions.

Ophyra larvae never fed for extended periods on a single house fly larva. After killing and feeding briefly on one, they moved on in search of another. When a house fly larva was killed by *Ophyra*, other house fly larvae were attracted to the site and began feeding on the dead larva. The clustering of house fly larvae aided *Ophyra* larvae in their search for prey. This hypothesis was corroborated by fitting the data in Table 14 to a binomial regression model and plotting the graph in Figure 20. The goodness of fit was highly significant ($p < 0.0001$). Larvae of *O. aenescens* can destroy more than 20 first-instar house fly larvae per day during the larval developmental period, but further testing is necessary to arrive at a more accurate number. On at least one occasion, a third-instar larva of *O. aenescens* was seen feeding on a pupa of *M. domestica* that had not completed the tanning process.

Since *Ophyra* adults were allowed to emerge prior to processing, no pupal weights were available to check for differences in the sizes of the adults. An attempt was made to find a correlation between eclosed pupal weight and adult weight, but results were negative. Adults raised in vermiculite did not appear to be smaller than adults from larvae raised in the usual manner.

Table 14. Results of *Ophyra aenescens* predation studies.

Reps	Treatments 1		2		3		4	5	6	7	8
	10 *O. aenescens*[a]	+ 25 *M. domestica*[b]	10 *O. aenescens*[a]	+ 100 *M. domestica*[b]	10 *O. aenescens*[a]	+ 200 *M. domestica*[b]	10 *O. aenescens*[a]	25 *M. domestica*[c]	100 *M. domestica*[c]	200 *M. domestica*[c]	100 *O. aenescens*[a]
1	1	0	6	0	9	0	0	0	0	0	0
2	2	0	6	0	9	0	0	0	0	0	0
3	3	0	5	0	8	0	0	0	0	0	0
4	1	0	8	0	7	0	0	0	0	0	0
$\bar{x}$	1.8A[d]	0.0	6.3A	0.0	8.3A	0.0	0.0A	0.0	0.0	0.0	0.0
% Mortality	82.5	100.0	37.5	100.0	17.5	100.0	100.0	100.0	100.0	100.0	100.0

[a] Indicates the number of *O. aenescens* larvae initially placed in each rep of treatments 1, 2, 3, 4, and 8.

[b] Represents the number of *M. domestica* larvae added daily to each rep of treatments 1, 2, and 3.

[c] Indicates the number of *M. domestica* larvae initially placed in each rep of treatments 5, 6, and 7.

[d] Represents the mean number of viable individuals at the end of the experiment.

Note: Means having common letters are significantly different ($p < .05$).

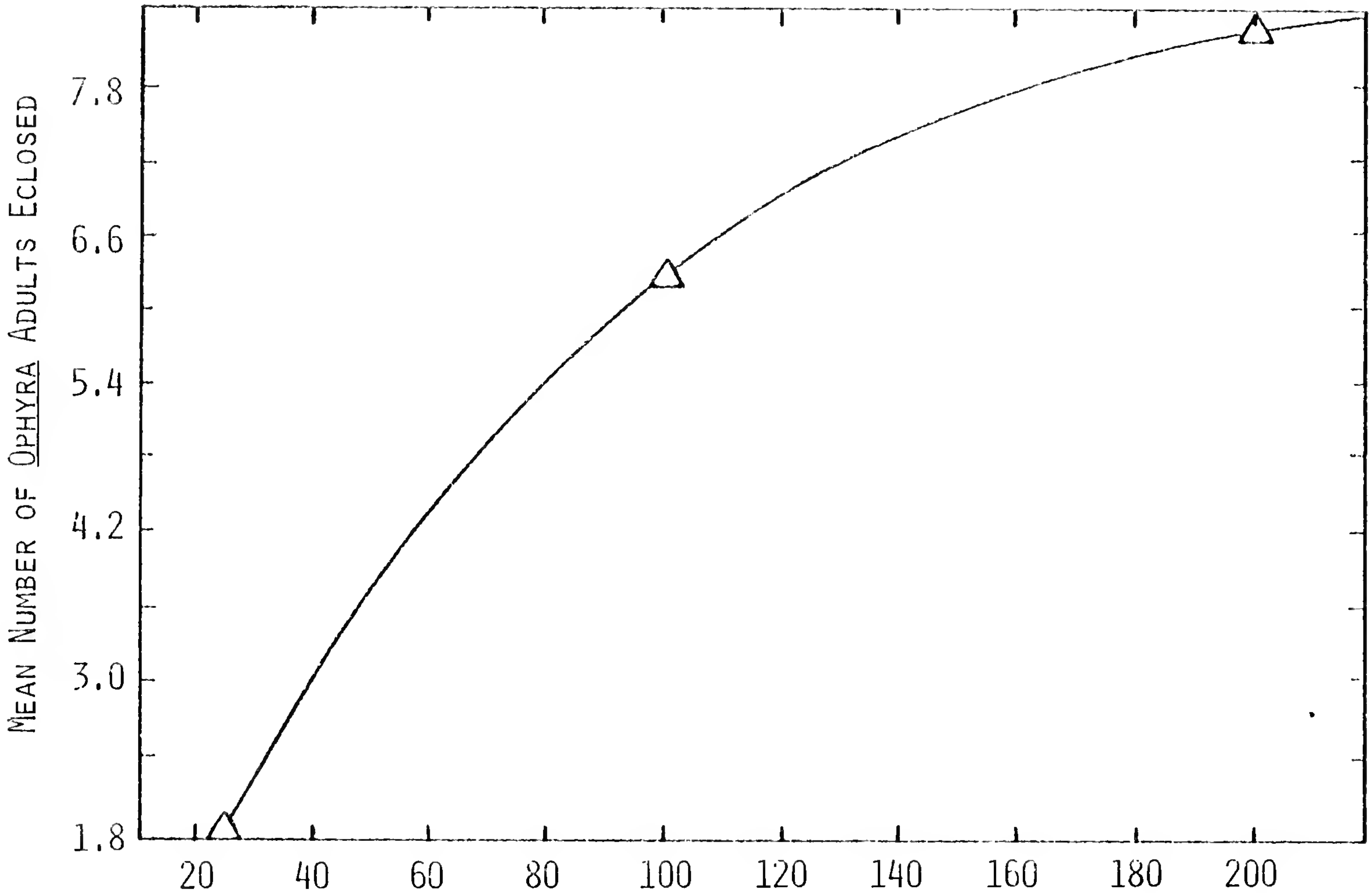


Figure 20. Regression curve for data from *Ophyra aenescens* predation study.

Comparisons of cephaloskeletons of *Ophyra aenescens* and *Musca domestica*. It was determined by Keilin and Tate (1930) that a correlation exists between certain anatomical structures of fly larvae and their modes of life. In order to gain further knowledge of the feeding habits of *O. aenescens* and *M. domestica* larvae through the use of anatomical evidence, the cephaloskeletons of third-instar larvae were dissected and mounted on microscope slides in balsam.

Areas of the cephaloskeletons compared (Figure 21) were the ventral surface of the basal sclerite(B.) and the oral sclerite(O.).

Results. The basal sclerites of *M. domestica* and *O. aenescens* are shown in Figures 22 and 23 respectively. Note the presence of longitudinal ridges(L.R.) in the basal sclerites of both species. In Figure 22, the salivary duct(S.D.) can be seen where it passes into the intermediate sclerite(I.).

The oral sclerites of *M. domestica* are shown in Figure 24. They consist of two lateral hooks(L.H.), nearly uniform in length, joined ventrally by a median ventral arc(M.V.A.). Both the median ventral arc and the lateral hooks are shrouded by the remains of the cuticle. Oral grooves(O.G.) leading to the oral aperature can be clearly seen.

The oral sclerites of *O. aenescens* (Figure 25) consist of two lateral hooks of unequal lengths. The left hook, shown protruding above the right hook, is the shorter of the two. It fits into a groove on the right hook which enables both hooks to work together. Both hooks are joined ventrally by a median ventral arc.

In addition to the oral sclerites, accessory oral sclerites are present in the bucco-pharyngeal armature of *O. aenescens* larvae (Figure 25). Beneath the lateral hooks are two oral bars(O.B.). The anterio-

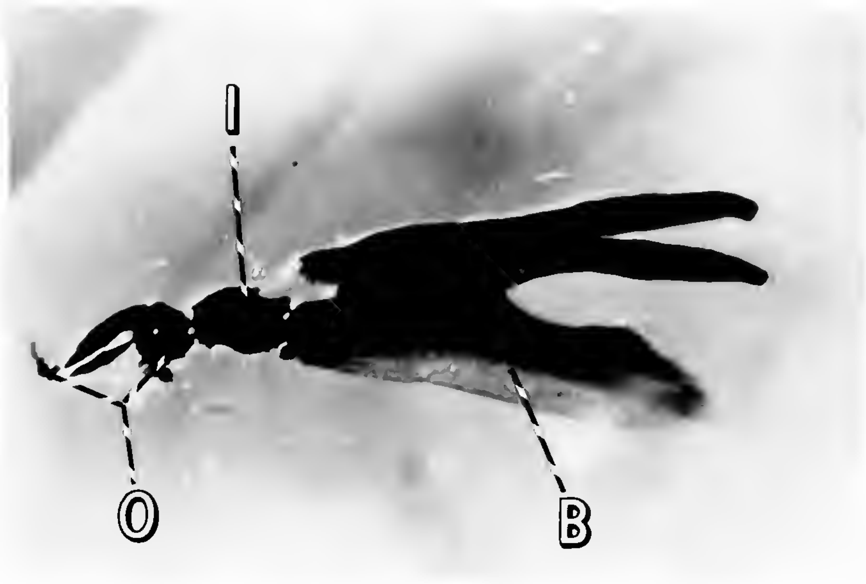


Figure 21. Areas on the cephaloskeleton of *Ophyra aenescens* compared with those of *Musca domestica*.

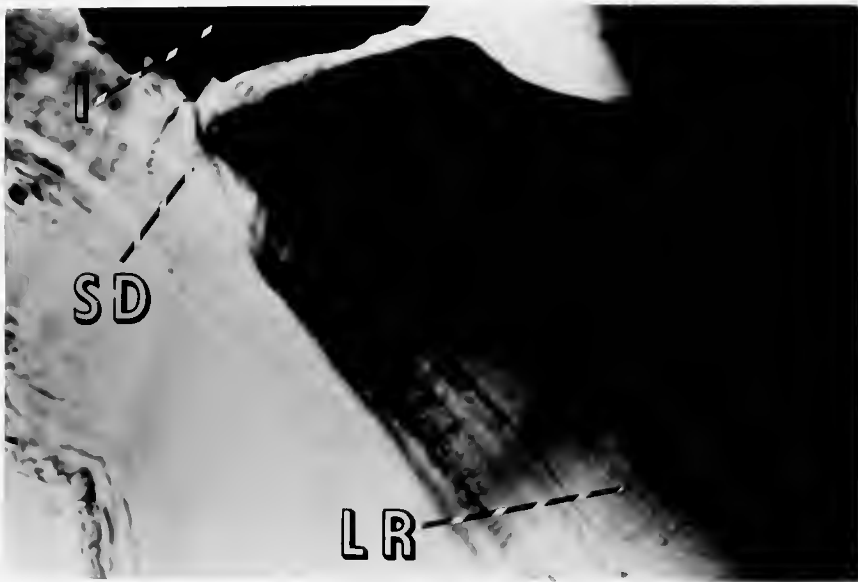


Figure 22. Basal sclerite of *Musca domestica* showing longitudinal ridges(L.R.) and salivary duct(S.D.).

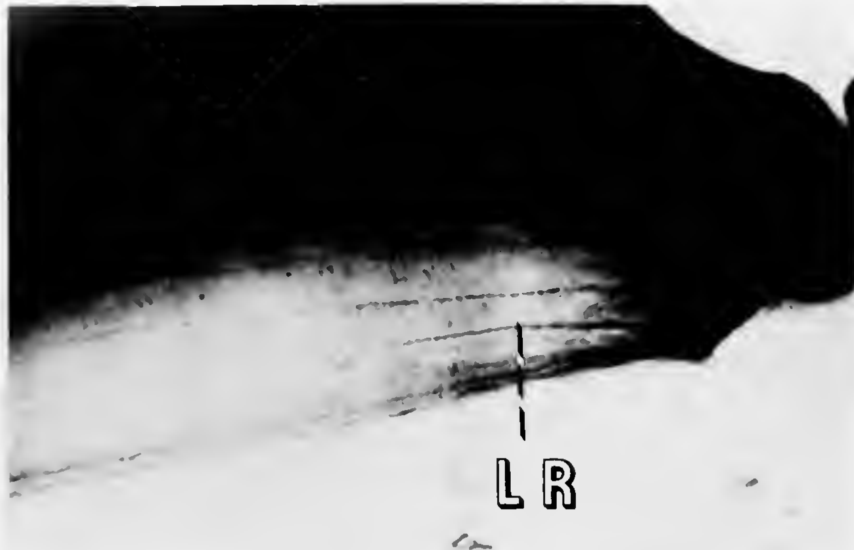


Figure 23. Basal sclerite of *Ophyra aenescens* showing longitudinal ridges(L.R.).

Figure 24. Oral sclerites of *Musca domestica* showing the intermediate sclerite(I.), the lateral hooks (L.H.), the median ventral arc(M.V.A.), and the oral grooves(O.G.).

Figure 25. Oral sclerites of *Ophyra aenescens* showing the lateral hooks(L.H.), the median ventral arc (M.V.A.), the oral bars(O.B.), the anterior ribbon(A.R.), and the cutaneous teeth(C.T.).

dorsal borders of the oral bars articulate with two additional sclerites that join anterior to the tips of the lateral hooks to form the anterior ribbon(A.R.). Beneath each oral bar is a row of cutaneous teeth(C.T.), but only one row is visible in Figure 25.

The longitudinal ridges in the basal sclerites of both species are indicative of saprophagous behavior and the accessory oral sclerites and unequal lateral hooks of *O. aenescens* are indicative of predaceous behavior (Keilin and Tate, 1930). The combination of characteristics, i.e. the longitudinal ridges and the oral sclerites, indicates that larvae of *O. aenescens* can live either in a saprophagous manner or they can be carnivorous.

Competition Studies with *Hermetia illucens*

Competition studies were performed in the laboratory to gain knowledge of interactions that could occur in the field if the habitats of *H. illucens* and those of three other flies, *M. domestica*, *O. aenescens*, and *S. robusta*, overlapped. Experimental designs and larval diets are shown in Tables 15 and 16. Larvae were grown in 360-ml plastic cups containing 200 ml of diet. Each cup represented a replication, and each treatment was replicated four times. Treatments consisting of only one species of fly were used as controls. After larvae or eggs were added, cups were covered with screen and placed in the growth chamber.

Trials were terminated when adults of *M. domestica*, *O. aenescens*, and *S. robusta* emerged in their respective control groups. Viable larvae of *H. illucens* were counted when experiments ended. Since the soldier fly larvae tended to stay at the bottom of the cups, they were easily located. The remaining medium was floated to separate adults from pupae of the competing fly species when applicable.

Table 15. Experimental designs of and the larval diets used in the competition studies involving larvae of *Hermetia illucens* vs. larvae of *Musca domestica* and *Ophyra aenescens*.

Treatment No.	Treatment
1	10[a] *O. aenescens* larvae[b]
2	25 *O. aenescens* larvae
3	10 *H. illucens* larvae
4	25 *H. illucens* larvae
5	10 *O. aenescens* larvae + 10 *H. illucens* larvae
6	25 *O. aenescens* larvae + 10 *H. illucens* larvae
7	10 *O. aenescens* larvae + 25 *H. illucens* larvae
8	10 *M. domestica* eggs
9	25 *M. domestica* eggs
10	50 *M. domestica* eggs
11	10 *H. illucens* larvae
12	25 *H. illucens* larvae
13	50 *H. illucens* larvae
14	10 *M. domestica* eggs + 10 *H. illucens* larvae
15	25 *M. domestica* eggs + 10 *H. illucens* larvae
16	25 *M. domestica* eggs + 25 *H. illucens* larvae
17	25 *M. domestica* eggs + 50 *H. illucens* larvae
18	50 *M. domestica* eggs + 10 *H. illucens* larvae

Larval Diet: 5 parts CSMA™ + 3 parts water

[a] Represents the number of larvae or eggs per replication with four replications per treatment.

[b] Larvae of *O. aenescens* were first instars; larvae of *H. illucens* were from 1 to 1.5 cm in length.

Table 16. Experimental design of and the larval diet used in the competition study involving larvae of *Hermetia illucens* and larvae of *Sarcophaga robusta*.

Treatment No.	Treatment
1	10[a] *H. illucens* larvae[b]
2	10 *S. robusta* larvae
3	25 *S. robusta* larvae
4	10 *S. robusta* larvae + 10 *H. illucens* larvae
5	25 *S. robusta* larvae + 10 *H. illucens* larvae
	Larval Diet: 200 ml CSMA™
	400 ml fish meal
	440 ml water
	ca. 800 ml very moist medium

[a] Represents the number of larvae per replication with four replications per treatment.

[b] Larvae of *S. robusta* were first instars; larvae of *H. illucens* were from 1 to 1.5 cm in length.

Results of studies with *Hermetia illucens* and *Ophyra aenescens*. When the number of *O. aenescens* larvae per replication was equal to or less than the number of *H. illucens* larvae, the larval mortality of *O. aenescens* increased significantly, but the mortality of *H. illucens* larvae was not significantly affected (Table 17). When larvae of *O. aenescens* outnumbered those of *H. illucens*, the larval mortality of *O. aenescens* was not affected, but the mortality of *H. illucens* was increased by a nonsignificant amount. This indicates that the larval development of *O. aenescens* can be suppressed, but not prevented, by the presence of *H. illucens* in the larval medium. This has not been demonstrated in the field.

Results of studies with *Hermetia illucens* and *Musca domestica*. Larvae of *H. illucens* were not able to completely prevent the development of larvae of *M. domestica* when the two species were reared together (Table 18). The mortality of *M. domestica* was increased to 90.0 and 86.0% when 25 larvae were reared with 25 and 50 larvae of *H. illucens* respectively. These were significant increases over control mortality.

Control mortality was high because house fly eggs were used instead of first-instar larvae, but at the time, additional larvae of *H. illucens* were not available and the experiment could not be repeated.

The ability of these two species to develop in the same containers during this experiment, but not during others, may have been due to variables such as container size, number of larvae per unit area of medium, and moisture content of the larval medium. Larvae of *H. illucens* tended to remain on the bottoms of the cups of medium, perhaps the wettest location in the cups. House fly larvae tended to remain in the upper two thirds of the cups of medium, perhaps seeking drier areas than

Table 17. Results of the competition study between *Hermetia illucens* and *Ophyra aenescens*.

	Treatments									
Reps	1	2	3	4	5		6		7	
	10 *O. aenescens*[a]	25 *O. aenescens*[b]	10 *H. illucens*	25 *H. illucens*	10 *O. aenescens* +	10 *H. illucens*	25 *O. aenescens* +	10 *H. illucens*	10 *O. aenescens* +	25 *H. illucens*
1	10	25	10	25	7	9	24	10	7	25
2	7	23	10	24	7	10	19	9	6	25
3	9	20	10	21	6	10	24	10	7	25
4	8	18	10	23	8	10	19	9	8	25
$\bar{x}$	8.5XY[c]	21.5	10.0	23.3	7.0Y	9.8	21.5	9.5	7.0Y	25.0
% Mortality	15.0	14.0	0.0	7.0	30.0	2.5	14.0	5.0	30.0	0.0

[a] Indicates number of *O. aenescens* larvae used in each rep.

[b] Indicates number of *H. illucens* larvae used in each rep.

[c] Represents mean number of viable individuals at the end of the experiment.

Note: Means having common letters are significantly different ($p < .05$).

Table 18. Results of the competition study between *Hermetia illucens* and *Musca domestica*.

Reps	Treatments 8	9	10	11	12	13	14	
	10 *M. domestica*[a]	25 *M. domestica*	50 *M. domestica*	10 *H. illucens*[b]	25 *H. illucens*	50 *H. illucens*	10 *M. domestica* +	10 *H. illucens*
1	8[c]	7	21	10	25	49	2	8
2	7	13	9	10	24	48	8	10
3	4	3	18	10	23	50	2	10
4	6	15	18	10	21	41	7	9
$\bar{x}$	6.3[c]	9.6YZ	16.5	10.0	23.3	47.0	4.8	9.3
% Mortality	37.5	62.0YZ	67.0	0.0	7.0	6.0	52.5	7.5

Table 18. Continued.

Reps	Treatments 15		16		17		18	
	25 *M. domestica* +	10 *H. illucens*	25 *M. domestica* +	25 *H. illucens*	25 *M. domestica* +	50 *H. illucens*	50 *M. domestica* +	10 *H. illucens*
1	12	10	3	24	5	47	19	10
2	8	10	1	23	5	48	13	10
3	6	10	3	25	2	48	21	10
4	6	10	3	25	2	48	11	10
$\bar{x}$	8.0	10.0	2.5Y	24.3	3.5Z	47.8	16.0	10.0
% Mortality	68.0	0.0	90.0	3.0	86.0	4.5	68.0	0.0

[a] Indicates the number of *M. domestica* eggs used in each rep.

[b] Indicates the number of *H. illucens* larvae used in each rep.

[c] Represents the mean number of viable individuals at the end of the experiment.

Note: Pairs of means having common letters are significantly different ($p < .05$).

H. illucens. Thus, the two species developed in separate locations in the same cups with little chance of coming in contact.

A situation such as this was seen in the field at the tilling site (Figure 31). Chips had been added to very wet manure which, when tilled, was ca. 29 cm above the level of the walks. A definite moisture gradient existed in the chips-manure mixture. Deep in the manure, at the level of the walks, there was a large population of soldier flies. Near the top of the manure-chips mixture, 2 to 4 cm beneath the surface, was a large house fly population. The populations of the two species were separated vertically by 25 to 27 cm and neither could be detected without digging into the manure. This was an unusual situation created by the depth of the manure-chips mixture and the moisture gradient within.

Results of studies with *Hermetia illucens* and *Sarcophaga robusta*. In treatments 4 and 5, where both species were reared together, the mortality of *S. robusta* larvae was increased significantly, but the mortality of *H. illucens* was not significantly affected (Table 19).

Since the larvae of *S. robusta* require only 3 days to complete their larval development, the reduction in their population by *H. illucens* was not expected. Because larvae of *S. robusta* and *H. illucens* prefer media similar in moisture content, it is possible that the bottoms of the cups of medium were the sites preferred by both fly species. *Sarcophaga robusta* may have been unable to compete successfully with *H. illucens* for the preferred sites and was displaced in numbers reflected by the increases in mortality shown in Table 19.

Table 19. Results of competition study between *Hermetia illucens* and *Sarcophaga robusta*.

Reps	Treatments 1	2	3	4		5	
	10 *H. illucens*[a]	10 *S. robusta*[b]	25 *S. robusta*	10 *S. robusta* +	10 *H. illucens*	25 *S. robusta* +	10 *H. illucens*
1	10[c]	7	24	4	10	12	10
2	10	8	21	2	9	12	10
3	10	8	23	2	10	12	10
4	10	8	19	2	10	8	10
Σ	40	31	87	10	39	44	40
$\bar{x}$	10.0	7.8X	21.8Y	2.5X	9.8	11.0Y	10.0
% Mortality	0.0	22.5	13.0	75.0	2.5	56.0	0.0

[a] Indicates number of *H. illucens* larvae used in each replication.

[b] Indicates number of *S. robusta* larvae used in each replication.

[c] Represents the number of viable individuals at the end of the experiment.

Note: Pairs of means($\bar{x}$) having common letters are significantly different ($p < .05$).

Insect Growth Regulators (IGR's) and Organophosphorus Larvicides

Feed-through studies with dimilin and methoprene. Methoprene (Zoecon™ ZR-515 10% tech.) and dimilin (Thompson-Hayward TH 6040 25% tech.) were added to poultry feed to produce diets of the following concentrations:

IGR	Amount added to 136.4 kg of basal layer diet (g)	Concentration of treatment diet (ppm)
TH 6040	5.44	10
TH 6040	0.54	1
ZR-515	13.60	10
ZR-515	1.36	1

Diets were fed to treatment groups of 44 week-old Babcock B-300 laying hens randomized in range houses measuring 2.4 by 4.6 m (Figure 26). The 10-ppm diets were fed for 107 days after which the 1-ppm diets were fed for 14 days. One week elapsed between the change of IGR treatment levels.

Manure was bioassayed three times while 10-ppm diets were fed, and twice while 1-ppm diets were being fed. Samples were seeded with eggs of *M. domestica*. Pupal traps were set twice while the 10-ppm diets were being fed.

Results. Feed consumption of hens fed methoprene and dimilin is shown in Table 20. At both dietary levels, birds eating the dimilin diets consumed feed in amounts that were significantly greater than those consumed by birds in the control groups. Birds eating methoprene diets consumed less feed that the birds in their respective control groups, but the differences were not significant.

During sampling period 9 (Table 20), hens were inadvertently allowed to run out of feed, which accounts for the consumption level of only 77.18 g/bird per day.

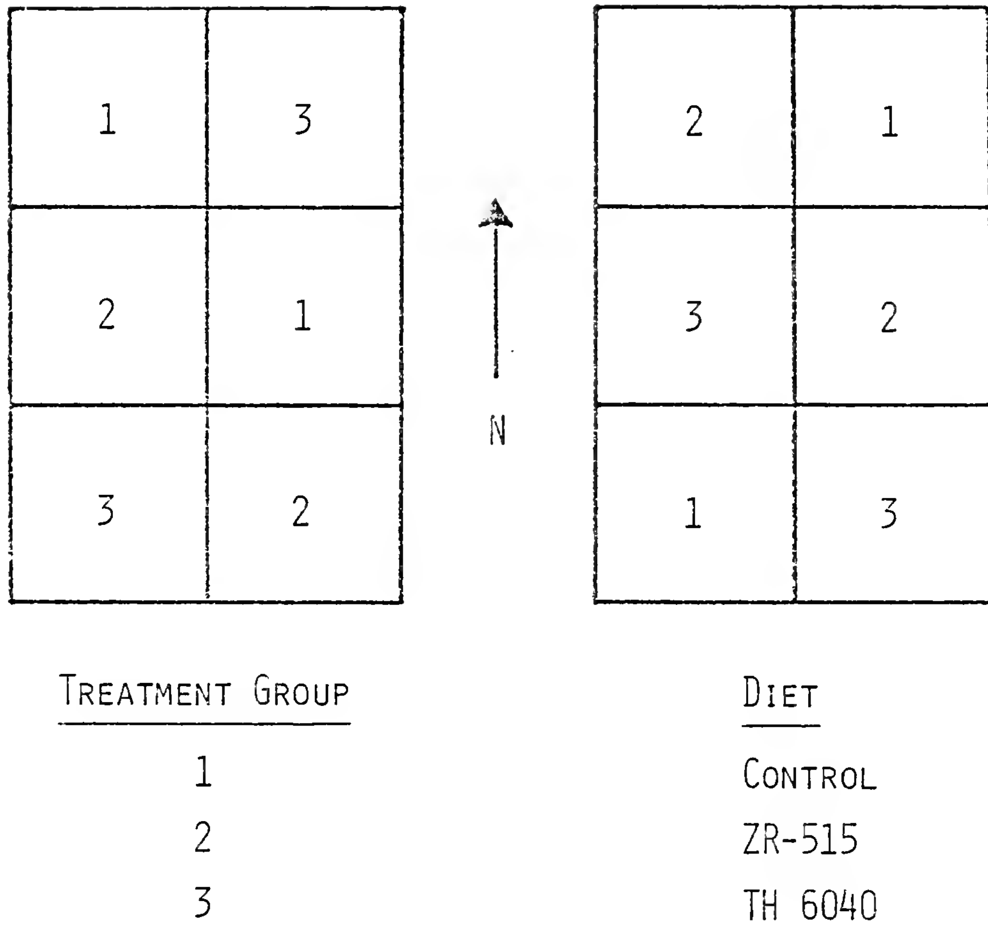


Figure 26. Assignment of diets containing ZR-515 and TH 6040 to treatment groups in range houses.

Table 20. Average daily feed consumption (g/bird per day) of hens fed diets containing TH 6040 and ZR-515 at 1 and 10 ppm.

	Growth Regulator		
Period[a]	ZR-515	TH 6040	Control
	Dietary Level -- 10 ppm		
1	106.69	108.96	104.42
2	111.23	113.50	104.42
3	102.15	118.04	102.15
4	104.42	113.50	97.61
5	113.50	122.58	111.23
6	118.04	127.12	118.04
7	113.50	131.66	111.23
8	104.42	122.58	108.96
9	77.18	127.12	106.69
$\bar{x}$	105.68A	120.56B	107.19A
	Dietary Level -- 1 ppm		
1	106.69	111.23	106.69
2	106.69	124.85	111.23
$\bar{x}$	106.69C	118.04D	108.96C

[a] Average daily feed consumption was calculated at the end of each 12-day period.

Note: Mean values in the same row without common letters are significantly different ($p < 0.01$) according to Duncan's Multiple Range Test.

There were no significant differences in egg production due to diet at the 10-ppm level (Table 21), but the birds consuming the dimilin diet were laying at a numerically higher level than the other two groups.

At the 1-ppm level, hens consuming the methoprene diet were laying significantly fewer eggs than the hens in the control groups. This was attributed to the hens being allowed to run out of feed as mentioned above. There were no significant differences in the production levels of the dimilin and control groups (Table 21), but the birds in the dimilin group maintained a higher numerical production level.

Results from the manure bioassay are shown in Table 22. At both dietary IGR levels, fly mortality was significantly greater than mortality in the control groups. At the 10-ppm level, methoprene and dimilin produced fly mortalities of 85.00 and 74.17% respectively. At 1 ppm, methoprene and dimilin produced fly mortalities of only 17.5% which is too low to be of any practical value.

The data from the pupal traps were nonconclusive (Table 23) since mortality in all three groups was nonsignificant. In the control group, 46.48% of the pupae failed to eclose for some unexplainable reason. The wood chips may have become contaminated with a toxic substance prior to use in the pupal traps. When Abbott's formula was used to correct the mortalities, only 16.11 and 12.80% of the mortalities was due to methoprene and dimilin respectively. These low mortalities may have been due to the rapid breakdown of the IGR's plus the lack of larval contact with the IGR's in the manure pack.

<u>Topical application of methoprene.</u> Experiment 1 -- The experimental design used at the tilling site is shown in Figure 27. Blocks 2

Table 21. Average hen-day production(%) of hens fed diets containing TH 6040 and ZR-515 at 1 and 10 ppm.

Period[a]	Growth Regulator: ZR-515	TH 6040	Control
	Dietary Level -- 10 ppm		
1	51.85	50.39	52.41
2	41.49	36.95	31.73
3	53.10	52.76	46.07
4	61.66	73.80	66.14
5	37.36	65.88	65.17
$\bar{x}$	49.09A	55.96A	52.30A
	Dietary Level -- 1 ppm		
1	26.94B	62.56C	57.87C

[a] Average hen-day production was calculated at the end of each 21-day period.

Note: Mean values in the same row without common letters are significantly different ($p < 0.01$) according to Duncan's Multiple Range Test.

Table 22. Bioassay of manure from hens fed diets containing TH 6040 and ZR-515 at 1 and 10 ppm.

Sample No.	Growth Regulator ZR-515	TH 6040	Control
	Dietary Level -- 10 ppm		
1 - 30[a]	1/10[b]	6/10	10/10
2 - 30	1/10	7/10	8/10
3 - 30	2/10	7/10	10/10
4 - 30	1/10	6/10	10/10
5 - 92	1/10	1/10	10/10
6 - 92	1/10	2/10	10/10
7 - 92	0/10	1/10	9/10
8 - 92	1/10	1/10	10/10
9 - 107	3/10	0/10	9/10
10 - 107	3/10	0/10	10/10
11 - 107	3/10	0/10	10/10
12 - 107	2/10	0/10	10/10
Σ	18/120	31/120	116/120
$\bar{x}$	1.50A	2.58A	9.67B
% Mortality	85.00	74.14	3.33
	Dietary Level -- 1 ppm		
1 - 11	10/10	8/10	10/10
2 - 11	10/10	6/10	9/10
3 - 11	10/10	9/10	10/10
4 - 11	9/10	7/10	10/10
5 - 14	6/10	9/10	10/10

Table 22. Continued.

Sample No.	Growth Regulator		
	ZR-515	TH 6040	Control
	Dietary Level -- 1 ppm		
6 - 14	7/10	8/10	9/10
7 - 14	8/10	10/10	8/10
8 - 14	6/10	9/10	8/10
Σ	66/80	66/80	74/80
$\bar{x}$	8.25C	8.25C	9.25C
% Mortality	17.50	17.50	7.50

[a] The numbers to the right of the hyphen indicate the day of the experiment on which the sample was collected, e.g. sample 1 was collected on day 30 of the experiment.

[b] Represents the number of adult house flies that formed when 10 eggs were set.

Note: Mean values in the same row without common letters are significantly different ($p < 0.05$).

Table 23. Data from pupal traps set in manure from hens fed diets containing TH 6040 and ZR-515 at 10 ppm.

	Growth Regulator		
Replication	ZR-515	TH 6040	Control
1	1/6[a]	6/16	7/13
2	10/20	11/27	16/28
3	2/8	3/11	12/21
4	9/15	15/21	3/9
Σ	22/49	35/75	38/71
% Mortality	55.10	53.33	46.48

[a] Represents the number of pupae that eclosed over the number of pupae found in the trap.

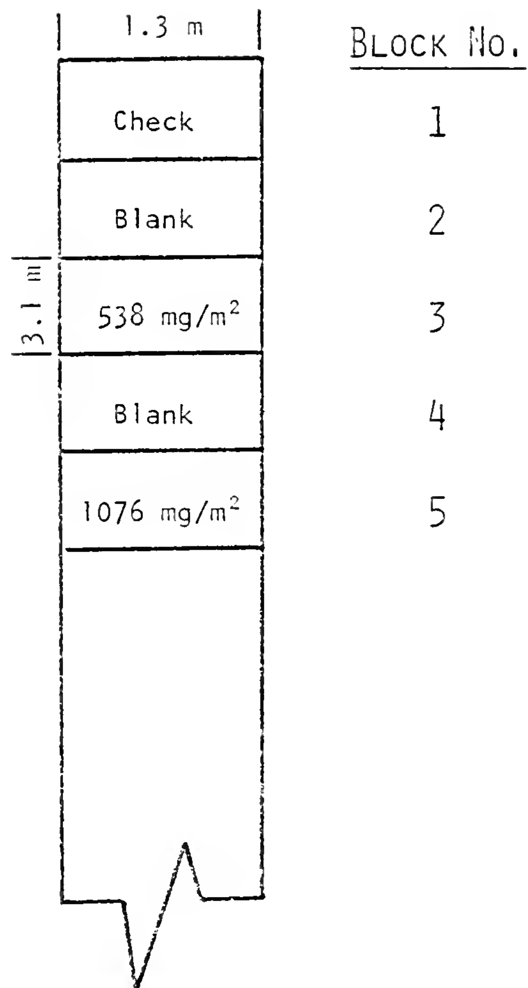


Figure 27. Experimental design for testing the effects of ZR-515 sand granules on larval populations of *Musca domestica* in the manure collection area of a poultry house.

and 4 were left untreated to prevent overlap of treatments. Methoprene (Zoecon 0.4% tech.) sand granules were applied at rates of 538 and 1076 mg/m^2. The weights of granules applied to blocks 3 and 5 (Figure 27) were 2.08 and 4.17 g respectively.

Immediately after treatments were applied and 3 days later, samples were collected for laboratory bioassay. Each sample was seeded with 25 eggs of *M. domestica*.

Results. Mortality produced by methoprene sand granules at 1076 mg/m^2 was significantly higher than the mortalities in the other two groups (Table 24). When corrected by Abbott's formula, mortalities produced by the 1076 and 538 mg/m^2 treatments were 94.3 and 42.9% respectively.

In the second group of samples, there were significant differences in the mortalities produced by all three treatments (Table 24). When corrected by Abbott's formula, mortalities produced by the 1076 and 538 mg/m^2 treatments were 95.82 and 62.91% respectively.

Results indicate that methoprene sand granules are effective for at least 3 days post-treatment. When the granules are applied at 1076 mg/m^2, fly mortality of more than 90% can be expected. Field tests, however, may not produce similar results as indicated by the feed-through trials.

Few pupae were formed in the sample replicates of manure treated with 1076 mg/m^2 of methoprene, because at this high concentration, the larvae were killed.

Experiment 2 -- At the University of Florida Poultry Science Farm, methoprene (Zoecon 0.4% tech.) sand granules were sprinkled on manure beneath two treatment blocks of 10 hens caged in pairs. Weights of the

Table 24. Number of pupae, number of pupae eclosed, and per cent mortality when larvae of *Musca domestica* were reared in poultry manure containing two levels of methoprene sand granules.

Sample No.	1076 mg/m² No. Pupae	1076 mg/m² No. Eclosed	538 mg/m² No. Pupae	538 mg/m² No. Eclosed	Check No. Pupae	Check No. Eclosed
1	0[a]	0	7	4	5	3
	0	0	18	4	10	3
	0	0	16	3	6	6
	7	2	14	9	25	23
Σ	7	2	55	20	55	35
$\bar{x}$		0.5A		5.0B		8.8B
% Mortality[b]		98.0		80.0		65.0
2	24	1	22	16	21	21
	0	0	14	3	16	16
	0	0	21	4	18	15
	0	0	30	4	28	27
Σ	24	1	87	27	83	79
$\bar{x}$		0.3C		6.8D		19.8E
% Mortality		99.0		78.0		24.0

Treatments: 1076 mg/m², 538 mg/m², Check.

[a] Represents the number of pupae formed from 25 house fly eggs.

[b] Equals the number of eclosed pupae subtracted from 100.

Note: Mean values in the same row without common letters are significantly different ($p < 0.01$).

granules used for the 1076 and 538 mg/m^2 treatments were 0.57 and 0.29 g respectively. Samples were collected as for experiment 1 above.

Results. In the first group of samples, house fly mortalities in groups treated with methoprene were significantly higher than the check mortalities (Table 25). When corrected by Abbott's formula, mortalities produced by the 1076 and 538 mg/m^2 treatments were 93.9 and 97.0% respectively.

In the second group of samples, mortalities in groups treated with methoprene were significantly higher than the check mortalities, despite the high check mortalities. When corrected by Abbott's formula, mortalities produced by the 1076 and 538 mg/m^2 treatments were 70.6 and 88.2% respectively.

Results indicate that methoprene sand granules are effective for at least 3 days post-treatment. When the granules are applied at 1076 mg/m^2, mortalities greater than 90% can be expected, at least on the first day. Results from field tests may differ from laboratory tests as previously stated. It cannot be explained why the lower concentration of methoprene produced numerically higher mortalities in both sample groups. Lack of pupae formation in the 538 mg/m^2 treatment in the second group of samples indicated the presence of a high level of methoprene, possibly due to collection of manure from an area where nonuniform application occurred.

High check mortalities in both experiments were attributed to the use of house fly eggs in the manure samples. House fly eggs hatch better in wet media and media in this experiment were somewhat dry.

Laboratory studies with CGA 72662. CGA 72662 (Ciba-Geigy™ 10% soluble concentrate) was added to the diets of six species of flies. The fly species and diets utilized are shown in Table 26.

Table 25. Number of pupae, number of pupae eclosed, and per cent mortality when larvae of *Musca domestica* were reared in poultry manure containing two levels of methoprene sand granules that were applied at the University of Florida Poultry Science Farm.

	Treatments					
	1076 mg/m^2		538 mg/m^2		Check	
Sample No.	No. Pupae	No. Eclosed	No. Pupae	No. Eclosed	No. Pupae	No. Eclosed
1	14[a]	1	18	0	21	21
	9	1	24	1	12	12
	11	0	22	1	14	14
	12	2	20	0	19	19
Σ	46	4	84	2	66	66
$\bar{x}$		1.0A		0.5A		16.5B
% Mortality[b]		96.0		98.0		34.0
2	5	5	1	1	14	13
	12	0	0	0	4	4
	9	3	2	2	10	10
	8	2	1	1	8	7
Σ	34	10	4	4	36	34
$\bar{x}$		2.5C		1.0C		8.5D
% Mortality		90.0		96.0		66.0

[a] Represents the number of pupae formed from 25 house fly eggs.

[b] Equals the number of eclosed pupae subtracted from 100.

Note: Mean values in the same row without common letters are significantly different ($p < 0.05$).

Table 26. Fly species and diets used for CGA 72662 laboratory studies.

Fly	Diet
M. domestica	350 ml CSMA™
	10 ml fish meal
	200 ml test solution or H_2O
H. illucens	500 ml CSMA™
	300 ml test solution or H_2O
S. robusta	100 ml CSMA™
	200 ml fish meal
	220 ml test solution or H_2O
P. regina and	150 ml CSMA™
F. canicularis	100 ml fish meal
	160 ml test solution or H_2O
O. aenescens	400 ml CSMA™
	100 ml horn fly dry mix[a]
	275 ml test solution or H_2O

[a] 960g wheat flour, 720g fish meal, 120g Na_2CO_3, 400g alfalfa meal (Greer, 1975).

Results. Results are summarized in Table 27 and the raw data are in Appendix 2. At the concentrations tested, CGA 72662 produced 100% larval mortality in all flies except *H. illucens* and *F. canicularis*, and 100% pupal mortality in *H. illucens*. Flies other than *H. illucens* and *F. canicularis* produced no pupae. No larval remains could be found in the growth media which indicates early larval death.

Concentrations of CGA 72662 were within the range of concentrations to be used in the field. Under field conditions, concentrations may become diluted after being applied to fresh poultry manure. Therefore, the concentrations of CGA 72662 used in this experiment were much higher than field populations of flies would be likely to encounter.

Larval house fly dosage-mortality curve for CGA 72662. The test concentrations of CGA 72662 added to house fly larval diets in this experiment were 1.0, 0.75, 0.50, 0.25, and 0.10 ppm. The diet used is shown in Table 26.

Results. The resulting mortality data are shown in Table 28. Two trials were performed and data were combined. In one trial, the number of larviform pupae found at different concentrations of CGA 72662 was recorded (Table 28). The lowest concentration producing larviform pupae was 0.25 ppm. All larviform pupae formed at this concentration eclosed. Those produced at higher concentrations did not eclose. At the highest concentration of CGA 72662, 1.0 ppm, only five uneclosed larviform pupae formed in the four treatment replications. It was therefore assumed that other larvae at this and higher concentrations died prior to reaching the pupal stage.

A larviform pupa formed during the experiment is shown in Figure 28. Larviform pupae were comparable in size to third-instar larvae and slightly darker in color than normally-formed pupae.

Table 27. Summary of per cent larval mortality in CGA 72662 laboratory studies.

		CGA 72662 Concentrations(%)						
Fly Species	Control	0.10	0.075	0.06	0.05	0.04	0.03	0.025
M. domestica	27.5	---	---	100.0	100.0	100.0	100.0	---
H. illucens[a]	0.0	100.0	87.5	---	85.0	---	---	52.5
S. robusta	7.5	100.0	100.0	---	100.0	---	---	100.0
O. aenescens	20.0	100.0	100.0	---	100.0	---	---	100.0
F. canicularis[a]	25.0	77.5	57.5	---	75.0	---	---	52.5
P. regina	5.0	100.0	100.0	---	100.0	---	---	100.0

[a] Values for *H. illucens* and *F. canicularis* represent percentages of pupae that formed. Pupal mortality was 100% in all cases except controls. Other fly species died in the larval stages.

Table 28. Larval mortality and larviform pupae formation resulting from various levels of CGA 72662 in growth media of house flies.

	Treatment (ppm)					
	1	2	3	4	5	6
	1.0	0.75	0.50	0.25	0.10	Control
	15/15[a]	14/15 (11)[b]	13/15 (10)[b]	3/15 (2)[b]	0/15	2/15
	15/15 (5)[b]	15/15 (4)	13/15 (13)	4/15 (3)	5/15	3/15
	15/15	15/15 (13)	13/15 (7)	2/15 (1)	2/15	1/15
	15/15	15/15 (9)	11/15 (8)	3/19 (3)	1/15	2/15
	14/15	8/15	7/15	4/15	3/15	0/15
	12/15	8/15	6/15	3/16	1/15	0/15
	13/15	11/15	5/15	2/16	2/15	0/15
	13/15	9/15	7/15	4/15	1/15	1/16
Σ	112/120	95/120	75/120	25/126	15/120	9/121
% Mortality	93.33	79.17	62.50	19.84	12.50	7.44
Corrected % Mortality[c]	92.80	77.49	59.49	13.40	5.47	

[a] Represents: $\frac{\text{initial no. of 1st instar larvae - no. of eclosed pupae or adults}}{\text{initial no. of 1st instar larvae}}$

[b] Represents number of larviform pupae found.

[c] Corrected using Abbott's formula: $\frac{\text{\% mortality in treatment - \% mortality check}}{\text{100 - \% mortality check}} \times 100$

Figure 28. A larviform pupa formed in medium containing between 0.5 and 1.0 ppm of CGA 72662.

The probability, log dose, upper and lower fiducial limits, and probits are shown in Table 29. The probit curve with the LC_{50} of 0.45 ppm is shown in Figure 29. The slope of the probit curve indicates relative heterogeneity in the susceptibility of the laboratory house fly population to CGA 72662 (Hoskins and Gordon, 1956).

Comparison of the efficacies of CGA 72662 and commercially used organophosphorus larvicides. CGA 72662 and three commercially used organophosphorus larvicides were applied to manure in one house at the tilling site. Sectioning of the manure collection area, assignment of treatments, and the application rates of the compounds are shown in Table 30. Treatment blocks measured 7.6 m along the walk by 1.2 m wide. The procedures for mixing the test concentrations are given in Table 31. Time interval for the collection of samples was as follows:

Sample No.	Time Interval
1	pretreatment, but on treatment day
2	4 days post-treatment
3	7 days post-treatment
4	11 days post-treatment
5	14 days post-treatment
6	28 days post-treatment
7	35 days post-treatment

Sampling ceased after the larval population mean of a treatment block exceeded that of the control block. All sampling ceased 35 days post-treatment because manure in the control block became too dry to support fly populations.

Results. The larval population means for all treatments during each sampling period are shown in Table 32 and Figure 30. The active period for the larvicides is shown in Table 33.

Table 29. The probabilities, log doses, upper and lower fiducial limits, and probits from the probit analysis of CGA 72662 dosage-mortality data.

OBS	PROB	LDOSE	LOWER	UPPER	PROBIT
1	0.01	-11.37	-11.77	-11.10	2.67
2	0.02	-11.21	-11.57	-10.98	2.95
3	0.03	-11.11	-11.45	-10.88	3.12
4	0.04	-11.03	-11.35	-10.81	3.25
5	0.05	-10.97	-11.28	-10.75	3.36
6	0.06	-10.92	-11.21	-10.72	3.45
7	0.07	-10.87	-11.16	-10.68	3.52
8	0.08	-10.83	-11.10	-10.64	3.59
9	0.09	-10.79	-11.06	-10.61	3.66
10	0.10	-10.76	-11.02	-10.58	3.72
11	0.15	-10.61	-10.84	-10.46	3.96
12	0.20	-10.50	-10.70	-10.36	4.16
13	0.25	-10.40	-10.58	-10.27	4.33
14	0.30	-10.31	-10.47	-10.20	4.48
15	0.35	-10.23	-10.38	-10.13	4.61
16	0.40	-10.16	-10.29	-10.06	4.75
17	0.45	-10.08	-10.20	-9.99	4.87
18	0.50	-10.01	-10.11	-9.92	5.00
19	0.55	-9.93	-10.03	-9.85	5.13
20	0.60	-9.86	-9.95	-9.78	5.25
21	0.65	-9.78	-9.87	-9.70	5.39
22	0.70	-9.70	-9.78	-9.61	5.52
23	0.75	-9.61	-9.70	-9.51	5.67
24	0.80	-9.51	-9.60	-9.40	5.84
25	0.85	-9.40	-9.50	-9.27	6.04
26	0.90	-9.26	-9.37	-9.10	6.28
27	0.91	-9.22	-9.34	-9.06	6.34
28	0.92	-9.18	-9.31	-9.01	6.41
29	0.93	-9.14	-9.27	-8.96	6.48
30	0.94	-9.10	-9.23	-8.90	6.55
31	0.95	-9.04	-9.19	-8.84	6.64
32	0.96	-8.98	-9.13	-8.76	6.75
33	0.97	-8.91	-9.07	-8.67	6.88
34	0.98	-8.80	-8.98	-8.55	7.05
35	0.99	-8.64	-8.85	-8.35	7.33

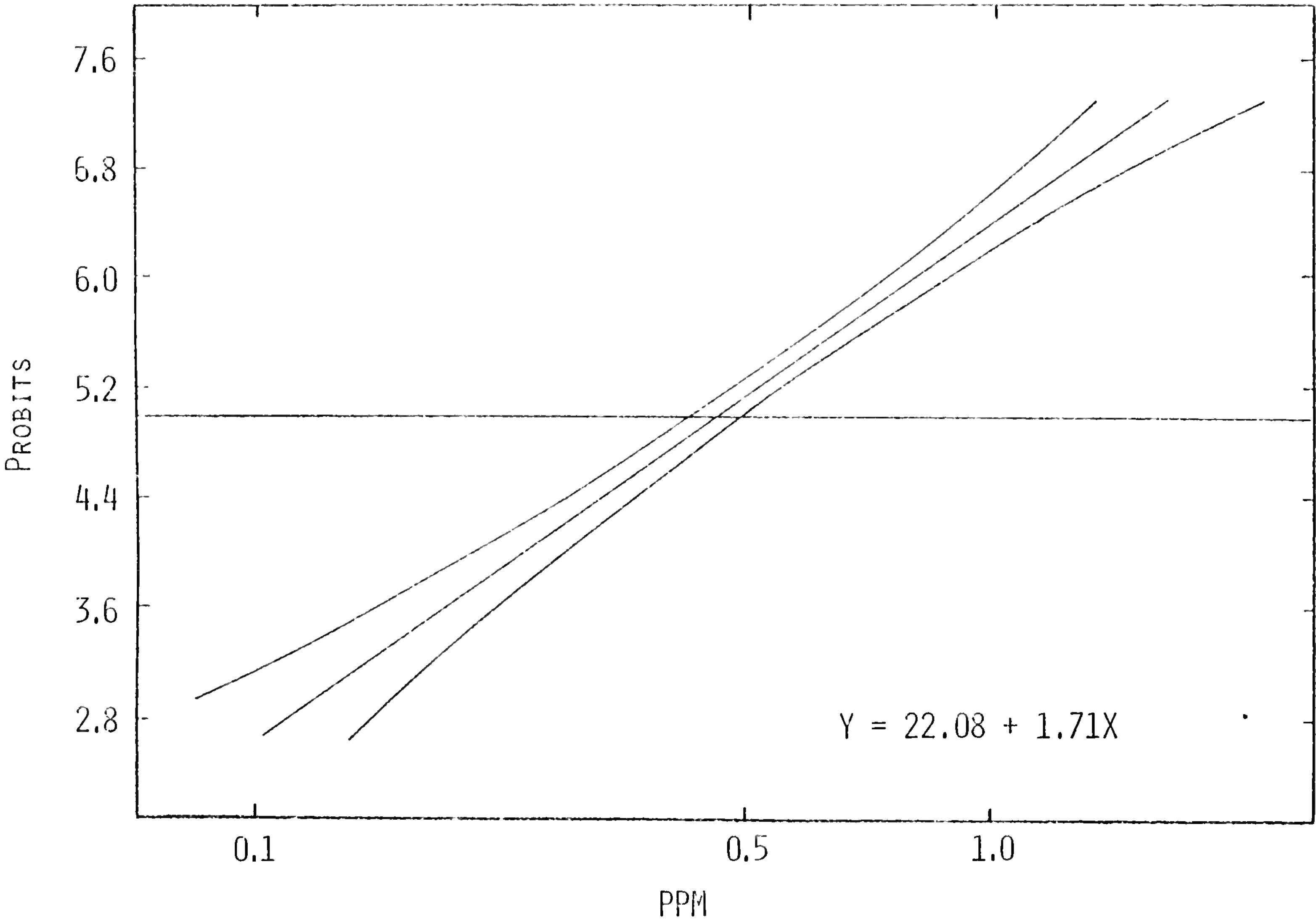
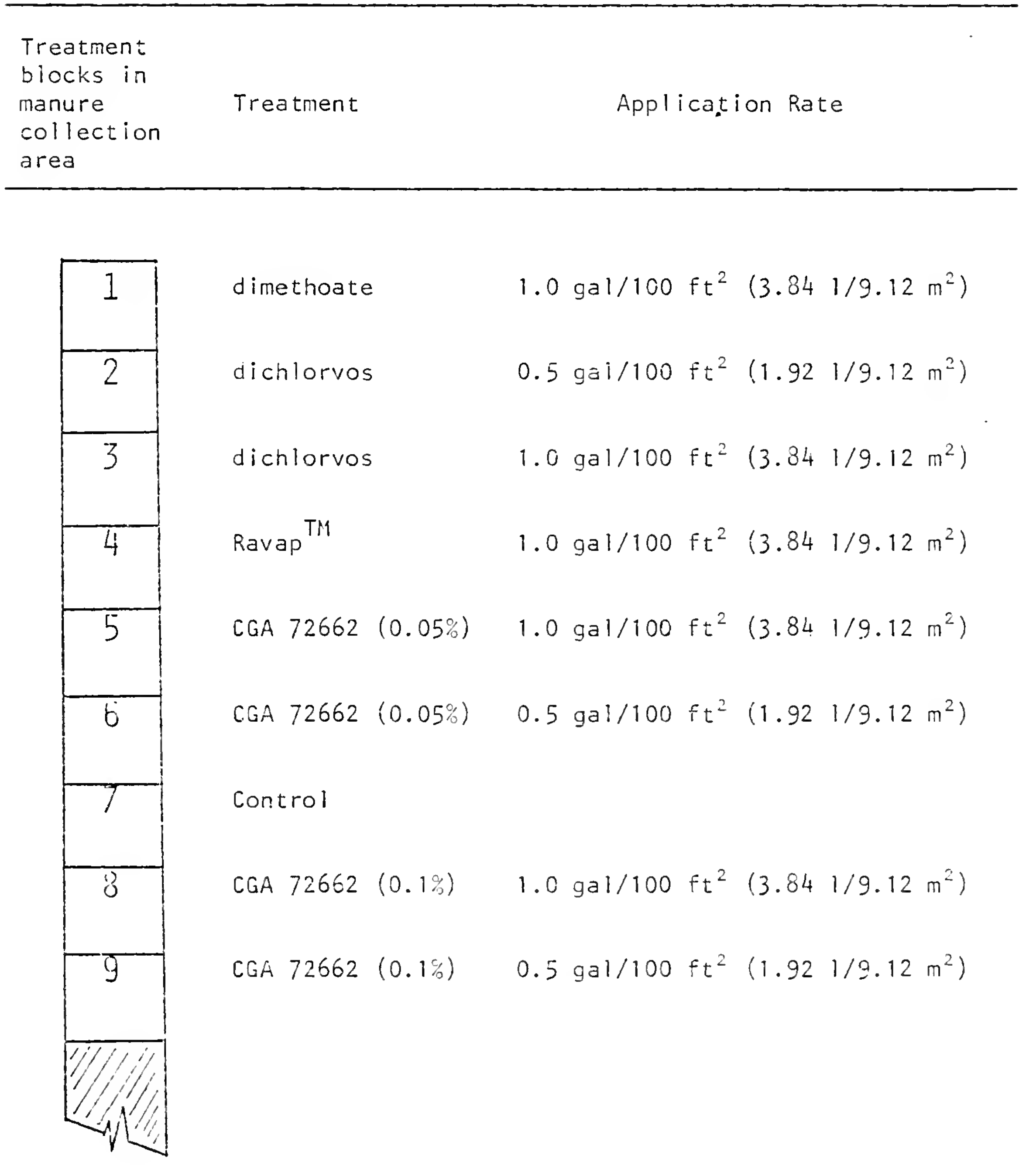


Figure 29. Probit curve, fiducial limits, LC_{50}, and regression equation for CGA 72662 dosage-mortality data.

Table 30. Sectioning of the manure collection area, assignment of treatments, and the application rates of CGA 72662 and the organophosphorus larvicides applied to poultry manure.



Treatment blocks in manure collection area	Treatment	Application Rate
1	dimethoate	1.0 gal/100 ft^2 (3.84 l/9.12 m^2)
2	dichlorvos	0.5 gal/100 ft^2 (1.92 l/9.12 m^2)
3	dichlorvos	1.0 gal/100 ft^2 (3.84 l/9.12 m^2)
4	Ravap^{TM}	1.0 gal/100 ft^2 (3.84 l/9.12 m^2)
5	CGA 72662 (0.05%)	1.0 gal/100 ft^2 (3.84 l/9.12 m^2)
6	CGA 72662 (0.05%)	0.5 gal/100 ft^2 (1.92 l/9.12 m^2)
7	Control	
8	CGA 72662 (0.1%)	1.0 gal/100 ft^2 (3.84 l/9.12 m^2)
9	CGA 72662 (0.1%)	0.5 gal/100 ft^2 (1.92 l/9.12 m^2)

Table 31. Mixing the test concentrations of CGA 72662 and the organophosphorus larvicides applied to poultry manure.

Compound Name	Formulation (% EC)	Contents of Test Concentrations: Compound (ml)	Contents of Test Concentrations: H_2O (ml)	Test Concentration (%)
dimethoate	23.4	192.0	3840	1.0
dichlorvos	23.4	76.8	3840	0.5
Ravap™	23 + 5.7	153.6	3840	1.0
CGA 72662	10	20.0	3840	0.05
CGA 72662	10	40.0	3840	0.1

Table 32. Larval population means for all treatments during each sampling period when poultry manure was treated with CGA 72662 and three organophosphorus larvicides.

		Treatments			
		dimethoate	dichlorvos		Ravap™
			1.92 1/9.12m^2	3.84 1/9.12m^2	
Sampling Period	Time Interval (days)	1	2	3	4
1	0	355.25A	235.00C	386.00A	305.00B
2	4	53.00A	13.00D	19.00C	20.00C
3	7	335.25B	421.00A	35.25B	269.00C
4	11	465.50A	302.50B	527.25A	33.50C
5	14				
6	28				
7	35				

Note: For each sampling period, treatment means without common letters are significantly different ($p < .05$) using the method of J. W. Tukey.

Table 32. Extended.

Treatments				
CGA 72662 (0.05%)		Control	CGA 72662 (0.1%)	
3.84 1/9.12m^2	1.92 1/9.12m^2		3.84 1/9.12m^2	1.92 1/9.12m^2
5	6	7	8	9
266.00C	261.50C	104.75D	117.25D	119.00D
10.00D	27.00B	20.50C	0.50E	0.25E
0.00F	0.00F	108.25D	0.00F	0.00F
0.00C	3.75C	62.25C	0.00C	0.00C
0.00B	62.25A	73.75A	0.00B	0.00B
6.25B	54.75A	55.75A	0.00B	0.00B
0.00B	103.50A	107.75A	0.00B	0.00B

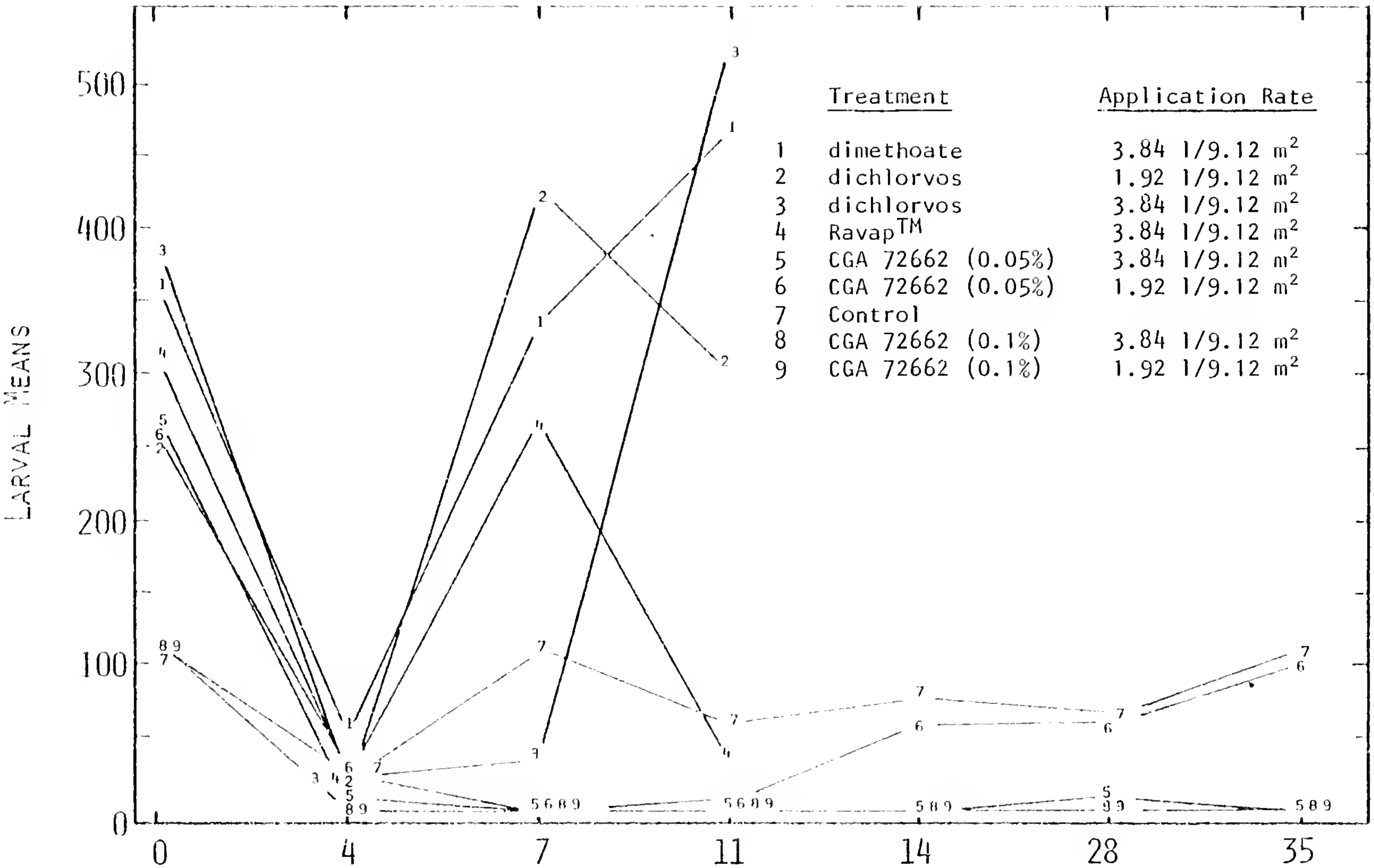


Figure 30. Larval population means for all treatments during each sampling period when poultry manure was treated with CGA 72662 and three organophosphorus larvicides.

Table 33. Larvicidal activity period of compounds tested in the CGA 72662 organophosphorus larvicide study.

Sampling Period	Time Interval (Days)	dimethoate	dichlorvos 1.92 l/9.12m^2	dichlorvos 3.84 l/9.12m^2	Ravap™	CGA 72662 (0.05%) 3.84 l/9.12m^2	CGA 72662 (0.05%) 1.92 l/9.12m^2	Control	CGA 72662 (0.1%) 3.84 l/9.12m^2	CGA 72662 (0.1%) 1.92 l/9.12m^2
		1	2	3	4	5	6	7	8	9
1	0									
2	4									
3	7									
4	11									
5	14									
6	28									
7	35									

At the beginning of the experiment, there were significant differences between larval population means in the treatment blocks, but the control block had the lowest mean of all blocks (Figure 30).

By the second sampling period, only blocks treated with dimethoate and CGA 72662 (0.05%) at 1.92 l/9.12 m^2 had larval population means that were significantly greater than that of the control block (Table 32, Figure 30). There were no significant differences between the larval population means of blocks treated with dichlorvos at 3.85 l/ 9.12 m^2 and Ravap, and the larval population mean of the control block. Larval means of all other blocks were significantly lower than the control block.

By the end of the third sampling period, fly resurgence had begun in the blocks treated with dimethoate, dichlorvos at 1.92 l/9.12 m^2, and Ravap, and the larval population means were all significantly greater than the larval population mean of the control group (Figure 30). Larval means of all other treatment blocks were significantly lower than the control block population. Larval populations in the blocks treated with both rates of CGA 72662 were reduced to zero.

At the end of the fourth sampling period, populations in blocks treated with dimethoate and dichlorvos at 1.92 l/9.12 m^2 continued to resurge, and resurgence also began in the block treated with dichlorvos at 3.84 l/9.12 m^2 (Figure 30). Sampling was discontinued in these blocks. The block treated with Ravap showed a decrease in larval population, but by the end of the fifth sampling period, the population was considered too numerous to count. Therefore, the larval population counted during the fourth sampling period was the last recorded count and sampling in the Ravap-treated block also ceased.

For the remainder of the experiment, there were no significant differences between the larval populations in the block treated with the lower level of CGA 72662 (0.05%) at 1.92 1/9.12 m^2 and the control block (Table 32, Figure 30). Populations in other blocks still being monitored were reduced to zero.

The activity period for dimethoate, dichlorvos at 1.92 1/9.12 m^2, and Ravap was 7 days (Table 33). The activity periods for dichlorvos at 3.84 1/9.12 m^2 and CGA 72662 (0.05%) at 1.92 1/9.12 m^2 were 11 and 14 days respectively. CGA 72662 (0.05%) at 3.84 1/9.12 m^2 and both rates of 0.1% CGA 72662 were still active 35 days after treatment was applied.

Larvicidal activity of CGA 72662 when tilled into wet manure. Manure from poultry houses at the tilling site was cleaned out leaving the bottoms of the manure collection areas ca. 14 cm below the surface of the walks. At this depth, the tines of the tiller were too short to reach the fresh manure being dropped. This, in conjunction with heavy rains and flooding, resulted in the rapid development of large populations of *H. illucens*.

To give consistency to the manure and to raise it to a level that the tiller tines could reach, wood shavings were added to the manure collection areas as previously described. The manure was tilled once, leaving the surface of the manure-wood shavings mixture ca. 29 cm above the surface of the walks.

Afterward, manure was not tilled for 1 week. During this time, large populations of house flies developed near the surface of the manure-wood shavings mixture, along with the soldier fly populations further beneath the surface (Figure 31).

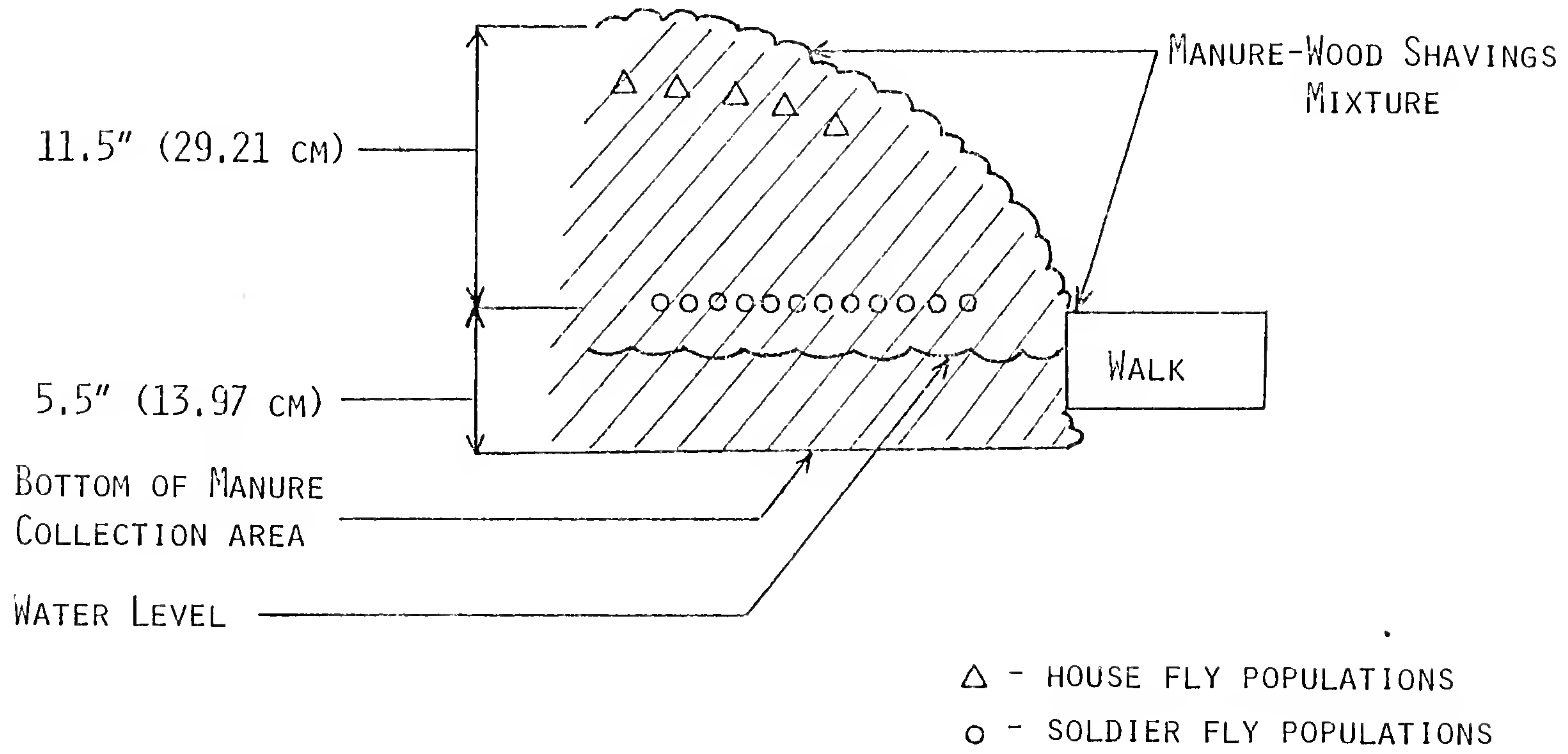


Figure 31. Cross-section of manure-wood shavings mixture 1 week after tilling, showing relative locations of house fly and soldier fly populations.

A section of the manure collection area of one house was divided into four blocks and treatments were assigned as shown in Figure 32. A 0.1% solution of CGA 72662 was mixed as shown below:

40 ml of CGA 72662 + 3.84 l H_2O = 0.1% solution

The application rate was 3.84 l/9.12 m^2. Treatment blocks 2 and 3 were tilled twice weekly and blocks 0 and 1 were left untilled.

Pretreatment samples were collected on the day the experiment began and additional samples were collected at weekly intervals. Larvae of house flies, soldier flies, and little house flies were counted as in the previous experiment. Sampling was discontinued after 5 weeks because of climatic conditions.

Results. Larval means from each treatment block for each sampling period are shown in Table 34.

The pretreatment house fly samples revealed significant differences between treatment-block population means. Blocks to be treated with CGA 72662 had numerically lower populations than the remaining two blocks.

One week after treatment, there were no significant differences between the house fly population means in the four blocks, but in the untilled block sprayed with CGA 72662, the house fly population mean was 2.75 as compared to 37.50 in the tilled CGA 72662 block (Table 34). Manure was so wet that house fly larvae were pupating just below the surface of the manure instead of at the edges of the manure pack. In the untilled GCA 72662 block, fly larvae coming up to pupate were being exposed to the concentrated CGA 72662 on the manure surface. In the tilled CGA 72662 block, CGA 72662 may have been diluted by the tilling and was slower acting.

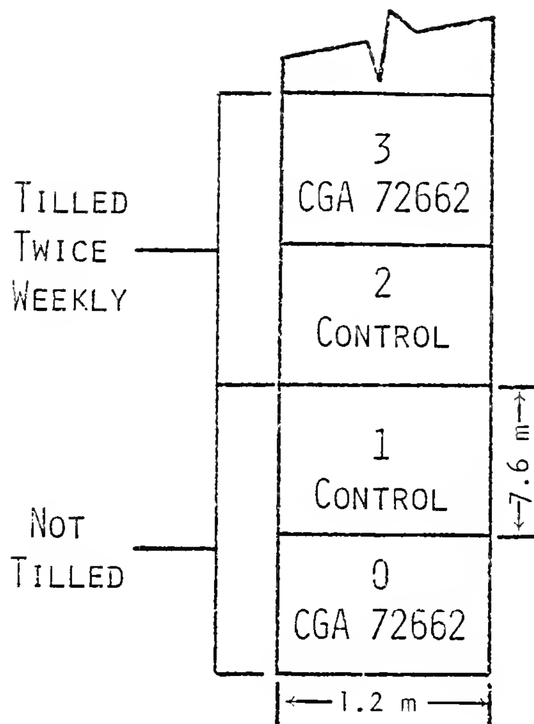


Figure 32. Treatment area, assignment of treatments, and tilling schedule in the CGA 72662 tilling trial.

Table 34. Weekly treatment means of house fly, soldier fly, and little house fly populations from manure treated with CGA 72662 and tilled twice weekly.

Week	Treatments[a] 0	1	2	3
	Musca domestica			
0 = Precount	97.00B	187.50A	136.00AB	143.00AB
1	2.75A	39.50A	17.50A	37.50A
2	0.00C	47.50A	22.75B	0.00C
3	0.00B	16.25A	16.25A	0.00B
4	93.50A	31.25B	35.25B	0.00C
5	209.00B	376.25A	150.25C	0.00C
	Hermetia illucens			
0 = Precount	31.25A	1.75B	2.25B	0.50B
1	18.50B	7.50BC	12.00AB	1.00C
2	0.00B	0.75B	7.75A	0.00B
3	0.00B	8.50A	6.00AB	0.00B
4	0.00B	15.25A	9.25A	0.00B
5	0.00B	39.00A	12.75AB	0.00B
	Fannia canicularis			
0 = Precount	10.00A	28.50A	27.50A	11.00A
1	0.00A	2.25A	1.50A	2.00A
2	0.00B	3.25AB	7.00A	0.00B
3	0.00A	0.25A	0.00A	0.00A
4	0.50A	0.00A	0.00A	0.00A
5	0.00A	0.00A	0.00A	0.00A

[a] Treatment 0 = CGA 72662, no tilling; Treatment 1 = control, no tilling; Treatment 2 = control, tilled twice weekly: Treatment 3 = CGA 72662, tilled twice weekly.

Note: Means in the same row having the same letter are not significantly different ($p < .05$).

By the second week after treatment, house fly populations in both blocks treated with CGA 72662 were reduced to zero. Population means in the tilled control were significantly lower than the population means in the untilled control.

By the fourth week post-treatment, house flies began to resurge in the untilled block treated with CGA 72662 and the larval population mean was significantly higher than the population means in either control block. Larval populations in the tilled CGA 72662 block remained at zero.

At the end of the fifth week, house fly resurgence had occurred in all treatment blocks except the tilled CGA 72662 block (Table 34, Figure 33).

Initial *H. illucens* populations were much lower than the house fly populations (Table 34). By the second week, soldier fly populations were reduced to zero in blocks treated with CGA 72662 and populations in both untreated controls were greater than populations in the CGA 72662 blocks. This situation continued through the end of the experiment (Table 34, Figure 34).

There were no significant differences in the little house fly population means at the beginning of the experiment, but by the second week, populations in both CGA 72662 blocks had been reduced to zero. By the third week there were no significant differences between little house fly population means. This situation remained the same through the end of the experiment (Table 34, Figure 35).

CGA 72662 added to drinking water as an oral larvicide. The test concentrations of CGA 72662 utilized were 5.0, 1.5, 10.0, and 20.0 ppm. The treatment and sample collection schedule is shown in Table 35.

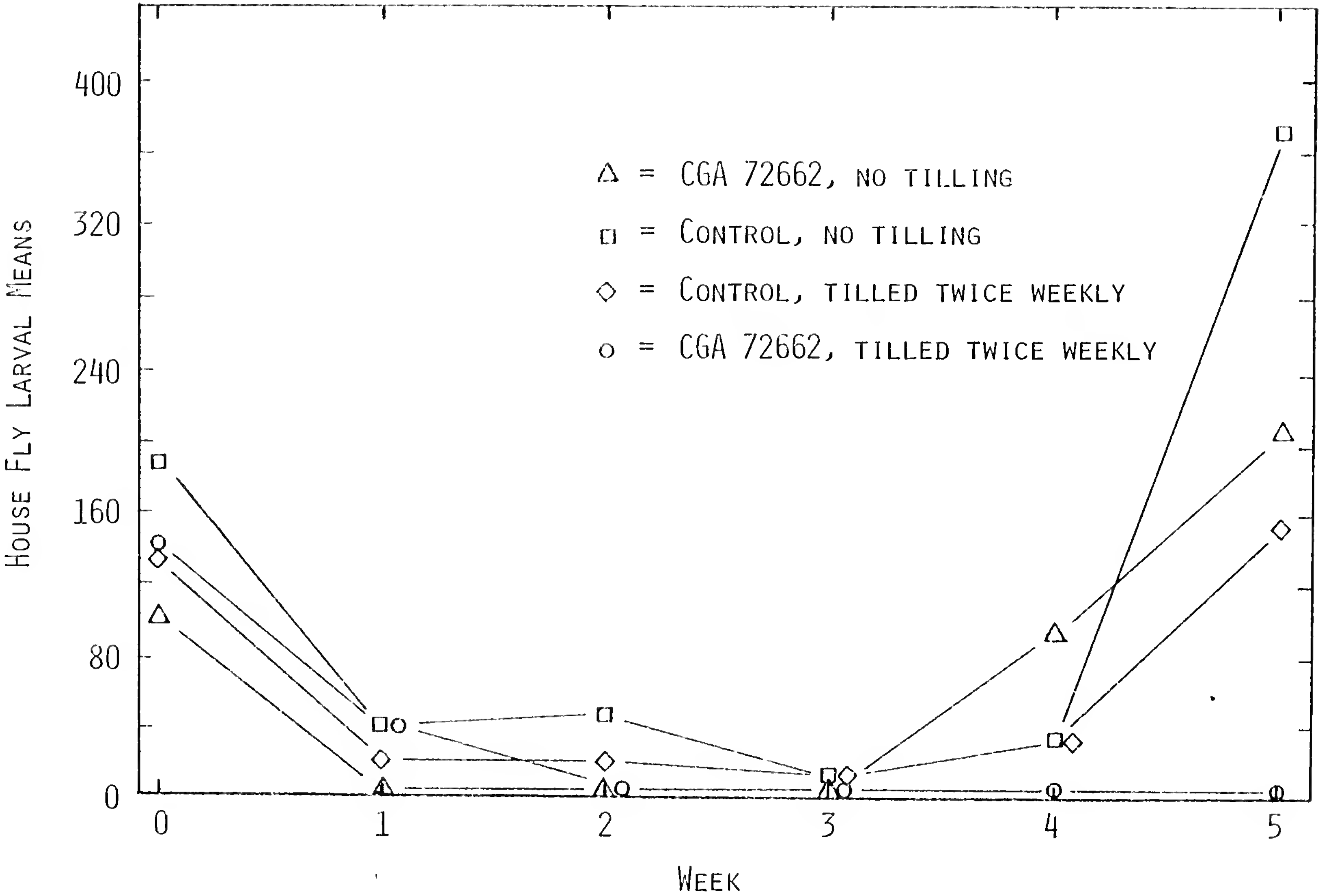


Figure 33. Weekly treatment means of house fly populations from manure treated with CGA 72662 and tilled twice weekly.

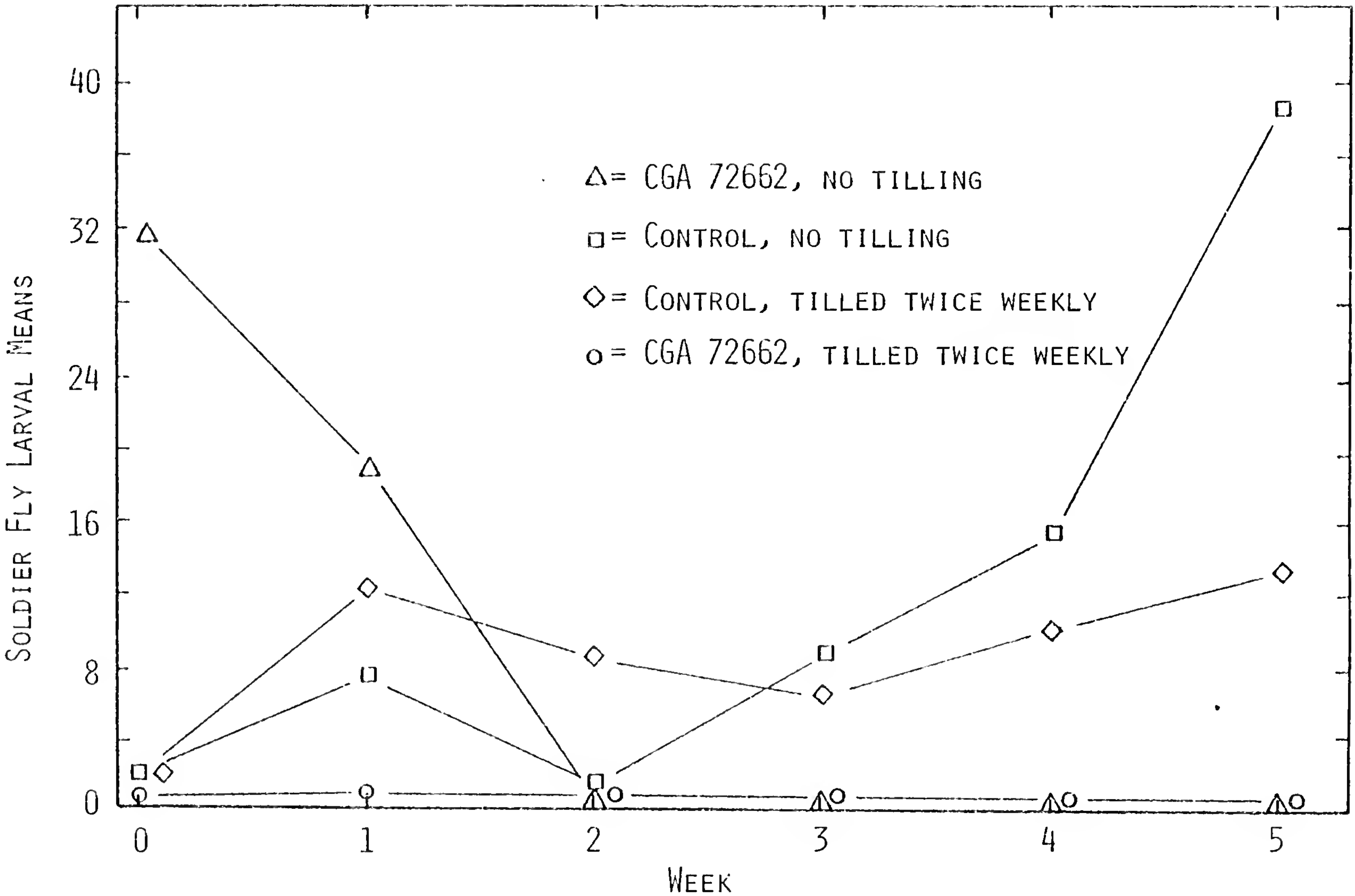


Figure 34. Weekly treatment means of soldier fly populations from manure treated with CGA 72662 and tilled twice weekly.

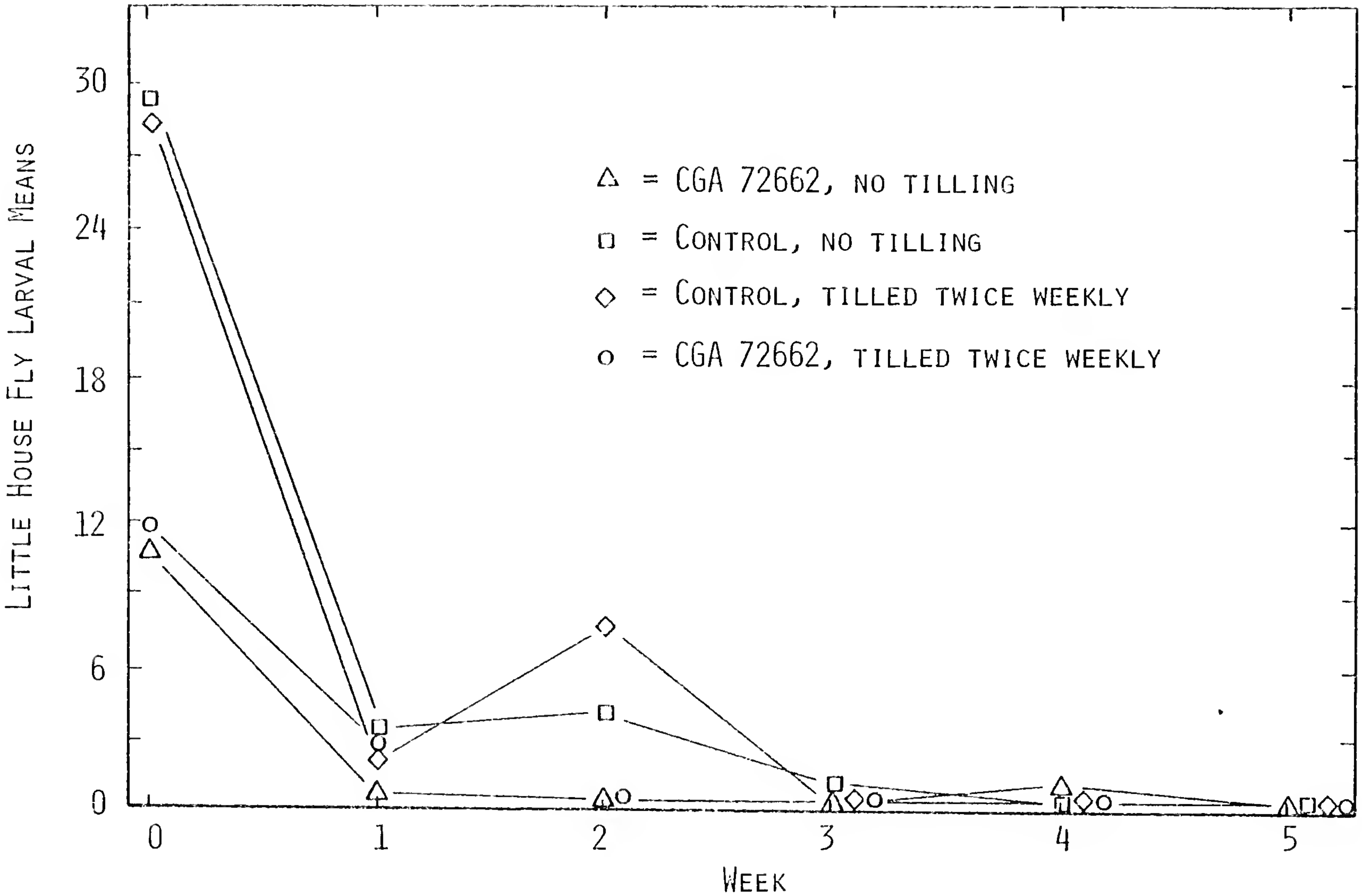


Figure 35. Weekly treatment means of little house fly populations from manure treated with CGA 72662 and tilled twice weekly.

Table 35. Treatment and sample collection schedule when CGA 72662 was added to the drinking water of laying hens as an oral larvicide.

Day 1	Began daily treatment with CGA 72662 at 1.5 and 5.0 ppm.
Day 7	First sample group of 5 cups of manure per treatment was collected.
Day 11	Second sample group of 10 cups of manure per treatment was collected.
Day 16	Discontinued daily treatment with CGA 72662 at 1.5 and 5.0 ppm. All birds were returned to 500 ml H_2O 3 times daily.
Day 22	Began daily treatment with CGA 72662 at 10 and 20 ppm. Replaced polyethylene under cages and in water troughs.
Day 29	First sample group of 5 cups of manure per treatment was collected.
Day 30	Discontinued daily treatment with CGA 72662 at 10 and 20 ppm. All birds were returned to 500 ml H_2O 3 times daily. Replaced polyethylene under cages.
Day 33	One sample group of 5 cups of manure per treatment was collected 3 days post-treatment.
Day 35	One sample group of 5 cups of manure per treatment was collected 5 days post-treatment.

Results. Both levels of CGA 72662 significantly increased house fly mortality in the first trial (Table 36). The mortalities of the 5.0-and 1.5-ppm treatments were 87.56 and 80.89% respectively, and corrected mortalities were 84.85 and 76.72% respectively.

The results of the second trial are shown in Table 37. Manure from both CGA 72662 treatments produced house fly mortalities significantly greater than the control mortality. Treatment levels of 20.0 and 10.0 ppm produced mortalities of 100.00 and 92.00% respectively, and corrected mortalities of 100.00 and 91.11% respectively.

Mortalities produced by manure from treated hens 3 days post-treatment were still significantly greater than mortality rates in control groups (Table 38). Manure from hens formerly receiving CGA 72662 at levels of 20.0 and 10.0 ppm produced mortalities of 99.33 and 80.00% respectively. Corrected mortalities were 98.97 and 69.39% respectively.

Results of samples bioassayed 5 days post-treatment are shown in Table 39. There were no significant differences in mortality due to treatment and, after correction, mortalities produced by manure from both former treatment groups were less than 10%.

Blacklight Electrocutor Grid Traps for Adult Fly Surveys

Fly catches from experimental blacklight electrocutor grid traps (Danalco™) were processed for 6 months. Species of flies observed includes *M. domestica*, *H. illucens*, *S. calcitrans*, *H. irritans*, and *Ophyra* sp.

Results. The monthly catches of the above species of flies in the two traps are shown in Table 40. In nearly all cases, trap A caught greater numbers of flies than trap B. Except for April and September,

Table 36. Mortality of immature house flies in the manure of laying hens collected when CGA 72662 was added to the drinking water at the rates of 10 and 20 ppm.

	20 ppm	10 ppm	Control
	15/15	14/15	2/15
	15/15	13/15	1/15
	15/15	14/15	3/20
	15/15	14/15	1/15
	15/15	14/15	1/15
Total Mortality	75/75	69/75	8/80
Mean Mortality	15.00	13.80	1.60[b]
% Mortality	100.00	92.00	10.00
Corrected % Mortality[a]	100.00	91.11	---

[a] Corrected using Abbott's formula:

$$\frac{\%\ \text{treatment mortality} - \%\ \text{check mortality}}{100 - \%\ \text{check mortality}} \times 100$$

[b] Significantly lower than other two treatments at $p < 0.05\%$.

Table 37. Mortality of immature house flies in the manure of laying hens collected when CGA 72662 was added to the drinking water at the rates of 1.5 and 5.0 ppm.

	5.0 ppm	1.5 ppm	Control
	13/15	6/15	4/15
	12/15	8/15	3/15
	10/15	11/15	3/16
	11/15	7/15	4/16
	11/15	8/15	0/17
	14/15	14/15	0/15
	13/15	13/15	7/15
	13/15	14/15	2/15
	14/15	15/15	1/15
	15/15	15/15	3/15
	14/15	15/15	2/15
	13/15	13/15	4/15
	15/15	15/15	2/15
	14/15	15/15	6/16
	15/15	13/15	0/15
Total Mortality	197/225	182/225	41/229
Mean Mortality	13.13A	12.13A	2.73B
% Mortality	87.56	80.89	17.90
Corrected % Mortality[a]	84.85	76.72	---

[a] Corrected using Abbott's formula:

$$\frac{\%\ \text{treatment mortality} - \%\ \text{check mortality}}{100 - \%\ \text{check mortality}} \times 100$$

Note: Means having different letters are significantly different ($p < 0.05$).

Table 38. Mortality of immature house flies in the manure of laying hens collected 3 days after treatment of drinking water with CGA 72662 at 10 and 20 ppm was terminated.

	20 ppm	10 ppm	Control
	15/15	13/15	6/15
	15/15	10/15	5/15
	15/15	12/15	2/15
	15/15	11/15	5/15
	15/15	13/15	5/15
	15/15	12/15	5/15
	14/15	13/15	12/15
	15/15	13/15	6/15
	15/15	11/15	4/15
	15/15	12/15	2/15
Total Mortality	149/150	120/150	52/150
Mean Mortality	14.90A	12.00B	5.20C
% Mortality	93.33	80.00	34.67
Corrected % Mortality[a]	98.97	69.39	---

[a] Corrected using Abbott's formula:

$$\frac{\%\ \text{treatment mortality} - \%\ \text{check mortality}}{100 - \%\ \text{check mortality}} \times 100$$

Note: Means having different letters are significantly different ($p < 0.05$).

Table 39. Mortality of immature house flies in the manure of laying hens collected 5 days after treatment of drinking water with CGA 72662 at 10 and 20 ppm was terminated.

	20 ppm	10 ppm	Control
	4/15	3/15	0/15
	1/15	2/15	2/15
	2/15	0/15	1/15
	2/15	1/15	0/15
	0/15	1/15	0/15
Total Mortality	9/75	7/75	3/75
Mean Mortality	1.80	1.40	0.60
% Mortality	12.00	9.33	4.00
Corrected % Mortality[a]	8.33	5.55	---

[a] Corrected using Abbott's formula:

$$\frac{\%\ \text{treatment mortality} - \%\ \text{check mortality}}{100 - \%\ \text{check mortality}} \times 100$$

Table 40. Monthly catches of *Musca domestica*, *Hermetia illucens*, *Stomoxys calcitrans*, *Hematobia irritans*, and *Ophyra* sp. in two blacklight electrocutor grid traps.

Trap	Months					
	Apr	May	Jun	Jul	Aug	Sep
			M. domestica			
A	21110	34380	67880	124930	68070	18460
B	11570	21470	14200	48940	26360	6930
			H. illucens			
A	40	140	180	140	90	10
B	50	100	20	100	50	50
			S. calcitrans			
A	950	1250	720	40	10	260
B	640	890	210	60	0	120
			H. irritans			
A	60	30	0	0	0	0
B	40	20	0	10	0	0
			Ophyra sp.			
A	200	770	250	180	20	0
B	220	530	60	60	10	0

trap A caught more soldier flies than trap B. Trap B caught greater numbers of *S. calcitrans* in July than trap A, but trap A caught the most *S. calcitrans* during the other months. There were slight numerical differences in catches of *H. irritans*, but trap A caught more than trap B except during July. Except for the month of April, trap A caught more *Ophyra* sp. than trap B.

Cow pastures bordered the tilling site on two sides. This accounts for the numbers of *S. calcitrans* and *H. irritans* caught in the traps.

Catches of *H. illucens* in April, May, and June probably consisted of adults that emerged from pupae formed the previous fall. Subsequent catches most likely consisted of adults that emerged during April and May.

Trap catches indicate relatively high populations of *Ophyra* during April, May, and June, but adults were never noted at the tilling site except in the traps.

House fly populations were low in April, peaked in July, and dropped again in September (Figure 36). The two traps presented two different views of house fly activity during the 6-month period. This indicates the necessity of using traps that are uniform when using trap data for survey work. The low June catch by trap B cannot be explained.

Efficacies of Granular Fly Baits

Laboratory and field testing were performed on granular baits provided by two different manufacturers. Both groups of tests will be described separately.

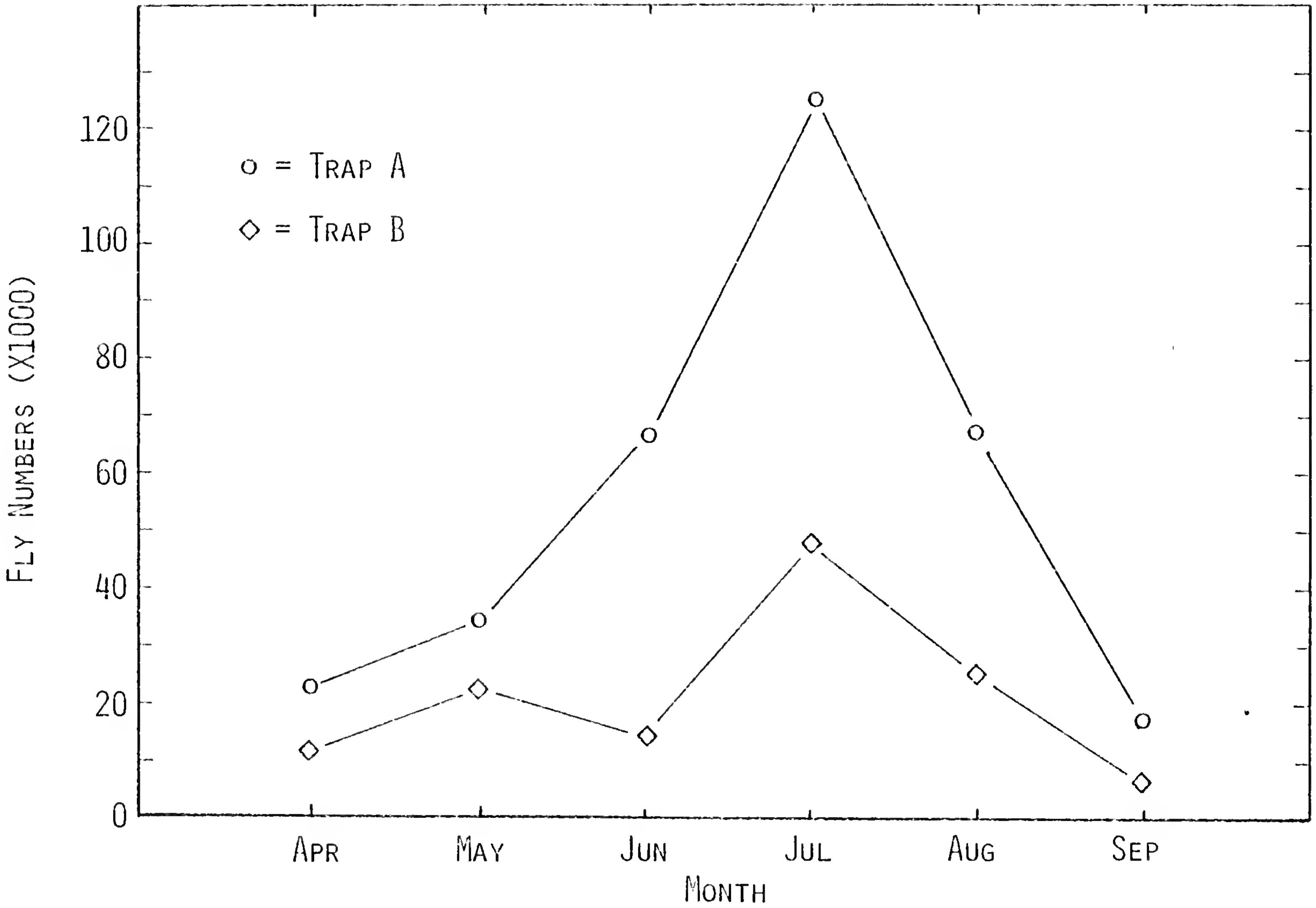


Figure 36. Fluctuation in house fly populations as recorded by two blacklight traps at the tilling site.

<u>Farnam baits.</u> The following baits were submitted by Farnam:

Kill'em Fly Killer II (Bomyl) - sand base
Kill'em Fly Bait (Bomyl) - corn cob base
Kill'em Fly Killer (Vapona + ronnel)
SX-70 Fly Bait (methomyl)

Baits were subjected to knockdown and residual tests. The amount of each bait used was 5 g.

<u>Knockdown test.</u> Results are shown in Table 41. Kill'em Fly Killer had the fastest knockdown, killing all flies in 10 min. Kill'em Fly Bait killed all flies within 1 hr and 40 min, and SX-70 Fly Bait and Kill'em Fly Killer II killed all flies in 4 hr and 40 min, and 4 hr and 50 min respectively.

<u>Residual test.</u> Results are shown in Table 42. By the 10th day of testing, all baits had picked up moisture from the air. SX-70 Fly Bait had the longest residual, still producing a mortality rate of greater than 25% after a 6-week testing period. Testing of Kill'em Fly Killer ceased on day 10 after the mortality rate dropped to 20% and testing of Kill'em Fly Killer II and Kill'em Fly Bait ceased on day 19 after mortality rates dropped to 30 and 10% respectively.

Although testing was discontinued on one bait on day 10 and two baits on day 19, these baits were maintained through the end of the experiment. As a check, flies were exposed to the discontinued baits once every week. Mortality never reached the last recorded values in Table 42 and continued to drop with time.

The sudden fluctuation in mortality rates produced by SX-70 Fly Bait during days 15 through 33 is unexplainable. Equally unexplainable is the 22.5% control mortality on day 12.

Table 41. Results of knockdown test with Farnam baits.

Time Interval	(1) Kill'em Fly Killer II[TM] (Bomyl) (sand base)					(2) Kill'em Fly Bait[TM] (Bomyl) (corn cob base)					(3) Kill'em Fly Killer[TM] (Vapona + ronnel)					(4) SX 70 Fly Bait[TM] (methomyl)				
	Replication				Per cent	Replication				Per cent	Replication				Per cent	Replication				Per cent
	A	B	C	D	Mortality	A	B	C	D	Mortality	A	B	C	D	Mortality	A	B	C	D	Mortality
0[a]	0	0	0	0[b]	0.0[c]	0	0	0	0	0.0	0	0	0	0	0.0	0	0	0	0	0.0
1	1	0	0	0	2.5	5	2	7	4	32.5	10	10	10	10	100.0	0	2	3	1	15.0
2	3	2	4	1	25.0	9	6	10	9	85.0						3	4	6	1	35.0
3	3	5	4	2	35.0	9	8	10	10	92.5						3	4	6	2	37.5
4	3	5	4	3	37.0	9	8	10	10	92.5						6	5	6	8	62.5
5	3	5	7	3	45.0	10	9	10	10	97.5						6	5	6	8	62.5
6	4	5	7	4	50.0	10	9	10	10	97.5						6	5	6	8	62.5
7	5	7	7	5	60.0	10	9	10	10	97.5						7	6	6	8	67.5
8	5	7	7	5	60.0	10	9	10	10	97.5						8	6	6	8	70.0
9	5	7	7	5	60.0	10	9	10	10	97.5						9	6	7	8	77.5
10	5	7	7	6	67.5	10	10	10	10	100.0						8	6	7	8	72.5
11	5	7	8	6	65.0											8	6	7	8	72.5
12	5	7	8	6	65.0											8	6	7	8	72.5
13	5	7	8	6	65.0											8	6	7	9	75.0
14	5	8	8	6	67.5											8	6	7	9	75.0
15	5	8	8	6	67.5											8	6	7	9	75.0
16	5	8	8	6	67.5											8	7	7	9	77.5
17	5	9	9	6	72.5											8	8	9	9	85.0
18	5	9	9	6	72.5											8	8	9	9	85.0
19	5	9	9	6	72.5											8	8	9	9	85.0
20	5	9	10	7	77.5											8	8	9	9	85.0
21	6	9	10	7	80.0											8	8	9	9	85.0
22	6	9	10	8	82.5											8	8	9	9	85.0
23	7	9	10	8	85.0											8	8	9	9	85.0
24	10	9	10	9	95.0											9	9	10	9	92.5
25	10	9	10	9	95.0											9	9	10	9	92.5
26	10	9	10	10	97.5											9	9	10	9	92.5
27	10	9	10	10	97.5											9	9	10	10	95.0
28	10	9	10	10	97.5											9	9	10	10	95.0
29	10	10	10	10	100.0											10	10	10	10	100.0

[a] Each number represents a 10 min interval.

[b] Each number represents the number of flies killed out of a total of 10.

[c] Per cent mortality = $\frac{\text{number dead}}{10} \times 100$

Note: All flies used in this experiment were five day-old female house flies, *Musca domestica* (L.), from a laboratory colony. Flies were sexed while anesthetized with CO_2.

Table 42. Results of residual tests with Farnam baits.

Time Interval	Baits: (1) Kill'em Fly Killer[TM] (Bonyl) (sand base) Replication A B C D	(1) Per cent Mortality	(2) Kill'em Fly Bait[TM] (Bonyl) (corn cob base) Replication A B C D	(2) Per cent Mortality	(3) Kill'em Fly Killer[TM] (Vapona + ronnel) Replication A B C D	(3) Per cent Mortality	(4) SX 70 Fly Bait[TM] (methomyl) Replication A B C D	(4) Per cent Mortality	Control with sugar water Replication A B C D	Control with sugar water Per cent Mortality	Control Replication A B C D	Control Per cent Mortality
1[a]	10 10 10 10[b]	100.0[c]	10 10 10 10	100.0	10 10 10 10	100.0	10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
2	10 10 10 10	100.0	10 10 10 10	100.0	10 10 10 10	100.0	10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
3	10 9 10 9	95.0	10 10 10 10	100.0	10 10 10 10	100.0	10 9 10 10	97.5	0 0 0 0	0.0	0 0 0 0	0.0
4	10 10 10 7	92.5	10 9 10 7	90.0	10 9 10 10	97.5	10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
5	9 10 9 9	92.5	9 9 9 9	90.0	8 5 8 7	70.0	10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
10	9 10 10 10	97.5	9 9 9 9	90.0	2 0 2 4	20.0	10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
11	10 10 8 9	92.5	10 10 10 10	100.0			10 10 10 10	100.0	0 0 0 0	0.0	1 0 2 0	7.5
12	10 7 10 9	90.0	8 10 10 10	95.0			10 10 10 10	100.0	1 0 0 0	2.5	3 3 3 0	22.5
15	8 6 - -	70.0	8 7 - -	75.0			8 9 - -	85.0	0 0 - -	0.0	0 0 - -	0.0
16	9 10 10 9	95.0	8 8 5 9	75.0			9 9 9 9	90.0	0 0 0 0	0.0	0 0 1 2	7.5
18	8 8 9 9	85.0	8 9 8 9	85.0			9 9 10 9	92.5	0 0 0 0	0.0	0 0 0 0	0.0
19	3 3 3 3	30.0	1 0 3 0	10.0			10 9 8 9	94.7	0 0 0 0	0.0	0 0 0 0	0.0
22							10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
23							10 10 10 10	100.0	0 0 0 0	0.0	0 0 0 0	0.0
24							8 6 3 10	81.8	0 0 0 0	0.0	0 0 0 0	0.0
25							10 7 7 9	91.7	0 0 0 1	2.5	0 0 0 2	5.0
26							7 9 5 6	71.7	3 4 4 4	37.5	0 2 0 0	5.0
29							7 6 8 6	93.1	0 0 0 0	0.0	0 0 0 0	0.0
31							1 6 4 2	41.3	0 0 0 0	0.0	0 0 0 0	0.0
32							6 7 1 10	92.3	0 0 0 0	0.0	0 0 0 0	0.0
33							7 9 8 9	94.3	0 0 0 0	0.0	0 1 0 0	2.5
36							7 9 9 8	91.7	0 0 0 0	0.0	0 0 0 0	0.0
37							9 5 8 9	86.1	0 0 0 0	0.0	0 0 0 0	0.0
38							7 6 7 8	73.7	0 0 0 0	0.0	0 0 0 0	0.0
39							2 4 2 3	28.9	0 0 0 0	0.0	0 0 0 0	0.0
40							4 4 5 4	46.0	0 0 0 0	0.0	0 0 0 0	0.0
43							5 3 2 2	31.0	0 0 0 0	0.0	0 0 0 0	0.0

[a] Each number represents a one-day time interval. Skipped numbers indicate weekends or holidays.

[b] Each number represents the number of flies killed out of a total of 10.

[c] Per cent mortality = $\frac{\text{number dead}}{10} \times 100$

Note: All flies used in this experiment were five-day old female house flies, *Musca domestica* (L.), from a laboratory colony. Flies were sexed while anesthetized with CO_2.

Field test. The baits described above plus Golden Malrin without Muscamone were tested in the field with and without the addition of Lure'em II attractant supplied by Farnam. Golden Malrin with Muscamone was used as a standard. When baits were tested alone, 5 g were used. When tested with the attractant, 5 g of bait and 3 g of the attractant were used together. The Lure'em II attractant was never used in the same bait station with Golden Malrin with Muscamone.

Results. Treatment means by treatment are shown in Table 43 and Figure 37. Treatment means by sex are shown in Table 44 and Figure 38. Raw data is in Appendix 3. Kill'em Fly Killer II plus Lure'em II attractant killed a significantly greater number of flies than the other treatments including the Golden Malrin with Muscamone standard (Table 43, Figure 37). Treatment means for Kill'em Fly Killer II plus Lure'em II attractant and Kill'em Fly Bait plus Lure'em II attractant were 158.25 and 116.13 flies per bait station respectively. All other means were less than 30.0 flies per bait station (Table 43).

Lure'em II attractant improved the kills of all baits except SX-70 Fly Bait. Golden Malrin without Muscamone plus Lure'em II attractant killed a significantly greater number of flies than the Golden Malrin with Muscamone standard. Kills produced by the standard bait were numerically lower than all other kills except those of Kill'em Fly Killer (Table 43).

Kill'em Fly Killer killed males and females of *M. domestica* in equal numbers (Table 44, Figure 38), but all other baits killed more females than males. Three baits killed significantly greater numbers of females than males. Lure'em II attractant increased the catches of females for all baits except SX-70 Fly Bait. Golden Malrin without

Table 43. Treatment means by treatment in Farnam bait field trial.

Treatment No.	Treatment	Treatment Mean
1 + 5	Kill'em Fly Killer II™ + Lure'em II™	158.25 A
2 + 5	Kill'em Fly Bait™ + Lure'em II	116.13 B
4	SX-70 Fly Bait™	28.63 C
4 + 5	SX-70 Fly Bait + Lure'em II	20.13 CD
6 + 5	Golden Malrin™ w/o Muscamone™ + Lure'em II	20.00 CDE
3 + 5	Kill'em Fly Killer™ + Lure'em II	19.63 DEF
6	Golden Malrin w/o Muscamone	15.75 DEFG
2	Kill'em Fly Bait	11.38 FGH
1	Kill'em Fly Killer II	8.00 GHI
7	Golden Malrin with Muscamone	7.88 GHI
7	Golden Malrin with Muscamone	6.13 HI
3	Kill'em Fly Killer	0.50 I

Note: Treatment means without common letters are significantly different ($p < 0.05$).

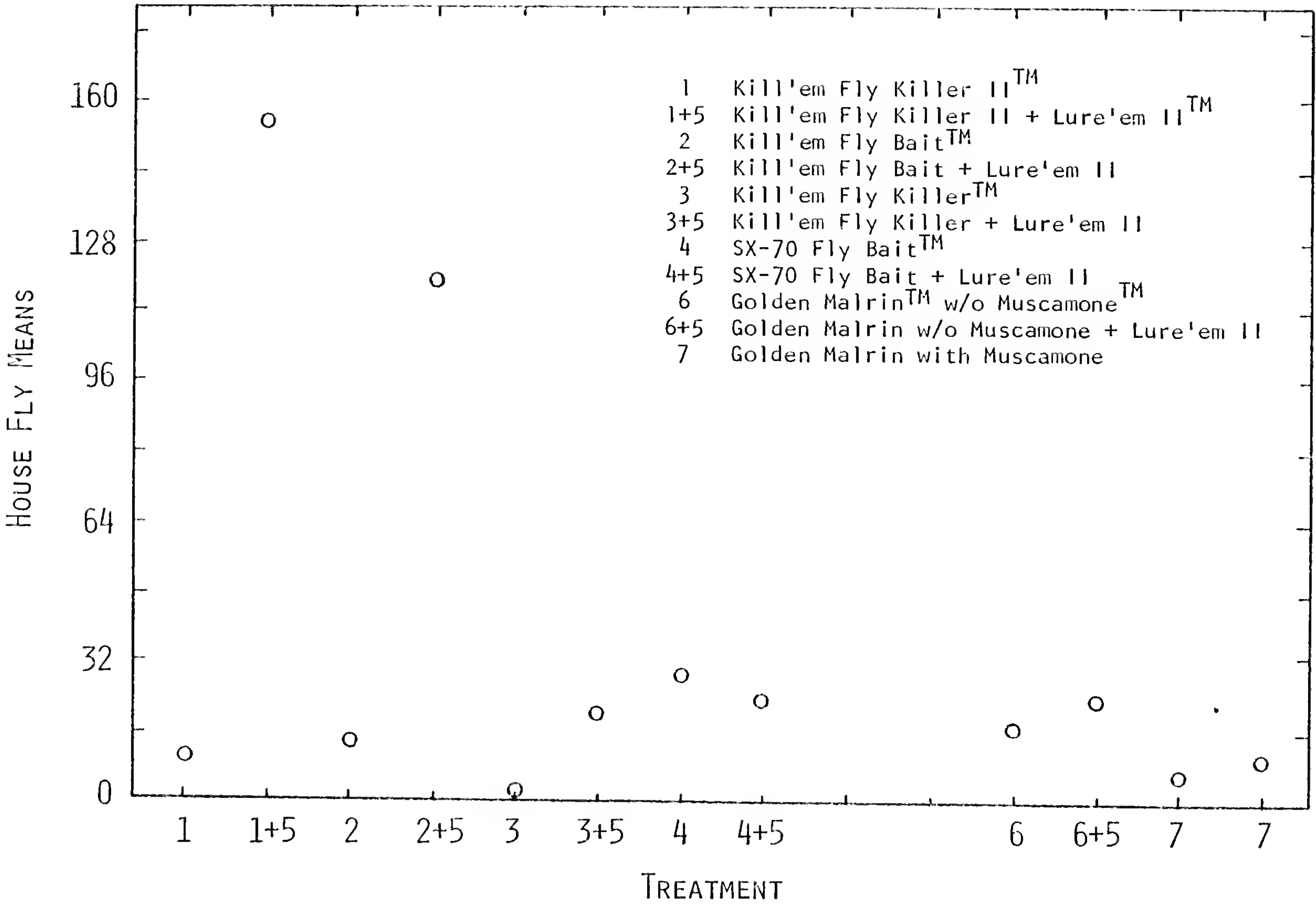


Figure 37. Farnam bait field trial treatment means.

Table 44. Treatment means by sex in Farnam bait field trial.

Treatment No.	Treatment	Sex	Treatment Mean
1	Kill'em Fly Killer II™	F	9.25*
		M	6.25*
1 + 5	Kill'em Fly Killer II + Lure'em II™	F	231.00†
		M	85.50†
2	Kill'em Fly Bait™	F	12.25
		M	10.50
2 + 5	Kill'em Fly Bait + Lure'em II	F	157.50
		M	74.75
3	Kill'em Fly Killer™	F	0.50
		M	0.50
3 + 5	Kill'em Fly Killer + Lure'em II	F	31.75
		M	7.50
4	SX-70 Fly Bait™	F	31.75
		M	25.50
4 + 5	SX-70 Fly Bait + Lure'em II	F	30.50*
		M	9.75*
6	Golden Malrin™ w/o Muscamone™	F	18.50
		M	13.00
6 + 5	Golden Malrin w/o Muscamone + Lure'em II	F	30.75
		M	9.25
7	Golden Malrin with Muscamone	F	9.25
		M	3.00
7	Golden Malrin with Muscamone	F	9.25
		M	6.50

* Indicates significant difference between sexes ($p < 0.05$).

† Indicates significant difference between sexes ($p < 0.01$).

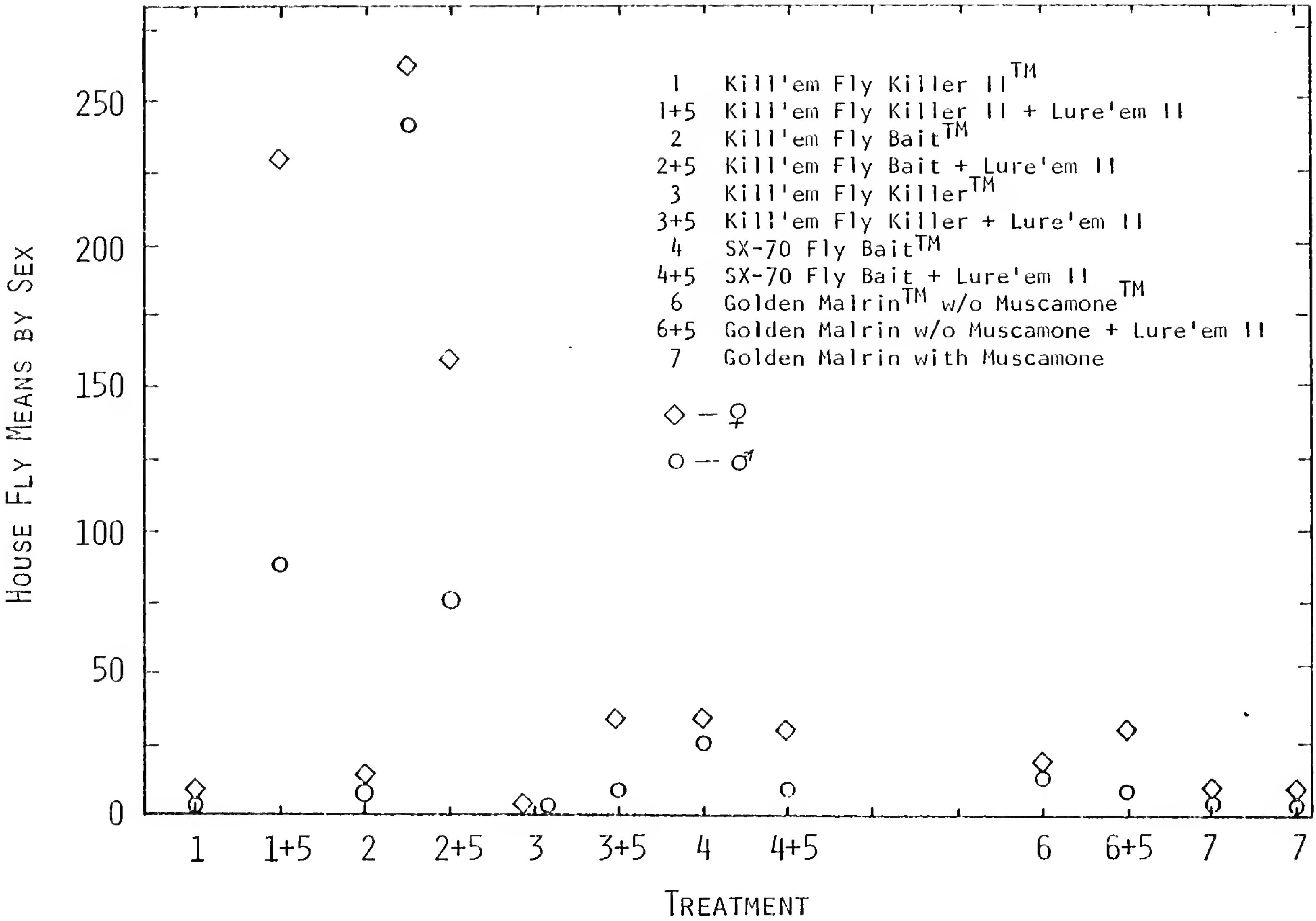


Figure 38. Farnam bait field trial treatment means by sex.

Muscamone plus Lure'em II attractant caught 3 times as many females as Golden Malrin with Muscamone.

Burroughs Wellcome bait. The bait submitted by Burroughs Wellcome was Atroban (BW 21Z) 0.25% sugar bait. Golden Malrin with Muscamone was used as a standard. These two baits were subjected to knockdown, residual, attractancy, and field tests. The amount of each bait used per replication was 5 g.

Knockdown test. Both baits required the same amount of time to produce mortality rates of 100% (Table 45). Atroban took much longer to actually kill flies than Golden Malrin with Muscamone, but after 60 min, flies exposed to Atroban were completely incapacitated. However, criterion for death was total lack of movement, and flies could not be considered dead until all movement ceased.

Residual test. Results are shown in Table 46. After 6 weeks of testing, Atroban and Golden Malrin with Muscamone baits were producing mortality rates of 87.5 and 100.0% respectively. Mortality was still occurring in the manner described in the knockdown test.

Attractiveness test. There were no dead flies in the Atroban bait station (Table 47). This indicates that Atroban was either unattractive to house flies or, due to the slow killing action of Atroban, flies died after leaving the bait station.

There were 25 dead flies in the Golden Malrin with Muscamone bait station.

Field test. Results are shown in Table 48. No flies were found in the Atroban bait stations, but a mean of 12.5 flies were found in the Golden Malrin with Muscamone bait stations. The ability of Atroban

Table 45. Results of the knockdown test using BW 21Z and Golden Malrin^{TM} with Muscamone^{TM} fly baits.

Time Interval	BW 21Z Replication A	B	C	D	BW 21Z Per cent Mortality	Golden Malrin with Muscamone Replication A	B	C	D	Golden Malrin with Muscamone Per cent Mortality	Control Replication A	B	C	D	Control Per cent Mortality
0[a]	0	0	0	0[c]	0.0[d]	0	0	0	0	0.0	0	0	0	0	0.0
1	0	0	0	0	0.0	2	0	1	4	17.5	0	0	0	0	0.0
2	0	1	0	0	2.5	2	0	1	4	17.5	0	0	0	0	0.0
3	0	1	0	1	5.0	5	0	1	4	25.0	0	0	0	0	0.0
4	0	2	0	1	7.5	5	0	2	4	27.5	0	0	0	0	0.0
5	0	2	0	1	7.5	5	0	3	4	30.0	0	0	0	0	0.0
6	0	2	0	1	7.5[e]	5	0	3	7	37.5	0	0	0	0	0.0
7	1	2	0	1	10.0	5	0	3	7	37.5	0	0	0	0	0.0
8[b]	1	4	0	2	17.5	5	0	3	7	37.5	0	0	0	0	0.0
9	4	4	2	3	32.5	6	4	7	7	60.0	0	0	0	0	0.0
10	6	6	5	6	57.5	8	7	7	9	77.5	0	0	0	0	0.0
11	8	8	6	6	70.0	9	7	8	9	82.5	0	0	0	0	0.0
12	9	9	8	8	85.0	9	9	9	10	92.5	0	0	0	0	0.0

Table 45. Continued.

Time Interval	Baits: BW 21Z Replication A	B	C	D	Per cent Mortality	Golden Malrin with Muscamone Replication A	B	C	D	Per cent Mortality	Control Replication A	B	C	D	Per cent Mortality
13	10	10	8	8	90.0	9	9	9	10	92.5	0	0	0	0	0.0
14	10	10	9	8	92.5	9	9	10	10	95.0	0	0	0	0	0.0
15	10	10	9	8	92.5	9	10	10	10	97.5	0	0	0	0	0.0
16	10	10	10	10	100.0	10	10	10	10	100.0	0	0	0	0	0.0

[a] Each number represents a 10-min interval.

[b] Each number represents a 30-min interval.

[c] Each number represents the number of flies killed out of 10.

[d] Per cent mortality = $\frac{\text{number dead}}{10} \times 100$

[e] All flies that were exposed to the BW 21Z bait were at this point either dead or completely incapacitated; since the criterion for death was not met, per cent mortality was figured at 7.5%.

Note: All flies used in this test were five-day-old female house flies, *Musca domestica* (L.), from a laboratory colony. Flies were sexed while anesthetized with CO_2.

Table 46. Results of the residual test using BW 21Z and Golden Malrin™ with Muscamone™ fly baits.

	Baits														
	BW 21Z					Golden Malrin with Muscamone					Control				
Time Interval	Replication A	B	C	D	Per cent Mortality	Replication A	B	C	D	Per cent Mortality	Replication A	B	C	D	Per cent Mortality
1[a]	10	9	10	10[b]	97.5[c]	10	10	9	10	97.5	0	0	0	0	0.0
2	10	10	10	10	100.0	10	10	10	10	100.0	3	0	0	0	7.5
3	3	9	10	10	80.0	10	10	10	10	100.0	0	0	0	0	0.0
4	10	7	8	9	85.0	9	9	10	9	92.5	2	0	0	0	5.0
6	5	10	10	10	87.5	10	10	10	10	100.0	0	0	0	0	0.0

[a] Each number represents the number of weeks after the completion of the knockdown test.

[b] Each number represents the number of flies killed out of 10.

[c] Per cent mortality = $\frac{\text{number dead}}{10} \times 100$

Note: All flies used in this test were five-day-old female house flies, *Musca domestica* (L.), from a laboratory colony. Flies were sexed while anesthetized with CO_2.

Table 47. Results of the attractiveness test using BW 21Z and Golden Malrin™ with Muscamone™ fly baits.

Bait	No. Flies Killed	Per cent Kill
BW 21Z	0[a]	0.0[b]
Golden Malrin with Muscamone	25	12.5

[a] Represents the number of flies killed out of 200.

[b] Per cent kill = $\frac{\text{number dead}}{200}$ X 100.

Note: All flies used in this test were five-day-old female house flies, *Musca domestica* (L.), from a laboratory colony. Flies were sexed while anesthetized with CO_2.

Table 48. Results of the field test using BW 21Z and Golden Malrin™ with Muscamone™ fly baits.

	Baits			
	BW 21Z		Golden Malrin with Muscamone	
Replicate	♂	♀	♂	♀
1	0	0	9	5
2	0	0	7	17
3	0	0	0	2
4	0	0	3	7
5	0	0	--	--[a]
Σ	0	0	19	31
Ave	0	0	4.75	7.75

[a] Bait station was lost.

to kill flies in the field was not demonstrated although its efficacy was adequately proven in the laboratory tests.

Contact Residuals

Adult house fly dosage-mortality curve for permethrin. Concentrations of permethrin (JFU 5819 25% permethrin EC, ICI Americas, Inc.) tested and responses are shown in Table 49. The probabilities, probits, log doses, and upper and lower fiducial limits are shown in Table 50. The dosage-mortality curve with fiducial limits is shown in Figure 39. The LD_{50} for JFU 5819 is 18.0 ppm and the regression equation is $Y = 14.25 + 1.46X$.

The high LD_{50} and the relatively shallow slope indicate that some amount of resistance to permethrin may already exist in this house fly strain and that more resistance is developing within the gene pool. Possible cross-resistance to chlorinated hydrocarbon insecticides may explain this phenomenon as indicated in the literature (Prasittisuk and Busvine, 1977).

The responses (Table 49) are not well distributed between the mortality values of 10 and 90%. This is due to the fluctuation in mortality rates produced by the same test concentrations on different generations of the house fly colony. After 10 to 15 trials, the set of data in Table 49 was the most reliable set produced.

Permethrin as a contact residual in the laboratory. Permethrin (JFU 5021A 2 lb/gal EC, ICI Americas, Inc.) was applied to cement, tin, and plywood templates to run-off at rates of 0.05 and 0.005%.

Female laboratory colony house flies, 3 to 5 days old, were transferred by use of a vacuum system to cylindrical window-screen cages 12.5 cm high and 7.0 cm in diameter. Open ends of the cages were placed

Table 49. Test concentrations and corresponding responses from the JFU 5819 laboratory bioassay.

Replication	Test Concentrations (ppm) 19	15	10	5	1	Control
1	6/8[a]	4/9	3/9	2/10	1/8	2/8
2	4/7	4/8	3/9	3/11	1/8	1/9
3	5/8	5/9	3/8	2/11	2/8	1/9
4	5/8	3/8	4/9	1/11	1/8	2/9
Σ	20/31	16/34	13/36	8/43	5/32	6/35
$\bar{x}$	5.00	4.00	3.25	2.00	1.25	1.50
% Mortality	64.52	47.06	37.14	18.60	15.63	17.14
Corrected % Mortality[b]	57.18	36.11	24.14	1.72	---	---

[a] Represents the number of flies killed out of the total number of flies tested.

[b] Corrected using Abbott's formula:

$$\frac{\text{\% treatment mortality} - \text{\% check mortality}}{100 - \text{\% check mortality}} \times 100$$

Table 50. The probabilities, probits, log doses, and upper and lower fiducial limits from the probit analysis of JFU 5819 dosage-mortality data.

OBS	PROB	PROBITS	LOGDOSE	LOWER	UPPER
1	0.01	2.67	-7.94	-11.15	-7.35
2	0.02	2.95	-7.76	-10.54	-7.23
3	0.03	3.12	-7.64	-10.15	-7.16
4	0.04	3.25	-7.55	-9.87	-7.10
5	0.05	3.36	-7.48	-9.63	-7.06
6	0.06	3.45	-7.42	-9.43	-7.10
7	0.07	3.52	-7.36	-9.26	-6.98
8	0.08	3.59	-7.31	-9.10	-6.95
9	0.09	3.66	-7.27	-8.96	-6.93
10	0.10	3.72	-7.23	-8.83	-6.90
11	0.15	3.96	-7.06	-8.29	-6.78
12	0.20	4.16	-6.93	-7.88	-6.69
13	0.25	4.33	-6.81	-7.53	-6.59
14	0.30	4.48	-6.71	-7.24	-6.49
15	0.35	4.61	-6.61	-6.99	-6.36
16	0.40	4.75	-6.52	-6.81	-6.20
17	0.45	4.87	-6.43	-6.67	-6.00
18	0.50	5.00	-6.35	-6.57	-5.77
19	0.55	5.13	-6.56	-6.48	-5.52
20	0.60	5.25	-6.17	-6.41	-5.25
21	0.65	5.39	-6.08	-6.34	-4.97
22	0.70	5.52	-5.99	-6.27	-4.67
23	0.75	5.67	-5.88	-6.20	-4.34
24	0.80	5.84	-5.77	-6.13	-3.98
25	0.85	6.04	-5.64	-6.04	-3.54
26	0.90	6.28	-5.47	-5.94	-3.00
27	0.91	6.34	-5.43	-5.91	-2.87
28	0.92	6.41	-5.38	-5.89	-2.72
29	0.93	6.48	-5.33	-5.86	-2.57
30	0.94	6.55	-5.28	-5.82	-2.39
31	0.95	6.64	-5.22	-5.79	-2.19
32	0.96	6.75	-5.15	-5.74	-1.95
33	0.97	6.88	-5.06	-5.69	-1.66
34	0.98	7.05	-4.94	-5.62	-1.28
35	0.99	7.33	-4.75	-5.50	-0.67

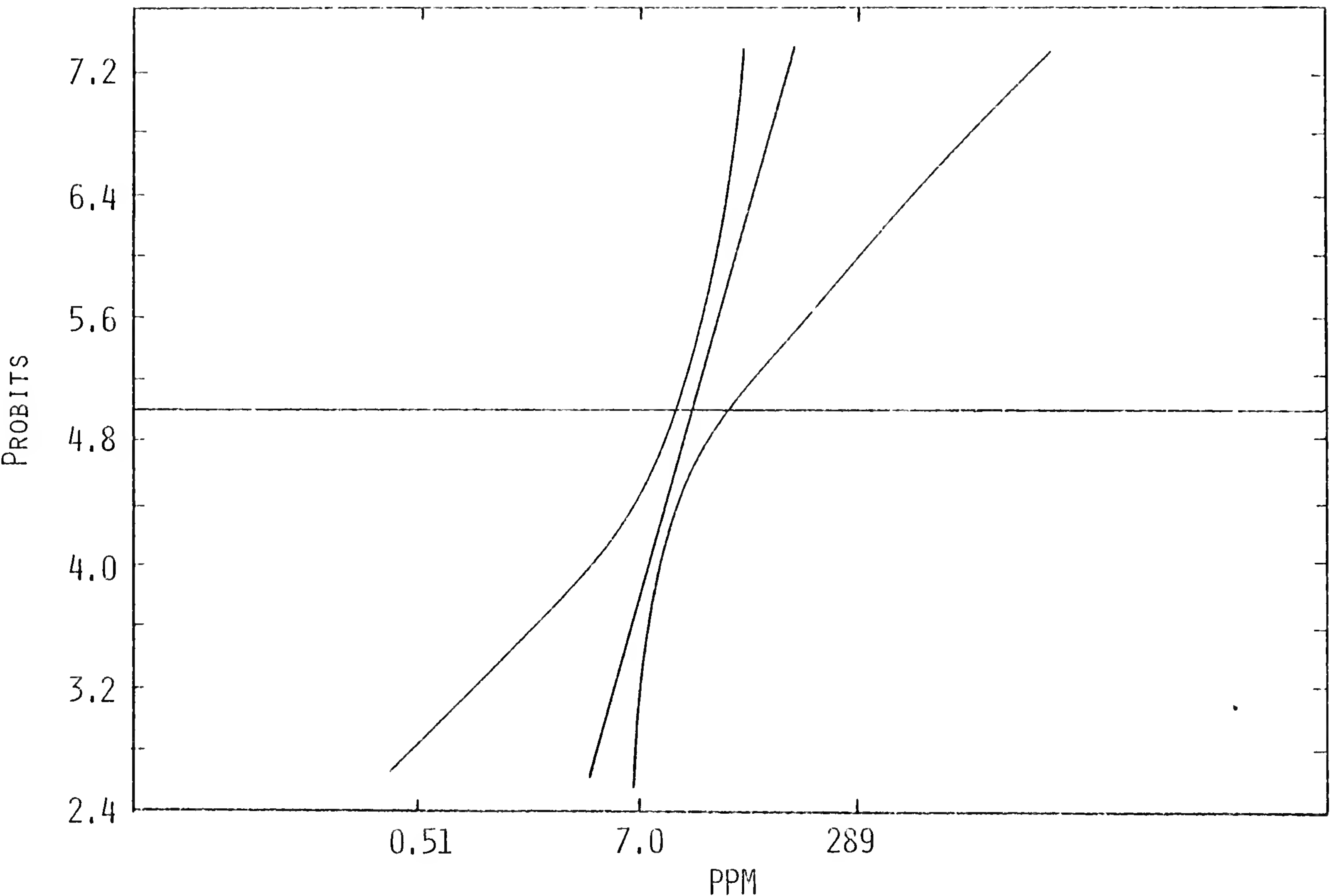


Figure 39. Probit curve, fiducial limits, and LC_{50} for JFU 5819 dosage-mortality data.

on the templates with four cages on each template and 10 flies per cage. A cotton ball soaked in a sucrose solution was placed on top of each cage as a food source. Mortality was recorded after 24 hr. Criterion for death was total lack of movement. Control templates were processed like the treated templates except that they were not sprayed with permethrin.

Testing continued at selected intervals until mortality dropped below 50%. Between tests, templates were stored outside under the eaves of the laboratory to simulate aging under field conditions. After each use, cages were baked in an oven at 148.9 C for a 24-hr period.

Results. Mortality on the tin templates dropped below 50% within 3 days (Table 51) and testing ceased after 8 days. Mortality on the cement block templates never exceeded 50% and testing was also discontinued after 8 days (Table 51).

The breakdown of JFU 5021A was slower on wood (Table 51). Mortality on the 0.005% template began to fluctuate 56 days post-treatment and dropped below 50% 75 days post-treatment. As a check, the template was tested again the following summer, but permethrin had broken down (Table 51) and testing was discontinued.

The 0.05% template continued to produce mortalities of 100% through 1976. When testing resumed in 1977, however, the mortality rate began to fluctuate (Table 51). Mortality dropped below 50% on 25 August and 8 November, but was back to 77.3% by 5 December. The reasons for this are not clear, but may be related to lower ambient testing temperature. The mean mortality for the year was 65.8%.

Table 51. Mortality and per cent mortality of house flies exposed to two levels of JFU 5021A applied as a contact residual on three different surfaces.

Date	Control		0.05%		0.005%	
	Mortality	Per cent Mortality	Mortality	Per cent Mortality	Mortality	Per cent Mortality
1976	Surface:	Tin	Application date:		9/15/76	
15 Sep	4/21	19.0	64/102	62.7	60/107	56.1
18 Sep	4/20	20.0	11/47	23.4	22/57	38.6
23 Sep	6/20	30.0	7/60	11.7	6/65	9.2
1976	Surface:	Cement block	Application date:		9/15/76	
15 Sep	3/23	13.0	58/118	49.2	40/88	45.6
18 Sep	4/25	16.0	18/64	28.1	11/47	23.4
23 Sep	1/20	5.0	5/60	8.3	5/53	9.4
1976	Surface:	Wood	Application date:		8/24/76	
14 Sep	7/65	10.8	45/45	100.0	58/58	100.0
15 Sep	15/122	11.5	113/121	93.4	68/113	60.2
29 Sep	3/50	6.0	53/57	93.0	10/54	18.5
19 Oct	3/31	9.7	37/37	100.0	25/43	58.1
7 Nov	10/80	12.5	81/81	100.0	40/86	46.5
1977	Surface:	Wood	Application date:		8/24/76	
1 Jul	8/49	16.3	41/47	87.2	3/46	6.5
25 Aug	0/46	0.0	23/49	46.9	0/39	0.0
20 Sep	14/62	22.6	36/54	66.7	Testing Discontinued	
11 Oct	5/50	10.0	37/52	71.2		
8 Nov	5/51	9.8	28/62	45.2		
5 Dec	10/48	20.8	33/45	77.3		
				$\overline{x} = 65.8$		

Table 51. Continued.

Date	Control		0.05%		0.005%	
	Mortality	Per cent Mortality	Mortality	Per cent Mortality	Mortality	Per cent Mortality
1978	Surface: Wood		Application date: 8/24/76			
31 Jan	6/42	14.3	35/42	83.3		
28 Feb	0/36	0.0	29/39	74.4		
21 Mar	1/39	2.6	26/37	70.8		
9 Jun	1/42	2.7	24/48	50.0		
6 Jul	0/33	0.0	5/40	12.5		
3 Aug	1/40	2.5	1/40	2.5		

Testing continued in 1978. Mortality was 50% on 9 June and dropped to 2.5% by 3 August. At this point the experiment was terminated. Permethrin at 0.05% was effective on wood for almost 23 months.

Fluctuation of control mortality throughout the experiment can only be attributed to an interaction between the flies and the untreated template surfaces.

Synthetic pyrethroids as contact residuals in the field. Three synthetic pyrethroid compounds, SD 43775 (Shell Development Co., 25% WP and 10% EC), BW 21Z (Burroughs Wellcome Co., 25% WP and 42.5% EC), and ICI 143 (ICI Americas, Inc., 5% EC), were applied to wooden panels in the amounts shown in Table 52.

Several weeks after panels were installed at the tilling site, there was no evidence to indicate that the compounds were killing flies as no dead flies were found in the aluminum guttering. In order to test the efficacy of the compounds, 3- to 5-day-old female laboratory colony house flies were processed and exposed to panel surfaces in the following manner. Flies were sexed while anesthetized with CO_2 and placed 10 to a cage in cages made from 180-ml plastic cups with lids. Cup bottoms had been removed and replaced with disks of window screen. In the field, cages were slipped under 23-cm lengths of 3-cm wide elastic that had been stapled to the panels for the purpose of holding the cages tightly to the surface. Lids were removed as cages were secured on the panels. One cage was attached to each panel for a 24-hr period. Criterion for death was total lack of movement.

Results. All compounds produced mortality rates up to 100% at 121 days post-treatment (Table 53). Knockdown was usually within 15 min though the onset of death required a much longer period. Flies fluttered

Table 52. Names, formulations, test concentrations, mixing instructions, and application rates of compounds applied to wooden panels.

Panel No.	Compound Name	Formulation and Type	Test Concentration (%)	Amount used to make 500 ml (g)	Amount applied to each Side of Panel (ml)	Application Instructions
C	Control	---	---	---	---	---
1	Shell 43775	25.0% WP	0.05	1.0	95.0	Apply to runoff
2	Shell 43775	10.0% EC	0.05	2.5	95.0	Apply to runoff
3	BW 21Z	25.0% WP	0.50	10.0	95.0	Apply to runoff
4	BW 21Z	25.0% WP	0.25	5.0	95.0	Apply to runoff
5	BW 21Z	25.0% WP	0.125	2.5	95.0	Apply to runoff
6	BW 21Z	42.5% EC	0.50	5.9	95.0	Apply to runoff
7	BW 21Z	42.5% EC	0.25	2.9	95.0	Apply to runoff
8	BW 21Z	42.5% EC	0.125	1.5	95.0	Apply to runoff
9	ICI 143	5.0% EC	0.50	50.0	41.0	Apply @ 1 gal/750 ft^2

Note: Compounds were mixed in water.

Table 53. Total and per cent mortality that occurred when 3- to 5-day-old female house flies were exposed to synthetic pyrethroids on wooden panels.

		Treatment Number									
		C	1	2	3	4	5	6	7	8	9
Days Post-treatment:	86	0/10	8/10	6/10	10/10	10/10	10/10	9/10	6/10	7/10	8/10
		0/10	9/10	10/10	10/10	1/4	9/10	9/10	9/10	9/10	9/10
		1/10	10/10	9/10	10/10	10/10	8/10	10/10	10/10	9/10	9/10
		0/10	8/10	6/10	10/10	2/2	10/10	10/10	8/10	10/10	9/10
Total Mortality		1/40	35/40	31/40	40/40	25/26	37/40	38/40	33/40	35/40	35/40
% Mortality		2.5	87.5	77.5	100.0	88.5	92.5	95.0	82.5	87.5	87.5
Days Post-treatment:	100	1/10	10/10	10/10	10/10	10/10	10/10	10/10	9/10	9/10	10/10
		0/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		4/10	10/10	10/10	10/10	10/10	8/10	10/10	10/10	10/10	9/10
		6/10	9/10	8/10	10/10	9/10	9/10	9/10	10/10	10/10	10/10
Total Mortality		11/40	39/40	38/40	40/40	39/40	37/40	39/40	39/40	39/40	39/40
% Mortality		27.5[a]	97.5	95.0	100.0	97.5	92.5	97.5	97.5	97.5	97.5

Days Post-treatment:	116	8/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		9/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		7/9	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
Total Mortality		34/39	40/40	40/40	40/40	40/40	40/40	40/40	40/40	40/40	40/40
% Mortality		87.2[b]	100.0	100.0	100.0	100.0	100.0	100.0	100.0	100.0	100.0
Days Post-treatment:	121	0/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		0/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		0/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
		0/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10	10/10
Total Mortality		0/40	40/40	40/40	40/40	40/40	40/40	40/40	40/40	40/40	40/40
% Mortality		0.0	100.0	100.0	100.0	100.0	100.0	100.0	100.0	100.0	100.0

[a] Two replications came in contact with pyrethrin fog when the producer fogged his egg room.

[b] Temperature was ca. 85 F (29.4 C), very hot and very dry.

Note: Treatment numbers are as follows: C = control, 1 & 2 = SD 43775, 3 through 8 = BW 21Z, 9 = ICI 143.

randomly throughout their cages and did not necessarily remain in contact with the treated surface. No further contact with the panels was needed to cause the flies to die.

At 100 days post-treatment, the poultryman fogged one end of his poultry house while a fly trial was in progress and the mortality rates of two control panels were affected (Table 53).

When a trial was performed 116 days post-treatment, ambient temperature was 29.4 C, but temperatures inside the poultry houses were above 35.0 C. High control mortality during this trial was attributed to the heat.

When an additional trial was attempted ca. 325 days post-treatment, a new flock of chickens had been housed on the farm. In between flocks, houses and panels had been thoroughly sprayed with a disinfectant and when flies were exposed to the panels, mortality was zero in all cases.

Northern Fowl Mites

Dosage-Mortality Curves for Selected Acaricides

Compounds tested were four synthetic pyrethroids [SBP-1382 (Penick 24.3% EC), BW 21Z (Burroughs Wellcome 42.5% EC), SD 43775 (Shell 10% WDL), and Ectiban (ICI Americas 26.0% EC)], Sevin (Union Carbide 99.9% crystaline powder), and malathion (American Cyanamid 96.4% EC).

Results. The test concentrations of acaricides and the total, per cent, and corrected per cent mite mortality for each concentration tested are shown in Table 54. Figure 40 shows all six dosage-mortality curves plotted on one set of axes and the regression equations and LC_{50}'s are shown in Table 55. Probabilities, probits, log doses, and upper and lower fiducial limits are shown in Appendices 4 through 9.

Table 54. Concentrations of acaricides and total, per cent, and corrected per cent mortality for each concentration tested against northern fowl mites.

Test Concentration (ppm)	Total Mortality	Per cent Mortality	Corrected % Mortality
	Penick 1382™		
10.00	54/60	90.00	88.10
8.00	54/61	88.52	86.90
6.00	40/62	64.52	58.33
4.00	38/61	62.30	54.76
2.00	25/60	41.67	30.95
1.00	25/62	40.32	28.57
Control	10/61	16.39	---
	BW 21Z		
4.25	44/60	73.33	70.99
2.75	33/60	55.00	51.06
1.75	26/60	43.33	38.36
1.25	19/60	31.67	25.68
1.00	15/60	25.00	18.43
Control	5/62	8.06	---
	Shell SD 43775		
10.00	51/60	85.00	82.74
8.00	45/60	75.00	71.23
6.00	40/60	66.67	61.64
4.00	29/60	48.33	40.53
2.00	21/60	35.00	25.19
1.00	12/61	19.67	7.55
Control	8/61	13.11	---

Table 54. Continued.

Test Concentration (ppm)	Total Mortality	Per cent Mortality	Corrected % Mortality
	ICI Ectiban™		
6.00	52/60	86.67	85.19
4.00	43/60	71.67	68.52
2.00	34/60	56.67	51.86
1.00	27/60	45.00	38.89
0.75	21/60	35.00	27.78
0.50	13/60	21.67	12.97
Control	6/60	10.00	---
	Carbaryl		
10.00	48/60	80.00	77.36
1.00	41/60	68.33	64.15
0.50	40/60	66.67	62.27
0.25	31/60	51.67	45.28
0.10	20/60	33.33	24.52
Control	7/60	11.67	---
	Malathion		
10.00	52/60	86.67	85.46
7.00	48/60	80.00	78.18
4.00	35/60	58.33	54.54
1.00	29/60	48.33	43.63
0.50	16/60	26.67	20.01
0.10	13/60	21.67	14.55
Control	5/60	8.33	---

Note: Each test concentration was replicated four times using 15 mature female northern fowl mites per replication.

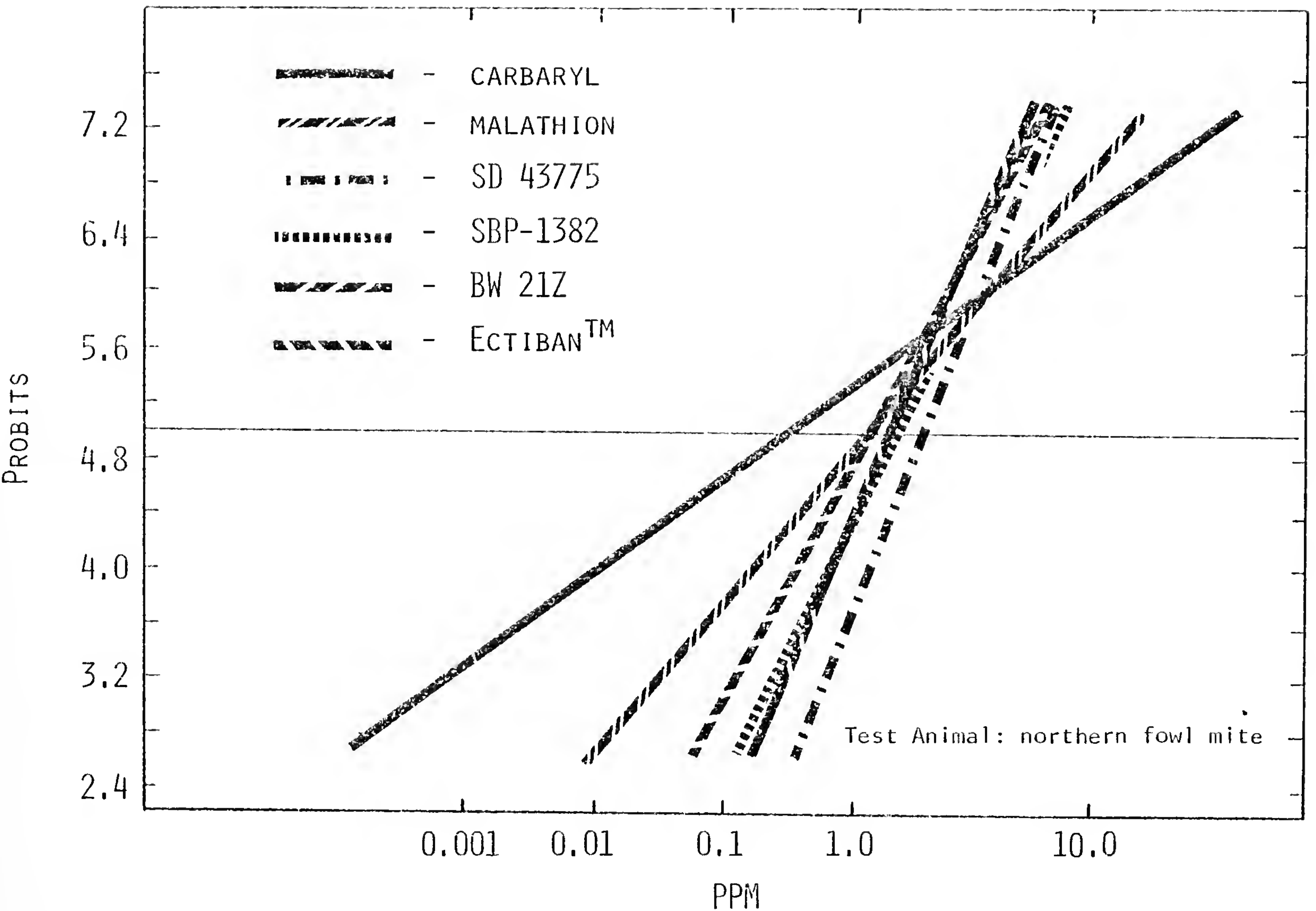


Figure 40. Probit curves for all acaricides tested, plotted on one set of axes.

Table 55. LC_{50}'s and regression equations for the acaricides tested.

Acaricide	LC_{50} (ppm)	Regression Equation
SBP 1382™	3.0	$Y = 11.46 + 0.80X$
BW 21 Z	2.5	$Y = 13.02 + 0.97X$
SD 43775	4.4	$Y = 12.70 + 0.99X$
Ectiban™	1.8	$Y = 11.51 + 0.75X$
Carbaryl	0.4	$Y = 7.90 + 0.29X$
Malathion	1.7	$Y = 9.15 + 0.48X$

Note: Acaricides were tested against the northern fowl mite.

The LC_{50}'s of the four synthetic pyrethroid compounds were very closely grouped (Table 55). Ectiban had the lowest LC_{50} at 1.8 ppm and SD 43775 had the highest at 4.4 ppm. The slopes of the synthetic pyrethroid curves were also closely grouped and approaching 1.0 (Table 55, Figure 40). This indicates that the susceptibility of the mite populations to these compounds was relatively heterogeneous. This may be due to chlorinated hydrocarbon cross-resistance which has been reported for other acari (Nolan et al., 1977).

The LC_{50} of malathion at 1.7 ppm was only slightly lower than the LC_{50} of Ectiban, but the LC_{50} of Sevin at 0.4 ppm was lower than the LC_{50} of SD 43775 by a factor of 10. The slopes of the malathion and Sevin curves were less than 0.5, thus the curves were flatter than the synthetic pyrethroid curves (Table 55, Figure 40). This indicates that the susceptibility of the mite populations to these two compounds was more heterogeneous than it was for the synthetic pyrethroids, and that the mite populations are in the process of becoming more resistant to Sevin and malathion (Hoskins and Gordon, 1956). Northern fowl mite resistance to malathion was recorded by Hall et al. (1978).

Although Sevin had the lowest LC_{50}, it had the highest LC_{90} of all compounds tested (Figure 40). In order to produce mortalities of up to 100% in the field, the concentration of Sevin would have to be much greater than the LC_{100} concentrations of malathion or the synthetic pyrethroids.

<u>Control of Endemic Florida Strains of Northern Fowl Mites with Carbaryl, Malathion, and Ravap</u>

Data were collected from one hen in each of the first 120 cages on the north side of the four California-style houses at the tilling site.

All three hens in each cage were examined initially, and the one having the highest mite population was selected as the test animal. For identification purposes, test animals were marked on the wing feathers with a water-proof black magic marker. A total of 480 hens was used in the experiment.

The formulations and mixing instructions, test concentrations and application rates for the acaricides tested are shown in Table 56. The treatment schedule is shown in Table 57. Malathion in treatment 1 was incorrectly mixed and applied before the error was discovered, as frequently occurs on commercial poultry operations. A second malathion treatment was added with malathion mixed at the proper concentration. Both treatments were maintained throughout the experiment.

On the day before the initial application of acaricides, a mite precount was made. Two weeks after making the precount, a second mite count was made. The following day, the second treatment was applied. Two weeks after making the second mite count, a third count was made, followed by a third treatment the following day. Two weeks after the third mite count was made, a fourth and final count was made.

Results. Weekly mite population means and converted population means are shown for each treatment in Table 58. A plot of mite population means per hen by treatment is shown in Figure 41.

There were significant differences between precount mite population means in the treatment groups (Table 58). This was unimportant, however, because the precount mite mean for the control group was in between the highest and lowest precount mite means, and because the final mite counts for all groups were significantly lower than the precount of the control group.

Table 56. The formulations, mixing procedures, test concentrations, and application rates for acaricides tested at the tilling site for control of northern fowl mites.

Acaricide	Formulation	Volume of Pesticide	Volume of H_2O (ml)	Test Concentration (%)	Application Rate
Malathion	56.1% EC	30 ml[a]	3840	0.44	3840 ml/60 to 80 birds[b]
Carbaryl	50.0% WP	22.7 g	3840	0.30	3840 ml/60 to 80 birds
Ravap[TM]	28.7% EC	48 ml	3840	0.36	3840 ml/60 to 80 birds

[a] Malathion used in week 0 of treatment 1 was 6 times (6X) this volume.

[b] Even though each replication contained 20 test birds, 60 birds were actually present and taken into account during acaricide application.

Table 57. Treatment schedule of acaricides tested at the tilling site for northern fowl mite control.

Treatment No.	Week 1	Week 3	Week 5
1	Malathion (6X)	Malathion (1X)	Malathion (1X)
2	Control	Control	Control
3	Carbaryl	Carbaryl	Carbaryl
4	Carbaryl	Carbaryl	Carbaryl
5	Ravap^{TM}	Ravap	Ravap
6	Malathion (1X)	Malathion (1X)	Malathion (1X)

Table 58. Mite population means and converted population means from hens treated with malathion, carbaryl, and Ravap™ at the tilling site for northern fowl mite control.

Count Number	Treatment[a] 1	2	3	4	5	6
	Mite population means per hen from field counts					
1 (Precount)	179.36A	116.88BCD	102.88C	181.46A	160.61AB	82.63D
2	20.43CDE	110.78A	50.89B	28.01CD	3.63E	34.00BC
3	19.00C	82.94A	8.23CD	34.20B	0.46D	15.81CD
4	6.81B	58.69A	3.42B	2.22B	3.13B	0.00B
	Converted population means per hen[b]					
1 (Precount)	1458.40	950.19	836.37	1478.64	1305.78	671.74
2	166.06	900.60	413.71	227.75	29.55	276.42
3	154.47	674.29	66.89	278.07	3.76	128.48
4	55.39	477.13	27.79	18.01	25.41	0.00

[a]Treatment 1 = malathion(6X)-malathion(1X)-malathion(1X).
Treatment 2 = control-control-control.
Treatment 3 = carbaryl-carbaryl-carbaryl.
Treatment 4 = carbaryl-malathion(1X)-carbaryl.
Treatment 5 = Ravap-Ravap-Ravap.
Treatment 6 = malathion(1X)-malathion(1X)-malathion(1X).

[b]Converted population mean = mite population mean from field count X 8.13.

Note: Mite population means per hen from field counts in the same row having unlike letters are significantly different ($p < 0.05$) using the method of J. W. Tukey.

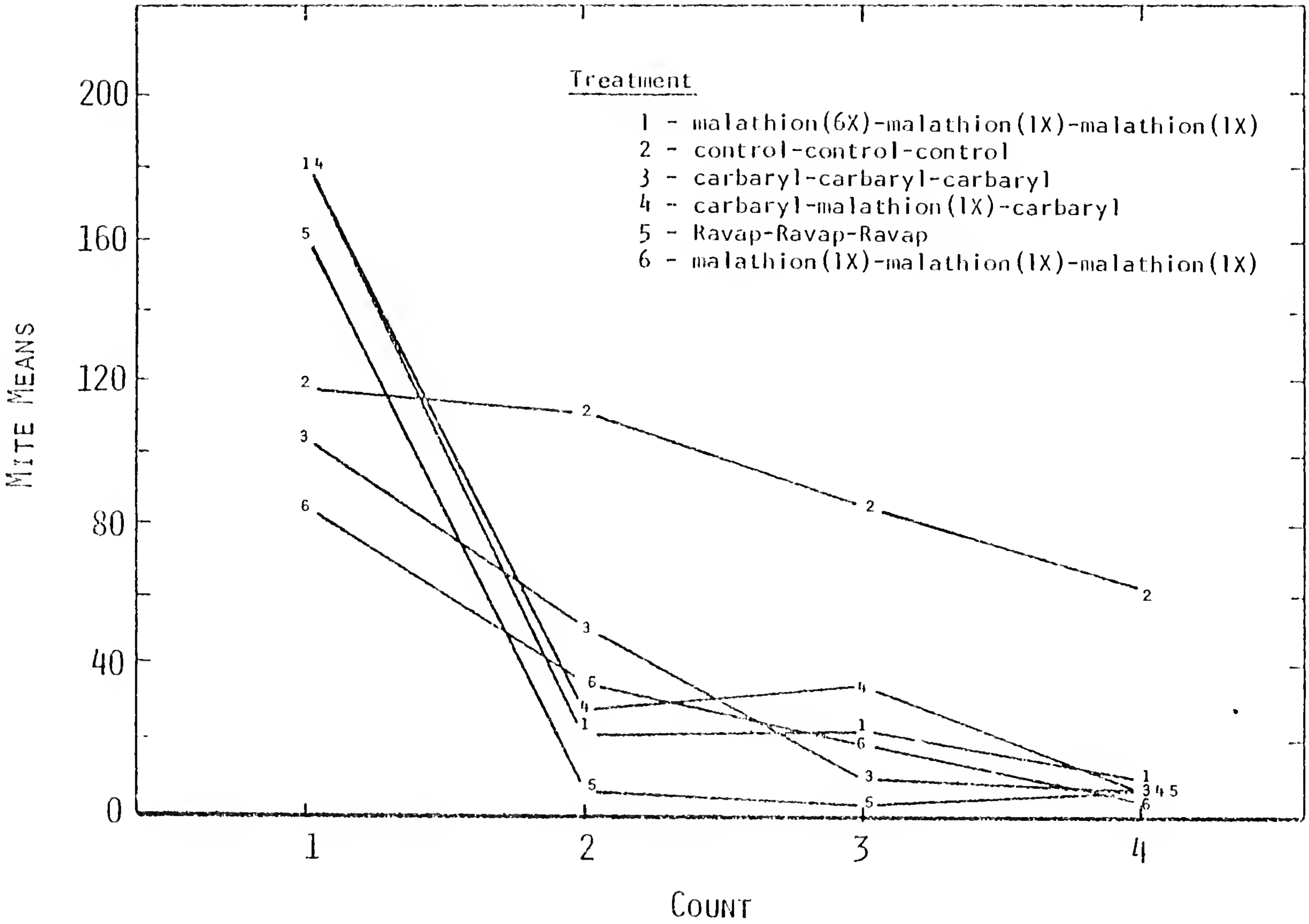


Figure 41. Mite population means from hens treated with malathion, carbaryl, and Ravap^{TM} at the tilling site.

Two weeks after the initial treatments were applied, the mite population means for all treatment groups were significantly lower than the mite population mean of the control group (Table 58, Figure 41). Significant differences also occurred between the means of treatment groups. The Ravap treatment group mite mean of 3.63 mites per bird was numerically lower than the mite means of the other groups. The next highest group was the 6X malathion group with a count of 20.43 mites per bird. The remaining groups were the carbaryl-malathion(1X)-carbaryl, malathion (1X)-malathion(1X)-malathion(1X), and carbaryl-carbaryl-carbaryl groups. The mite mean of the control was 110.78 mites per bird.

Two weeks after the second treatments were applied, the mite means for all treatment groups were still significantly lower than the mite mean of the control group (Table 58, Figure 41). Significant differences also occurred between means of treatment groups. The Ravap treatment group mite mean of 0.46 mites per bird was numerically lower than the mite means of the other groups. The next highest counts were in the carbaryl-carbaryl-carbaryl group, followed by the two malathion groups and the carbaryl-malathion(1X)-carbaryl group. The mite mean of the control group was 82.94 mites per bird.

At the time of the last mite count, mite population means of all treatment groups were significantly lower than the mite population mean of the control group (Table 58, Figure 41) and no significant differences occurred between treatment groups. The malathion(1X)-malathion(1X)-malathion(1X) group mite population mean was reduced to zero. The next highest group was the carbaryl-malathion(1X)-carbaryl group followed by the Ravap-Ravap-Ravap, carbaryl-carbaryl-carbaryl, and malathion(6X)-

malathion(1X)-malathion(1X) groups. The control group had a mean mite count of 58.69 mites per bird.

All treatments significantly reduced mite populations in 6 weeks, but Ravap reduced mite populations more dramatically than the other acaricides and in a shorter period of time.

Efficacy of Two Synthetic Pyrethroid Compounds Against Northern Fowl Mites on Laying Hens in Floor Pens

Seventy-five 41-week-old White Leghorn laying hens at the University of Florida Poultry Science Research Farm in Chipley, Florida, were randomly divided into three groups of 25 hens each. Groups were housed in pens measuring 3.65 by 3.65 m with wood shavings as floor litter. Natural light was supplemented with morning and evening incandescent lights so hens were exposed to 15 hr of light per day. A 16.9% protein laying mash and water were provided free choice. Eggs were collected daily by pen.

The synthetic pyrethroid test materials, mixing procedures, and application rates are shown in Table 59. Immediately following a pre-treatment mite count on each hen, treatments were applied. A post-treatment mite count was conducted 36 days later. Prior to treatment, birds were leg-banded to identify them during the pre- and post-treatment mite counts.

Results. Pre- and post-treatment field-estimated and converted mite population counts and treatment means for each treatment are shown in Table 60. Pre- and post-treatment field-estimated mite population means are represented graphically in Figure 42.

There were no significant differences between pretreatment mite population means (Figure 42), and the mite populations on the control

Table 59. Formulations, mixing procedures, and application rates for synthetic pyrethroids applied to floor birds in Chipley, Fl., for northern fowl mite control.

Synthetic Pyrethroid	Volume of Pesticide(ml)	Volume of Water(l)	Final Solution(%)	Application Rate
BW 21Z (42.5% EC)	4.5	3.84	0.05	3.84 l/100 birds
SD 43775 (42.5% EC)	9.0	3.84	0.10	3.84 l/100 birds

Table 60. Pre- and post-treatment field-estimated and converted mite population counts and treatment means for each treatment from floor birds treated with two synthetic pyrethroids in Chipley, Fl., for northern fowl mite control.

Observation	Pretreatment Counts		Post-treatment Counts	
	Field Estimates	Converted[a]	Field Estimates	Converted
		Treatment 1 (BW 21Z)		
1	999	8121.87	0	0
2	500	4065.00	0	0
3	100	813.00	0	0
4	100	813.00	0	0
5	25	203.25	0	0
6	50	406.50	0	0
7	100	813.00	0	0
8	75	609.75	0	0
9	100	813.00	0	0
10	100	813.00	0	0
11	100	813.00	0	0
12	250	2032.50	0	0
13	100	813.00	0	0
14	110	894.30	0	0
15	50	406.50	0	0
16	130	1056.90	0	0
17	100	813.00	0	0
18	300	2439.00	0	0
19	65	528.45	0	0
20	55	447.15	0	0
21	175	1422.75	0	0
22	100	813.00	0	0
23	110	894.30	0	0
24	150	1219.50	0	0
25	100	813.00	0	0
Mite Mean	161.76	1315.11	0	0

Table 60. Continued.

Observation	Pretreatment Counts		Post-treatment Counts	
	Field Estimates	Converted	Field Estimates	Converted
		Treatment 2 (SD 43775)		
1	25	203.25	0	0
2	500	4065.00	0	0
3	100	813.00	0	0
4	30	243.90	0	0
5	75	609.75	0	0
6	120	975.60	0	0
7	120	975.60	0	0
8	200	1626.00	0	0
9	100	813.00	0	0
10	100	813.00	0	0
11	50	406.50	0	0
12	150	1219.50	0	0
13	75	609.75	0	0
14	150	1219.50	0	0
15	200	1626.00	0	0
16	95	772.35	0	0
17	150	1219.50	0	0
18	30	243.90	0	0
19	65	528.45	0	0
20	150	1219.50	0	0
21	50	406.50	0	0
22	50	406.50	0	0
23	150	1219.50	0	0
24	75	609.75	0	0
25	35	284.55	0	0
Mite Mean	113.0	918.69	0	0

Table 60. Continued.

Observation	Pretreatment Counts		Post-treatment Counts	
	Field Estimates	Converted	Field Estimates	Converted
		Treatment 3 (control)		
1	50	406.50	125	1016.25
2	75	609.75	175	1422.75
3	35	284.55	75	609.75
4	35	284.55	90	731.70
5	110	891.00	75	609.75
6	25	203.25	65	528.45
7	65	528.45	300	2439.00
8	50	406.50	95	772.35
9	55	447.15	175	1422.75
10	25	203.25	95	772.35
11	56	455.28	185	1504.05
12	125	1016.25	300	2439.00
13	100	813.00	150	1219.50
14	180	1463.40	95	772.35
15	100	313.00	275	2235.75
16	75	609.75	255	2073.15
17	300	2439.00	250	2032.50
18	50	406.50	90	731.70
19	300	2439.00	375	3048.75
20	75	609.75	275	2235.75
21	110	891.00	300	2439.00
22	75	609.75	400	3252.00
23	150	1219.50	800	6504.00
24	50	406.50	100	813.00
25	200	1626.00	85	691.05
Mite Mean	98.84	803.57	208.20	1692.67

[a] Converted mite count = field estimation X 8.13.

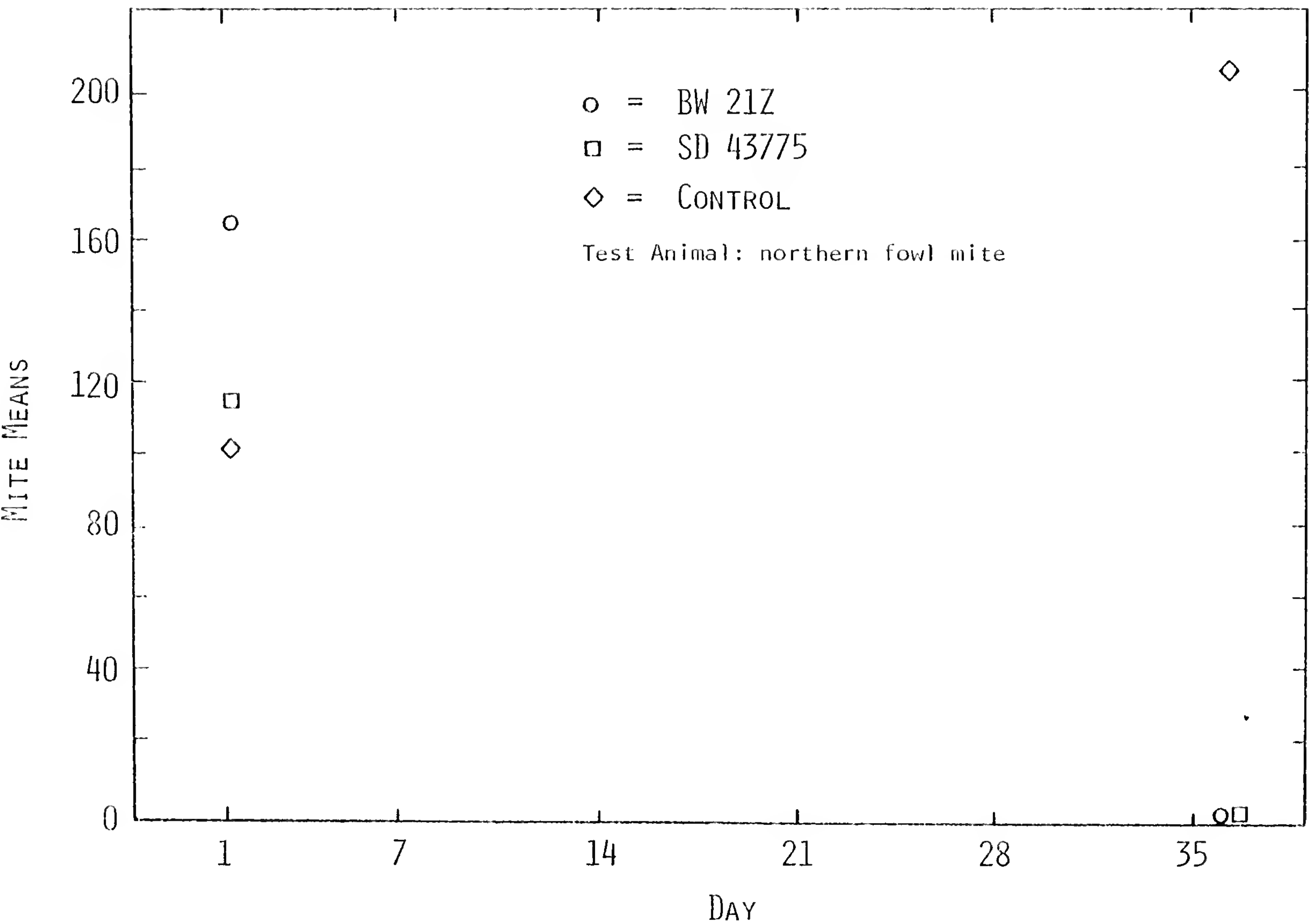


Figure 42. Pre- and post-treatment field-estimated mite population means from floor birds treated with two synthetic pyrethroid compounds at Chipley, Fl.

birds were numerically lower than the mite populations on birds in the two treatment groups.

On day 36, there were significant differences between the mite population means on the treated birds and the control birds (Figure 42). Mite populations on the treated birds were reduced to zero, but the mite populations on the control birds averaged 208.2 per bird.

The mean egg production by treatment by day is shown in Table 61. Analysis of production data was run by day and by the 36-day production mean. There was no significant difference in production due to treatment shown by either analysis.

The Effects of Northern Fowl Mites on Egg Production

Twelve commercial strains of White Leghorn laying hens on the University of Florida Poultry Science Research Farm in Chipley, Florida, were used to evaluate the effects of northern fowl mites on egg production. Birds were housed two to a cage in cages measuring 31 by 25 by 46 cm and were 41 weeks old when the experiment began.

Two houses were utilized for the trial. House 200 was a California-style house 82.3 m long by 3.7 m wide with the center aisle on an east-west axis. House 100 was 91.4 m long, 9.1 m wide, and oriented parallel to house 200. House 100 was partially enclosed with aluminum siding and wild-bird-proofed with 1-cm hardware cloth. All the hens in house 200 and the caged layers housed in the west end of house 100 were utilized for the experiment. The number of hens tested in each house was the same.

Both houses contained four rows of stair-step cages (Figure 43). Each row was divided into twelve 24-cage blocks with 12 cages on the upper row directly above 12 on the lower row. The 12 strains of hens

Table 61. Daily egg production means of birds treated with two synthetic pyrethroids in Chipley, Fl.

Day	Treatment 1	Treatment 2	Treatment 3
2	0.24	0.24	0.56
3	0.56	0.84	1.04
4	0.92	0.80	0.80
5	0.80	0.84	0.80
6	0.64	0.64	0.68
7	0.92	0.88	0.76
8	0.72	0.48	0.88
9	0.40	0.88	0.32
10	1.00	0.80	0.76
11	0.76	0.76	0.64
12	0.80	0.76	0.60
13	0.76	0.52	0.76
14	0.92	0.92	0.80
15	0.52	0.52	0.60
16	0.80	0.80	0.60
17	0.60	0.44	0.56
18	0.84	0.64	0.76
19	0.84	0.56	0.92
20	0.60	0.48	0.44
21	0.80	0.76	0.80
22	0.92	0.80	0.76
23	0.56	0.56	0.36
24	0.72	0.68	0.52
25	0.68	0.44	0.56
26	0.60	0.84	0.80
27	1.20	0.84	1.00
28	0.64	0.68	0.52
29	0.68	0.56	0.72

Table 61. Continued.

	Treatment		
Day	1	2	3
30	0.80	0.56	0.72
31	0.64	0.40	0.58
32	0.88	0.84	0.72
33	0.68	0.64	0.64
34	0.72	0.24	0.36
35	0.96	0.48	0.56
36	0.52	0.60	0.40

Note: Treatment 1 = BW 21Z at 0.05%
Treatment 2 = SD 43775 at 0.10%
Treatment 3 = Control
Treatments were applied to floor-housed birds.

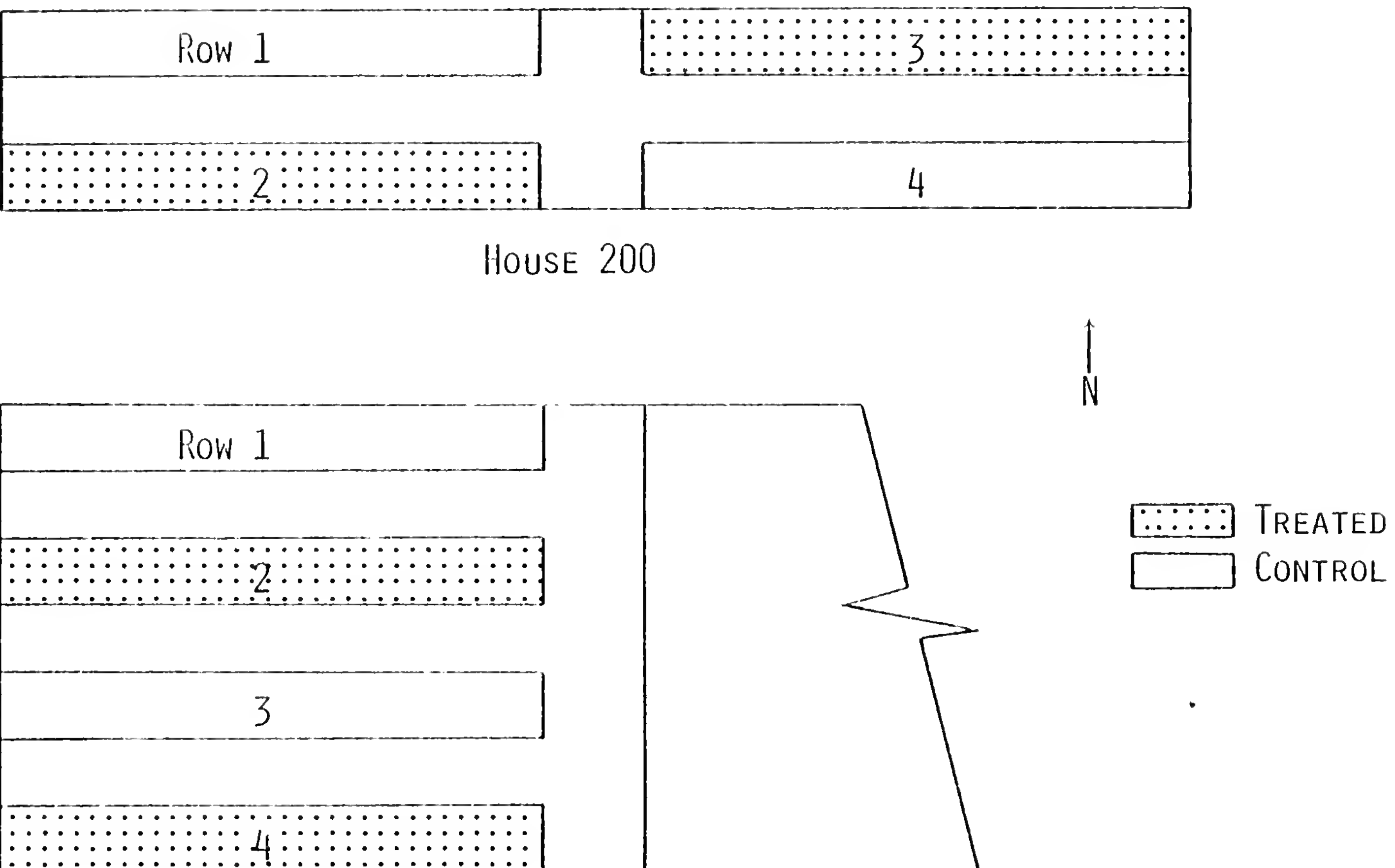


Figure 43. Houses 100 and 200 showing locations of strain replications and treatment areas.

were randomized throughout the houses so that each row of cages housed one 24-cage block of each strain. Blocks were divided in half. Hens in one half received a diet with a varying protein level and hens in the other half received a diet with a fixed protein level.

All hens on the lower rows of cages in house 200 were examined for northern fowl mites. One bird in each cage was marked on the wing with a black water-proof magic marker so both birds could be identified individually during each mite examination. Mite populations were counted on the day of but prior to the first acaricide treatment and again 36 days later (Table 62). Mites on birds in house 100 were not monitored.

Egg production records were maintained in both houses. Eggs were collected daily and production was calculated on a hen-day basis for both feed protein levels within each block. Because of results from previous trials, Ravap was the acaricide of choice. The test concentration of 0.36% was mixed by adding 48 ml of Ravap 28.7% EC to 3.8 l of water. The application rate was 3.84 l per 60 to 80 birds. Ravap was applied on days 1 and 29 (Table 62) to hens in rows 2 and 3 in both houses (Figure 43).

Morning and evening artificial lights supplemented natural light and provided hens with a total of 15 hr of light per day. Water and feed were offered *ad libitum*.

A fly control system, manufactured by Chem-amatic™ in Bellview, Florida, had been installed under the cages in house 200. When an automatic timer activated the system, a 0.033% pyrethrin solution was dispensed through spray heads located at fixed intervals throughout the house. The height of the spray heads was just above the level of the walks.

Table 62. Data collection and treatment application schedule for the Ravap^{TM} northern fowl mite trial in Chipley, Fl.

3 days prior to pre-treatment mite count	Began collection of egg production data on a daily basis.
Day 1	Mite pretreatment count completed; Ravap was applied.
Day 29	Second Ravap application.
Day 36	Post-treatment mite count completed.
Day 45	Terminated collection of daily egg production data.

Results. With the exceptions of strains 8 and 9, Ravap reduced mite populations to zero with two applications, as shown in Table 63. Mite numbers were reduced significantly ($p < 0.0001$) in all cases.

Although there were no significant differences between mite population means of treatment groups prior to treatment, significant differences between strains did occur (Table 64). This was due to the wide variation in mite numbers found on strain replications in the four quarters of house 200. Table 65 shows a significant difference between post-treatment group mite means. Note that the mite populations in the untreated group had doubled since the pretreatment count. Also shown is the ranking of mite means by strain. This is essentially a ranking of means from the untreated groups, since the mite populations in the treated groups had been virtually eliminated. Means shown in Table 65 were reduced by a factor of 2 since both treatments were considered in their computation.

In Table 66, pre- and post-treatment mite counts in the control group were added together and the means computed. This table gives some indication of strain resistance to mite populations. The strains, as ranked here, either maintained high mite populations throughout the trial or had rapid increases in population numbers during the trial.

Upon our arrival in Chipley, Florida, to begin the pretreatment mite count, it was noted that the pyrethrin fly control system previously mentioned was in partial operation on the north side of house 200. Since one group from each treatment was on the north side of the house, the house was divided into east and west halves and both halves were analyzed separately. Tables 67 and 68 show the pretreatment mite counts by treatment and strain for the west and east ends respectively of house

Table 63. Pre- and post-treatment mite population means by treatment and strain from caged-layer trial at Chipley, Fl.

Strain	Pretreatment				Post-treatment			
	Trt 1[a]	1-X[b]	Trt 2	2-X	Trt 1	1-X	Trt 2	2-X
1	10.4	84.6	1541.7	12534.0	0.0	0.0	742.0	6032.5
2	37.7	306.5	1954.2	15887.6	0.0	0.0	1731.8	14079.5
3	66.0	536.6	1279.7	10404.0	0.0	0.0	1891.7	15379.5
4	202.2	1643.9	414.9	3373.1	0.0	0.0	283.7	2306.5
5	500.0	4065.0	729.4	5930.0	0.0	0.0	728.1	5919.5
6	3714.6	30199.7	278.7	2265.8	0.0	0.0	645.2	5245.5
7	4151.0	33747.6	314.9	2560.1	0.0	0.0	1289.9	10486.9
8	2530.3	20571.3	562.5	4573.1	55.5	451.2	1063.5	8646.3
9	1447.9	11771.4	170.8	1388.6	0.5	4.1	4183.4	34011.0
10	2234.0	18162.4	59.6	489.5	0.0	0.0	3464.4	28165.6
11	839.6	6825.9	140.2	1139.8	0.0	0.0	1790.8	14559.2
12	351.1	2854.4	3666.7	29810.3	0.0	0.0	5896.9	47941.8

[a] Treatment 1 = Ravap
Treatment 2 = Control

[b] Mite population means converted to actual mean mite number by process described in text (actual mean = mite population mean X 8.13).

Table 64. Pretreatment mite population means by strain (treatment ignored) and by treatment group from the caged-layer trial at Chipley, Fl.

GROUPING			MEAN[a]	N	STRAIN
	A		2253.157895	95	7
B	A		2026.315789	95	12
B	A		2014.736842	95	6
B	A	C	1546.385417	96	8
B	A	C	1146.808511	94	10
B		C	945.937500	96	2
		C	809.375000	96	9
		C	776.041667	96	1
		C	673.210526	95	3
		C	614.583333	96	5
		C	493.684211	95	11
		C	309.677419	93	4

GROUPING	MEAN	N	TRT[b]
A	1346.782837	571	1
A	932.092820	571	2

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap™, Treatment 2 = Control.

Table 65. Post-treatment mite population means by strain (treatment ignored) and by treatment group from the caged-layer trial at Chipley, Fl.

GROUPING		MEAN[a]	N	STRAIN
	A	2979.473684	95	12
	B	2113.957395	95	9
	B	1732.180351	94	10
	C	945.533333	96	3
	C	876.329787	94	11
	C	865.335417	96	2
D	C	633.157895	95	7
D	C	564.321053	95	8
D	C	370.989533	95	1
D	C	304.052500	[illegible]	6
D	C	319.210526	95	5
D		144.945652	92	4

GROUPING	MEAN	N	TRT[b]
A	1980.[illegible]34211	570	2
B	4.627417	569	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Table 66. Means of the combined pre- and post-treatment mite counts of the control group (treatment 2) from the caged-layer trial at Chipley, Fl.

GROUPING				MEAN[a]	N	STRAIN
	A			5563.541667	48	12
	B			4354.187500	48	9
C	B			3635.937500	48	2
C	B			3523.936170	47	10
C	B	D		3171.354167	48	3
C		D	E	2283.645833	48	1
		D	E	1934.239130	46	11
			E	1626.041667	48	8
			E	1604.787234	47	7
			E	1457.291667	48	5
			E	923.936170	47	6
			E	698.617021	47	4

[a] Means with the same letter are not significantly different ($p < .05$).

Table 67. Transformed pretreatment mite population means by strain (treatment ignored) and by treatment group for the west end of house 200.

GROUPING			MEAN[a]	N	STRAIN
	A		43.924778	47	6
	B		30.570533	45	7
C	B		29.479933	47	10
C	B	D	22.041345	46	9
C	B	D	21.810354	48	8
C	B	D	20.610200	47	12
C	E	D	17.917438	45	3
F	E	D	13.297715	47	2
F	E		9.820219	47	11
F	E		9.244700	45	1
F			5.622646	46	4
F			3.283420	48	5

GROUPING	MEAN	N	TRT[b]
A	21.911304	286	1
B	15.378395	284	2

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Note: Data was transformed using $\sqrt{N+1}$.

Table 68. Transformed pretreatment mite population means by strain (treatment ignored) and by treatment group for the east end of house 200.

GROUPING			MEAN[a]	N	STRAIN
	A		34.613145	[illegible]	8
	A		32.284779	[illegible]	12
B	A		27.840593	47	7
B	A	C	26.471543	[illegible]	5
B	A	C	25.072731	48	2
B		C	21.129223	46	1
B		C	19.379564	48	6
B		C	18.794734	47	10
B		C	15.686355	[illegible]	11
B		C	15.367762	[illegible]	9
B		C	14.778031	47	4
		C	12.948361	47	3

GROUPING	MEAN	N	TRT[b]
A	22.322796	2[illegible]5	1
A	21.703110	237	2

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Note: Data was transformed using $\sqrt{N+1}$.

200. The mite populations in the control group in the west end of the house were significantly lower than those of the treated group. This is reflected in the differences in the mean rankings by strain between the east and west ends of the house (Tables 67 and 68). The northwest section, where the pyrethrin spray system was partially operating, was where the control birds were located. Tables 69 and 70 show the post-treatment mite counts by treatment and strain for the west and east ends respectively of house 200. Since the mite means for the control groups in both ends of the house were almost numerically equal it was discounted that the pyrethrin spray system had any effect on the mite populations.

Egg production means by treatment and strain for house 100, house 200, and houses 100 and 200 combined, are shown in Tables 71, 72, and 73 respectively. There were no significant differences in egg production due to treatment when strain differences were ignored. Mean rankings by strain were very similar in all three tables.

Table 74 gives the egg production means by week, with and without treatment being taken into consideration. Weeks 6 and 7 were significantly different from each other and from weeks 1 through 5, but there were never any significant differences between treatments at any point. There were no differences in egg production due to feed protein level and results are not reported.

A plot of weekly egg production means by treatment is shown in Figure 44. A nonsignificant drop (ca. 1%) in egg production occurred 3 days after both applications of Ravap (Table 74, Figure 44), possibly due to treatment stress. In each case, birds recovered and within 7 days began to lay at a numerically higher rate than the control birds.

Table 69. Transformed post-treatment mite population means by strain (treatment ignored) and by treatment group for the west end of house 200.

GROUPING	MEAN[a]	N	STRAIN
A	34.749415	47	9
A	33.131943	47	12
A	23.581391	47	10
B	19.262126	47	11
B	15.659921	46	3
B	15.595522	48	8
B	14.845799	48	7
C B	12.316359	48	1
C B	10.627505	43	2
C B	10.530330	47	6
C B	10.231445	48	5
C	5.307104	45	4

GROUPING	MEAN	N	TRT[b]
A	33.912224	234	2
B	1.397101	234	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Note: Data was transformed using $\sqrt{N+1}$.

Table 70. Transformed post-treatment mite population means by strain (treatment ignored) and by treatment group for the east end of house 200.

GROUPING				MEAN[a]	N	STRAIN
	A			37.058741	48	12
	B			24.169411	48	9
	B			24.138737	48	2
	B			22.041772	48	3
C	B			20.962822	47	10
C	B	D		16.352755	47	11
C	E	D		14.297567	47	8
C	E	D		13.990758	47	7
	E	D		11.342103	48	6
	E	D		11.191015	48	5
	E			8.397534	48	1
	E			7.953779	47	4

GROUPING	MEAN	N	TRT[b]
A	34.350701	236	2
B	1.000000	235	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Note: Data was transformed using $\sqrt{N+1}$.

Table 71. Egg production means by strain (treatment ignored) and by treatment group for house 100.

GROUPING		MEAN[a]	N	STRAIN
	A	84.201454	392	10
	A	83.422092	392	6
	B	81.148036	392	7
C	B	80.832832	392	4
C	B	80.477321	392	3
C	D	78.974031	392	2
E	D	77.938189	392	5
E	F	76.110230	392	9
G	F	74.648240	392	12
G		73.585403	392	11
	H	71.599872	392	1
	I	68.815704	392	8

GROUPING	MEAN	N	TRT[b]
A	77.991862	2352	2
A	77.308873	2352	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Table 72. Egg production means by strain (treatment ignored) and by treatment group for house 200.

GROUPING		MEAN[a]	N	STRAIN
	A	83.333622	392	10
	A	83.159617	392	6
B	A	81.579209	392	3
B	C	80.625255	392	7
	C	78.937500	392	2
	D	76.856964	392	9
E	D	75.409566	392	5
E	D	75.215332	392	4
E		74.459052	392	12
E		74.067117	392	11
	F	71.422602	392	1
	G	68.428418	392	8

GROUPING	MEAN	N	TRT[b]
A	77.062993	2352	1
A	76.867721	2352	2

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Table 73. Egg production means by strain (treatment ignored) and by treatment group for houses 100 and 200 combined.

GROUPING	MEAN[a]	N	STRAIN
A	83.767538	734	10
A			
A	83.200855	734	6
B	81.028265	734	3
B			
B	80.286045	734	7
C	78.930705	734	2
C			
C	78.049082	734	4
D	76.673873	734	5
D			
D	76.403597	734	[illegible]
E	74.552771	733	12
E			
E	73.526263	734	11
F	71.511237	734	1
G	68.622564	734	8

GROUPING	MEAN	N	TRT[b]
A	77.420472	4703	2
A			
A	77.185933	4704	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap[TM], Treatment 2 = Control.

Table 74. Egg production means by week (treatment ignored) and by treatment group in the caged-layer trial at Chipley, Fl.

GROUPING	MEAN[a]	N	WEEK
A	79.770491	1344	2
A			
A	79.603653	1344	5
A			
A	79.533661	1344	3
A			
A	79.512312	1344	4
A			
A	79.436979	1344	1
B	77.514859	1344	6
C	65.017498	1343	7

WEEKLY PRODUCTION MEANS BY TREATMENT

OBS	WEEK	TRT[b]	MEAN
1	1	1	79.4365
2	1	2	79.5514
3	2	1	79.7446
4	2	2	79.7694
5	3	1	79.3134
6	3	2	79.7893
7	4	1	79.4197
8	4	2	79.5909
9	5	1	78.9119
10	5	2	79.2123
11	6	1	77.8480
12	6	2	77.1590
13	7	1	65.9020
14	7	2	64.3917

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 = Ravap™, Treatment 2 = Control.

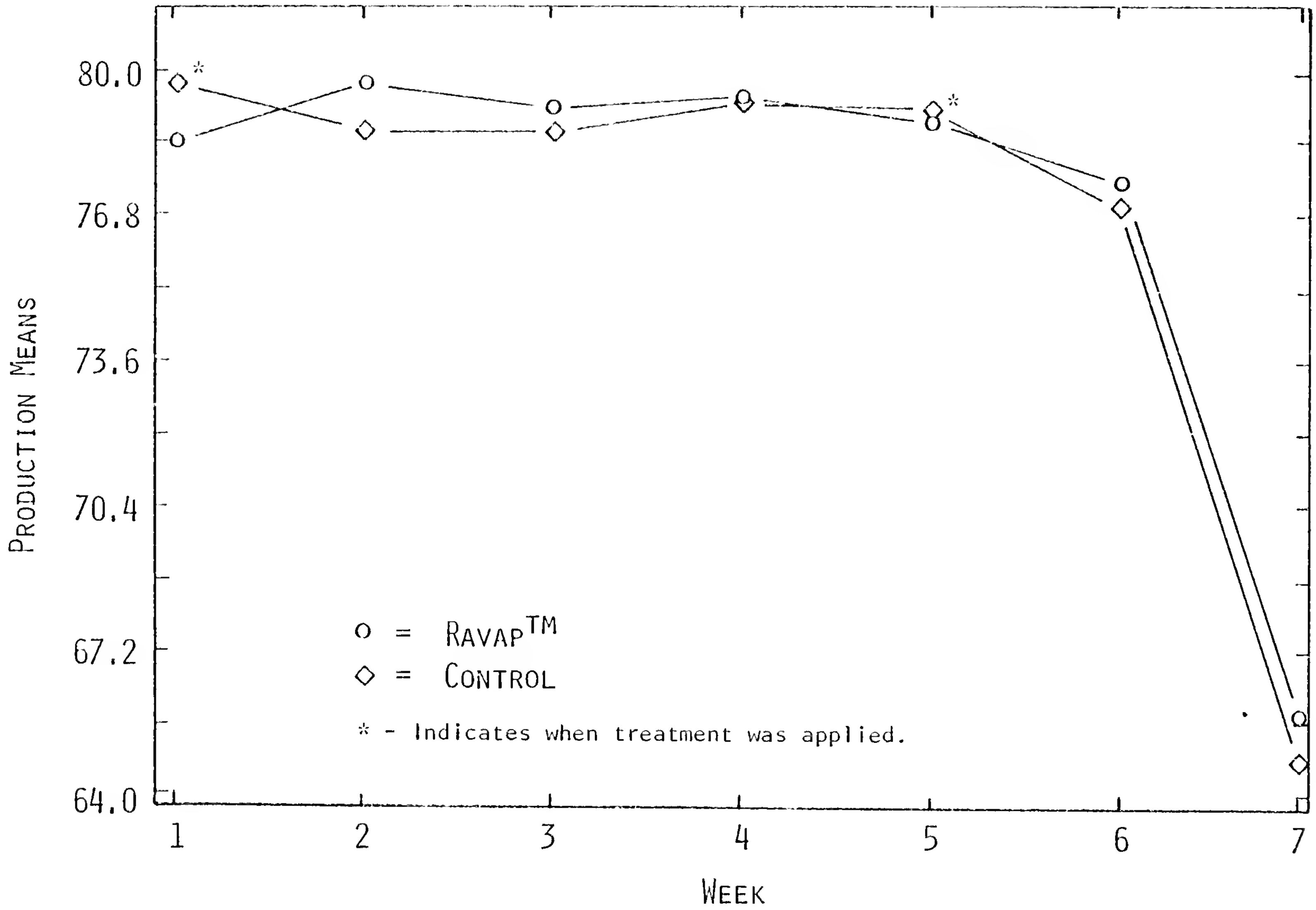


Figure 44. Weekly egg production means by treatment from the caged-layer trial at Chipley, Fl.

The decrease in egg production beginning in week 5 was attributed to the beginning of an outbreak of fatty liver syndrome (R. H. Harms, *personal communication*). Patent symptoms did not appear until several weeks after the experiment ended.

Analysis of individual strains by a t-test revealed a significant difference in production in one strain due to treatment (Table 75). The treated birds in strain 4 were producing eggs at a level slightly above the average production level of the 12 strains combined. The production level of the treated strain 4 hens represented a 3.67% increase in production ($p < 0.0137$) over the untreated strain 4 hens. As can be seen in Table 77, and other tables previously mentioned, strain 4 hens maintained low mite populations throughout the trial. Strain 4 was also one of the few strains to exhibit a natural decrease in mite population at the end of the trial (Table 77).

In an effort to correlate production data with effects caused by the previously mentioned pyrethrin sprayer, each quarter of house 200 was analyzed as a different treatment, i.e. treatments 1 through 4. Treatment means (treatments 1 through 4) shown in Table 76 were not significantly different and again, any effects of the pyrethrin sprayer were discounted.

In an attempt to show a correlation between mite numbers and egg production, a plot of egg production means versus mite means by strain was made (Figure 45). According to this graph, there appears to be no correlation. Birds with mite populations of ca. 640 or ca. 2320 mites per bird were both laying at the 81 percentile.

However, it appears that it is not mite numbers alone that affect production, but a combination of mite numbers and time (Table 77).

Table 75. T-test of egg production treatment means by strain from the caged-layer trial at Chipley, Fl.

Strain	Treatment[a]	Mean (%)	Δ[b]	Prob < T (%)
1	1	70.46	-1.92	82.53
	2	72.38		
2	1	78.32	-1.34	63.43
	2	79.66		
3	1	81.80	+0.44	24.09
	2	81.36		
4	1	77.05	+3.67	98.63
	2	73.38		
5	1	76.42	+2.03	84.34
	2	74.39		
6	1	83.03	-0.26	14.05
	2	83.29		
7	1	80.47	-0.31	17.01
	2	80.78		
8	1	69.15	+1.44	70.15
	2	67.71		
9	1	76.84	-0.09	4.79
	2	76.93		
10	1	83.52	+0.37	20.62
	2	83.15		
11	1	73.20	-1.73	76.80
	2	74.93		
12	1	74.48	+0.02	1.30
	2	74.46		
		$\bar{x}$ = 76.97		

[a] Treatment 1 = Ravap™, Treatment 2 = Control.

[b] Mean difference between treated and untreated birds.

Table 76. Egg production means when each quarter of house 200 was analyzed as a separate treatment.

GROUPING	MEAN[a]	N	TRT[b]
A	77.660802	1180	3
A			
A	75.391932	1170	4
A			
A	76.[illegible]31514	1175	2
A			
A	75.432477	1191	1

[a] Means with the same letter are not significantly different ($p < .05$).

[b] Treatment 1 SW = Ravap[TM], Treatment 2 NW = Control + pyrethrin, Treatment 3 NE = Ravap[TM] + pyrethrin, Treatment 4 SE = Control.

Table 77. Egg production means, pre- and post-treatment mite population means, and the change in mite population numbers on untreated hens from the caged-layer trial at Chipley, Fl.

Strain	Pretreatment Mite Mean	Post-treatment Mite Mean	Change in Mite Population During Trial	Overall Egg Production Mean (%)
6	278.7	645.2	↑ 2.0X	83.29
10[b]	59.6	3464.4	↑ 50.0X	83.15
3[a,b]	1279.7	1891.7	↑ 1.5X	81.36
7	314.9	1289.9	↑ 4.0X	80.78
2[a,b]	1954.2	1731.8	↓ 0.9X	79.66
9[b]	170.8	4183.4	↑ 25.0X	76.93[c]
11[b]	140.2	1790.8	↑ 12.0X	74.93
12[a,b]	3666.7	5896.9	↑ 1.5X	74.46
5[a]	729.4	728.1	No Change	74.39
4	414.9	283.7	↓ 0.7X	73.38
1[a]	1541.7	742.0	↓ 0.5X	72.38
8[a]	562.5	1063.5	↑ 2.0X	67.71

[a] Six strains with the highest pretreatment mite counts.

[b] Six strains with the highest post-treatment mite counts.

[c] Mean egg production: 76.87%.

<u>Note:</u> Arrows (↑,↓) indicate an increase or decrease in egg production.

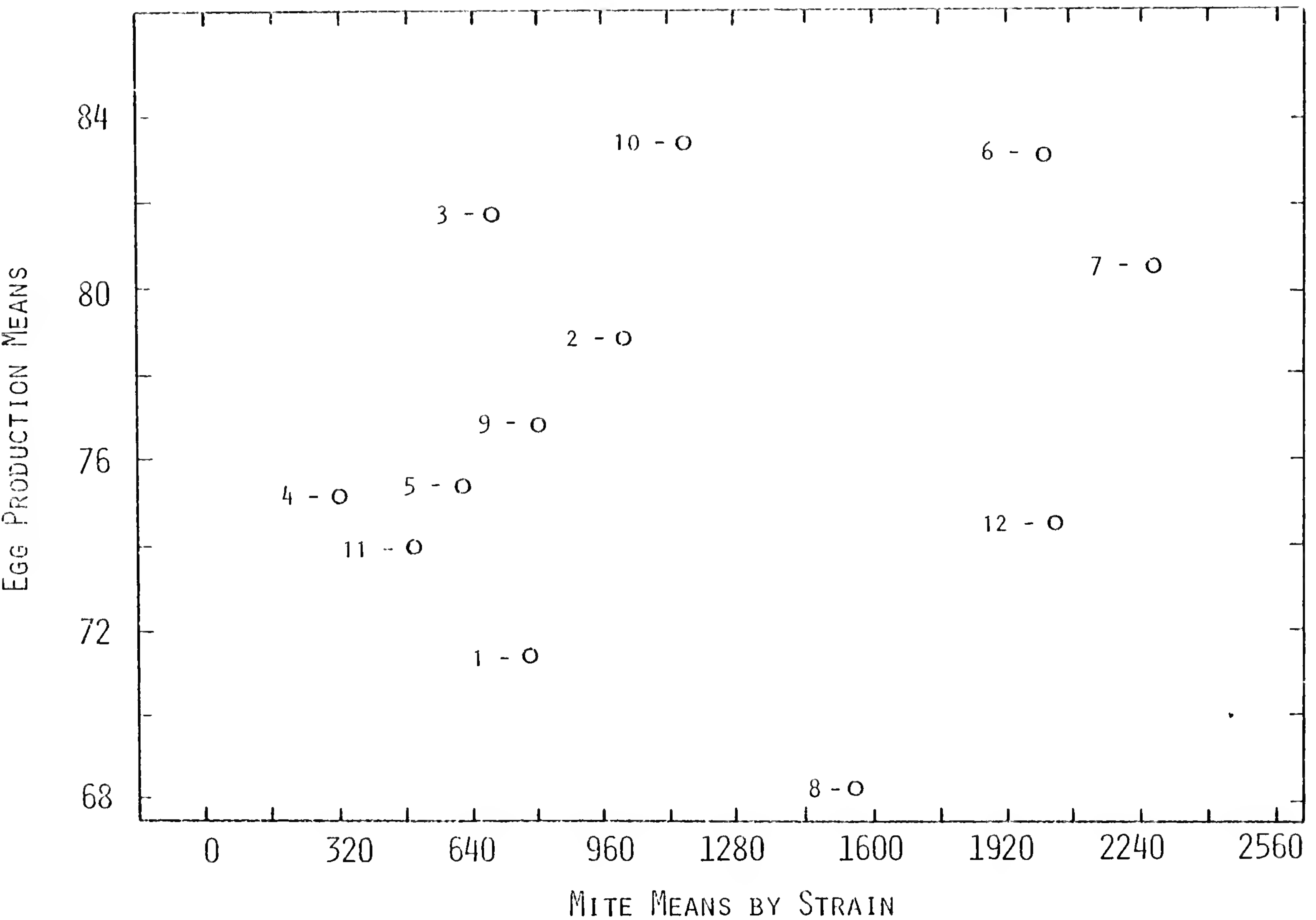


Figure 45. Plot of egg production means vs. precount mite means by strain from caged-layer trial at Chipley, Fl. (production means have not been adjusted to reflect strain differences).

Four out of the six strains with the highest pretreatment mite counts were laying below the average production level at the end of the trial. All six strains showed little or no change in mite populations during the trial. Of the remaining six strains, four of which were laying above the average, only one strain, strain 4, had a mite population change of less than +2X. Only two strains, strains 2 and 3, produced above the production mean and maintained mite populations ranking among the six highest in the pre- and post-treatment counts.

No matter what conclusions may be drawn from Table 77, it is interesting to note that the treated counterparts of the strains in Table 77, all with different pretreatment mite populations, had their populations reduced to zero or close to it. Yet, only one strain had a significant increase in production due to treatment. A longer trial including initial development of mite populations is necessary to more completely investigate the factors involved in this phenomenon.

DISCUSSION

The Value of Rotovation as a Method of Manure Management

After three years of experience dealing with tilling and manure management, I have made the following observations concerning rotovation as a manure management tool.

The Use of Tilling for Drying Manure

Manure may stay dry without tilling. Tilling manure does not necessarily guarantee that manure will be dry (McKeen and Rooney, 1976), and abstention from tilling does not necessarily guarantee that manure will be wet. During the 2 years that tilling experiments were conducted, manure at the tilling site was very wet. These normally unfavorable conditions were very suitable for our experimental purposes. After tilling experiments had been terminated, a new flock of chickens was housed and the manure the following year was the driest it had been in over 3 years.

The wet conditions during the first 2 years were attributed to two main factors. First, the summers were unseasonably hot. On one particular day, 50 chickens died as a result of the heat. Second, in an effort to cool themselves, chickens drank large amounts of water which caused droppings to be semiliquid throughout the spring, summer, and fall.

Rotovation as a Management Tool

Rotovation is not a substitute for good management practices. Instead, it is an additional management practice which can be used to dry

manure and reduce fly levels. Poultrymen who think that rotovation can be used as a shortcut to make up for lack of continuous sound management will soon discover that fly and manure problems will continue to exist even after the purchase of a tiller.

Tilling once or twice a month will not correct problems caused by leaking waterers or water seepage. In fact, tilling on an infrequent and random basis can be worse than not tilling at all. If manure is fairly dry and crusted over, periodic tilling will only expose wet areas beneath the crust and encourage fly breeding. This is particularly true during the months of July through September.

In the experiments performed in this study, most tilling was done in manure that was wetter than preferred. With sufficient time and effort, wet manure can be tilled enough to improve its consistency and make removal possible. This is not the purpose for which the tiller was designed.

If the decision is made to utilize rotovation as a manure drying technique, it must be implemented at the right time to ensure success.

Implementation of a Tilling Program

The time to begin a tilling program is in between flocks. As soon as a flock has been removed from the farm, work should begin to properly prepare the manure collection areas under the cages for a tilling program. When the old manure is cleaned out, a layer of manure should be left under the cages at a level that the tiller's tines can penetrate. This is also necessary to maintain a balance in biological control agents (Peck and Anderson, 1970). Tilling should begin immediately to dry this manure base as much as possible prior to the arrival of the new

flock. (Poultry farms are usually vacant 4 to 6 weeks between flocks and flocks are changed every 15 to 18 months.) When the new flock has arrived, the tilling program will have already begun and the manure drying process can begin on the first day.

If a poultryman waits until a flock has been housed before deciding to till, he has missed the opportunity to prepare dry beds in which to stir the manure. If he waits longer before deciding, he also has to contend with the layer of wet manure being deposited during every day he has waited. Extra time and effort will be necessary to bring manure moisture levels below those preferred by house flies.

In trials performed at the tilling site, tilling always began after a flock was housed. Manure was tilled a maximum of twice a day with a 30 min time interval between tillings. The moisture content of very wet manure was reduced to almost acceptable levels within 3 weeks. Tilling more than twice a day with longer time intervals should help shorten the drying period, but operation and labor costs are increased.

Tilling Frequency

Tilling frequency depends on the condition of the manure. Powdery dry manure is routinely tilled once a day by poultrymen in the Tampa area of Florida (C. F. Hinton, *personal communication*). Wet manure should be tilled two or more times daily depending on the individual situation. Except with semiliquid manure, progress can be seen within 1 week if manure is tilled frequently enough.

Tilling during rainy weather is not recommended especially if manure is not dry and dusty. Once manure is dry enough to be dusty, wetting it again is very difficult. On occasions when rain blew into

houses at the tilling site which contained manure in the state of dryness mentioned above, manure remained essentially unaffected. Tilling was resumed the following day if the weather cleared.

As wet manure loses its moisture content, it breaks into particles. The particle size will be quite large initially, but will continue to decrease as manure dries. When manure becomes powdery dry, particles essentially cease to exist and manure has a texture similar to coarse sand.

Particle formation hastens the drying process by increasing the manure surface area; it also allows parasitic wasps to more thoroughly search the manure for pupal hosts (Hinton, 1977). Particle formation provides the poultryman with visible evidence that the manure is drying.

<u>When to Till</u>

Tilling should be done during the heat of the day to increase the water evaporation from freshly turned manure. However, if manure is wet, tilling in the morning before egg collecting begins is beneficial as well as tilling several times during the afternoon. All tilling at the tilling site was done between 12 noon and 2 p.m. while the egg gatherers were at lunch. This allowed the entire farm to be tilled non-stop without interfering with the egg pickup.

<u>Leave a Dry Base When Poultry Houses are Cleaned Out</u>

A dry base that the tiller's tines can reach must be left each time manure is removed from poultry houses. Laborers cleaning out poultry houses at the tilling site always removed the dry manure base and some of the sand beneath it. More than one tilling experiment was halted because the tiller's tines could not touch the bottoms of the manure collection areas after the houses were cleaned out.

Two factors were responsible for this problem. First, the laborers prided themselves in doing a thorough job of removing all the visible manure plus a little more. Second, the poultryman refused to monitor the activities of the laborers and insist that manure be removed only to the level that he specified. As an observer, I could only make suggestions that often went unheeded.

Tilling Reduces Manure Volume and the Number of Clean-Outs Per Year

Poultrymen are currently cleaning out manure three times during the 15-month period that a flock is housed. However, regular use of a tiller has reduced clean-outs on some north Florida poultry farms to two per 15-month period. The present cost of cleaning out a poultry house is $3.28 per m, or $300 for a 91.4 m house. Reduction in the number of clean-outs per flock by one third would save the owner of the tilling site $1200 per year.

This savings is increased when one considers the value of the manure as a fertilizer (Hinton, 1977). In 1976, farms in the Tampa area were selling rotovated manure for $10 per ton and attempts were being made to increase the interest of the citrus industry in this readily available product.

Rotovated manure can be spread on fields or sold in bags for fertilizer without further drying. In this form, the manure is unattractive to flies and does not burn plants or pastures (C. F. Hinton, *personal communication*).

Tiller-Related Costs and Returns

Initial cost of purchasing a tiller is high but operational costs are low. Selpat sells the tiller and the modified Kuboda as a unit,

although the two can be purchased separately. The price in 1976 was approximately $3000. This capital can be regained in 3 to 5 years if a tilling program is begun and adhered to. Returns are in the forms of reduced manure clean-out schedules (described in the previous section), increased value of manure as a fertilizer (also described above) and the reduction of adult and immature fly control costs.

An average of 33.68 min were required to till all four houses at the tilling site. Average time required to till one 91.4-m poultry house was 8.42 min (Table 78). Time values in Table 78 also represent time spent moving equipment and egg carts out of the path of the tiller, so actual time spent tilling was slightly less than the time indicated. Some time was also spent keeping up public relations with the farm owners.

Approximately 0.25 l of fuel and 0.14 tractor hr were required to till one house (Table 78). All four houses at the tilling site could be tilled once a day for 16.55 days on one tank of fuel (19.2 l).

Using the figures given in Table 78 and assuming the cost of fuel and labor to be $0.26/l and $3 per hr respectively, it would cost the owner of the tilling site $640.58/year to till his four California-style houses once a day. This translates to $0.032/bird per year, which is a reasonable cost for fly control. This figure quickly increases, however, if houses must be tilled more than once a day. Hinton (1977) quotes tilling costs of $.03 to $.10 per bird as compared to $.06 to $.08 per bird for the purchase of pesticides. His figures are for a 50,000-bird flock.

Table 78. Mean operator and tractor time and the amount of fuel required to till one 91.4-m California-style poultry house.

Operator Time (min)[a]	
20.00	8.00
10.00	6.67
7.33	6.00
5.67	8.00
7.75	7.76
10.00	7.00
7.00	7.00
8.00	7.50
6.67	6.67
12.50	12.50
10.00	7.50
6.67	6.67
8.00	7.00
	10.00
Σ	227.27
per house	8.42

	Odometer Reading (hr)[b]	Fuel Utilized (l)[c]
Start	71.90	19.20(full)
Finish	80.50	0.96
Total	8.60[d]	18.24[d]
Per house	0.14	0.29

[a] Time required to physically operate the tiller.

[b] Amount of time the tractor motor operated according to the tractor odometer.

[c] Amount of fuel used while tractor was in operation (includes idling time).

[d] Represents the tractor operating time and fuel utilized to till one 91.4-m house 63 times, or the tilling site farm (four 91.4-m houses) 15.75 times.

Operational costs, i.e. parts and service, over the 2-year testing period were less than $50. One part had to be rewelded, the oil and filter were changed, and antifreeze was added to the radiator. Chicken manure caused rapid corrosion of metal parts, and at the end of a 2-year period, the shields covering the tiller's tines were practically rusted away and replacements were needed. These shields were very important since they prevented manure from being thrown up into the cages during tilling and staining the eggs.

<u>Tilling can be Incorporated in an Integrated Control Program</u>

Insect growth regulators such as CGA 72662 can be applied topically to manure as a spot treatment in hot spots, areas with thriving populations of fly larvae. When tilled into manure, CGA 72662 will prevent fly resurgence for up to 5 weeks. Similar tests were not attempted with methoprene because it decomposes so rapidly.

Wood chips can be added to hot spots or wet spots and tilled in to give manure consistency and aid in the drying process. When chips are added to semiliquid manure, flat mounds will begin to form if manure is tilled daily for 1 week. Once chips are added, manure can become attractive to house flies if moisture levels are right (Kilpatrick and Schoof, 1959). Tilling should continue in an attempt to reduce moisture levels below those preferred for fly breeding.

Addition of sand to semiliquid manure causes manure to thicken and become heavy. The extra weight causes manure to be difficult to handle and can cause damage to manure spreading equipment. Tilling semiliquid manure without the addition of a stabilizer is of no value and drying does not begin until some texture can be given to the manure.

Ophyra aenescens Larvae as Predators of *Musca domestica* Larvae

In the competition study, *M. domestica* viability was suppressed significantly by *O. aenescens* in all cases (Table 12). This was indicative of predation but not proof. When larvae of *O. aenescens* were reared in vermiculite, the daily addition of house fly larvae as a possible food source significantly reduced the mortality rate of *O. aenescens* in all cases (Table 14). Attempts to rear *O. aenescens* larvae in vermiculite failed when no house fly larvae were added. This confirmed that larvae of *O. aenescens* are not cannibalistic, but they are predators of house fly larvae. Predation by *O. aenescens* was also observed during the study.

Peck (1969) proved that *O. leucostoma*, a sister species also found in poultry manure, is predaceous upon house fly larvae using a vermiculite study similar to the one I used. Studies indicated that each larva of *O. aenescens* could destroy more than 20 house fly larvae during the 5-day larval feeding period. Larvae of *O. leucostoma* were found to destroy from 20 (Anderson and Poorbaugh, 1964b) to 30.6 (Peck, 1969) house fly larvae during their developmental period.

Anderson and Poorbaugh (1964b) stated that when house fly larvae were added to cups of vermiculite containing larvae of *O. leucostoma*, they were chased out of the vermiculite by the *O. leucostoma* larvae. *Ophyra leucostoma* larvae killed more house fly larvae than they could eat and dead larvae attracted more house fly larvae. This enabled *O. leucostoma* to increase the number of house fly kills (Anderson and Poorbaugh, 1964b).

Larvae of *O. aenescens* in vermiculite became very active when larvae of *M. domestica* were added, but did not chase *M. domestica* larvae as described by Anderson and Poorbaugh (1964b). The nature of house fly larvae to congregate around and feed upon the dead of their own kind was also observed in the *O. aenescens* experiments.

When larvae of *O. leucostoma* were reared with house fly larvae in nutrient medium, *O. leucostoma* killed as many house flies as when the two species were reared together in vermiculite. This indicated the preference of *O. leucostoma* for house flies over the constituents of the nutrient medium (Anderson and Poorbaugh, 1964b). This was not the case with *O. aenescens*, as indicated by the competition study.

Morphological Proof that *Ophyra aenescens* is Predaceous

Séguy (1923) stated that larvae of *Ophyra* are predaceous but gave no reasons to substantiate the claim. Morphological evidence was presented by Keilin and Tate (1930) who showed that larvae of *O. leucostoma* are predaceous and saprophagous.

The morphological features of the cephaloskeleton of *O. aenescens* are comparable to those of *O. leucostoma*. The accessory oral sclerites and the longitudinal ridges on the venter of the basal sclerite indicate that larvae of *O. aenescens* are predaceous and saprophagous. The cephaloskeleton of *M. domestica* lacks the accessory oral sclerites, but ridges are present on the venter of the basal sclerite. Larvae with these characteristics are saprophagous but not predaceous (Keilin and Tate, 1930).

The Value of *O. aenescens* as a Biocontrol Agent

In manure having the desired moisture range, *O. aenescens* may aid in house fly control if laboratory results are indicative of field results. At the tilling site, larvae of *Ophyra* sp. were only found singly or in pairs, and never in pockets or hot spots like *M. domestica*. This type of behavior would limit the value of *Ophyra* as a house fly predator.

If a manure management program is drying manure enough to provide good house fly control, this would also preclude the development of other diptera such as *Ophyra*, whose moisture requirements are similar to those of *M. domestica*.

Adults of *Ophyra* were never noticed at the tilling site except in light trap catches (Table 40). In large numbers, these flies can be especially bothersome. Adults are particularly bold and are not intimidated by the usual methods employed to chase flies from clothing or body surfaces (P. G. Koehler, *personal communication*).

Rearing *Ophyra aenescens* in the Laboratory

Ophyra aenescens was colonized and reared with the same relative ease as *M. domestica*. Life cycle from larva to adult averaged 14 days at 29.4 C and the average life span for males and females was 16.5 to 20 days respectively. These results were almost numerically the same as those of Johnson and Venard (1957).

When kept in the growth chamber at 29.4 C, larvae of *O. aenescens* raised the temperature of their growth medium to 42.4 C on day 2. The temperature dropped daily and reached 30.0 C on day 7. When Johnson and Venard (1957) found larvae of *O. aenescens* in the field, temperatures of larvae media ranged from 35.6 C to 41.1 C.

High larval densities should be maintained for maximum development of larvae. Less than 25 larvae per 300 ml of diet resulted in increased larval mortality. Fortification of the larval diet significantly increased larval viability.

The Influence of Larvae of *Hermetia illucens* on Other Species of Fly Larvae

Larval development of *M. domestica, O. aenescens,* and *S. robusta* was not completely suppressed when each species was reared individually in containers with larvae of *H. illucens*. Earlier attempts to demonstrate complete suppression of other fly species by *H. illucens* in the laboratory were inconclusive (Fletcher et al., 1956; Furman et al., 1959).

Soldier fly larvae tend to remain on the bottoms of the rearing containers and the C.S.M.A. in these areas becomes very slimy. The slime does not repel the larvae of *M. domestica*, *O. aenescens*, or *S. robusta* or otherwise keep them from entering these areas, but it may somehow inhibit their development.

The order in which the fly species were affected by *H. illucens* was *M. domestica* > *S. robusta* > *O. aenescens*. This was expected for *M. domestica* since other experiments indicated that *H. illucens* would not allow *M. domestica* to develop at all (Furman et al., 1959). These results were not expected for *S. robusta* with a larval development period lasting only 3 days. *S. robusta* larvae prefer wetter habitats than *M. domestica* and perhaps this brought them into closer contact with *H. illucens*. Larvae of *O. aenescens* were affected least of all by *H. illucens* larvae. Mortality rates for *H. illucens* larvae indicate that they were not being preyed upon by *O. aenescens*. *Ophyra* larvae may not be able to penetrate the soldier fly cuticle.

In the field, large populations of *H. illucens* may totally prevent other species of flies from breeding in their midst. Control of house flies on poultry farms with naturally occurring populations of *H. illucens* was noted by Tingle et al. (1975) and advocated by Vazquez-Gonzalez et al. (1962). At the tilling site, populations of house flies and soldier flies were found living at different depts in the same manure pack. The depths were determined by moisture level. This is an example of species packing in which two or more species utilize the same resource without interference (Price, 1975). This unusual situation was created when wood chips were added to wet manure.

Soldier fly larvae are not popular with poultrymen, especially after they liquidate manure and then cover the walks when migrating to dry areas for pupation. This fly is not recommended as a biocontrol agent in this country. Good manure management techniques should prevent serious outbreaks of *H. illucens*.

The Efficacy of Dimilin as a Feed Additive

When fed to hens at 10 ppm, dimilin produced fly mortalities of 74.17% in the laboratory and 53.33% in the field. When fed at 1 ppm, laboratory mortalities were 17.50%. Miller et al. (1975) fed dimilin to hens at 6.2 to 12.5 ppm and achieved 100% control of flies.

The addition of dimilin to feed at 1 and 10 ppm caused hens to eat significantly greater amounts of feed and produce eggs at a level that was numerically but not significantly higher than control hens. Eggs produced by hens consuming dimilin in the diet have been found to contain residues (Miller et al., 1975). This, along with the fluorine and chlorine atoms attached to the dimilin molecule (Kenaga and End, 1974), has helped to prevent dimilin from becoming labeled for commercial use.

The Efficacy of Methoprene as a Feed Additive

When fed to hens at 10 ppm, methoprene produced fly mortalities of 85.00% in the laboratory and 55.10% in the field. When fed at 1 ppm, methoprene produced a laboratory mortality of 17.50%. Morgan et al. (1975) produced 70.9% fly mortality with 5 ppm methoprene diets, but a mortality of 99.3% required 100 ppm methoprene diets. Low mortalities produced under field conditions were attributed to the rapid decomposition of methoprene (Schaefer and Dupras, 1973). This rapid decomposition eliminates methoprene for use as an oral larvicide. Larval molts are not affected by low concentrations of methoprene and the critical pupal molt occurs after the larvae have left the area of methoprene concentration.

Consumption of methoprene caused no significant differences in feed consumption or egg production except in one case where hens were allowed to run out of feed. Morgan et al. (1975) found no differences in hen weights due to methoprene diets.

Methoprene as a Topical Larvicide

Methoprene applied topically to manure at 1076 mg/m^2 produced mortalities of 90.0 and 99.0% in laboratory-strain flies 3 days post-treatment. Applied at 538 mg/m^2, 3-day post-treatment mortalities of 73.0 and 96.0% were produced, but this result may not be indicative of what is occurring in the field.

With a residual of 3 days or longer, methoprene applied topically would be ideal for spot treatment of house flies in poultry manure. The granules are in a form that is convenient to use and could be dispensed from the rear of a rototiller. Tilling would increase the chances of larvae coming into contact with methoprene.

Parasitism rates of pupal parasites have been shown to double in manure treated with methoprene (Butler and Greer, 1974), due to the action of methoprene that prevents pupae from emerging. Used for spot treating, methoprene could be applied at rates high enough to affect larval molts as well as the pupal molt. Other investigators found methoprene to be ineffective as a topical larvicide in poultry manure (Morgan et al., 1975).

Laboratory Studies with CGA 72662

At high concentrations, CGA 72662 kills the early instars of fly larvae. Only *H. illucens* and *F. canicularis* formed pupae at the initial levels of CGA 72662 tested, and none of these pupae emerged (Table 27).

An LC_{50} of 0.45 ppm was found for house fly larvae. Although this is well below 1 ppm, the slope of the dosage-mortality curve indicates that the susceptibility of the house fly population tested to CGA 72662 is relatively heterogeneous (Hoskins and Gordon, 1956).

CGA 72662 concentrations of 0.25 ppm produced larviform pupae that eclosed like normal pupae. As concentrations continued to increase, numbers of larviform pupae increased, and the number of uneclosed larviform pupae increased (Table 28). At 1.0 ppm five uneclosed larviform pupae formed in the four treatment replications and no other larvae or pupae were found. This indicates that at concentrations of 1.0 ppm and above, larvae were being killed prior to reaching the pupal stage.

CGA 72662, Dimethoate, Dichlorvos, and Ravap as Topically Applied Larvicides

Effective larval control lasted for 7 days with Ravap and dimethoate, and from 7 to 11 days with dichlorvos. Seven days of control

is all a poultryman can expect to get from compounds that are presently available (J. F. Butler, *personal communication*).

The lowest level of CGA 72662 (0.05%) began to break down in 14 days, but the other levels of CGA 72662 were still active 35 days post-treatment. CGA 72662 (0.1%) remained active for 35 days even when tilled into wet manure. Potential resistance problems would result if CGA 72662 concentrations were not periodically increased to ensure the production of 100% larval mortalities.

In a liquid form, CGA 72662 may not be as convenient to use as methoprene granules. However, when used for spot treatment of house flies in manure, the residual of CGA 72662 would prevent fly activity for long periods and allow treated areas to dry out. Retreatment would not be as frequent as with methoprene. CGA 72662, like dimilin, kills fly larvae in all stages and prevents rapid fly resurgence. Ciba-Geigy has done residual testing but the results are unknown at this time.

The Efficacy of CGA 72662 in Water

CGA 72662 added to drinking water of hens at rates of 1.5 to 20.0 ppm produced larval mortality rates in the laboratory house fly strain of 80.89 to 100.00% respectively, and was still effective 3 days post-treatment. Ciba-Geigy representatives claim that fly control ceases the day that use of treated water ceases (D. B. Hays, *personal communication*).

Although the use of a larvicide in drinking water may be considered impractical by members of the poultry industry (R. H. Harms, *personal communication*), Ciba-Geigy feels that all routes of application should be open to the poultryman (D. B. Hays, *personal communication*).

Light Traps for Surveying Adult Fly Populations

Each of two light traps at the tilling site caught different numbers of the monitored fly species from month to month and presented two different pictures of monthly changes in fly population numbers. Because of such variation in catches, light traps are recommended for survey work but are not considered consistent enough to accurately estimate fly populations (Pickens et al., 1972).

Driggers (1971) and Pickens et al. (1975) increased fly catches by lowering traps from ceiling level to ground level. At the tilling site, traps were hung at ceiling level because ground level traps would have interfered with the rototiller and would have been covered with manure as a result of the rototiller.

The yellow trap consistently caught more flies than the black trap. This was due to the differences in wave lengths emitted by the light sources of the two traps. Although the traps themselves were two different colors, it is doubtful that trap color influenced the fly catch (Mitchell et al., 1975).

Although their influence on fly populations may be questionable, light traps are looked upon favorably by some poultrymen. Light traps provide results that the poultryman can see and hear all day long.

Granular Baits for House Fly Control

A combination of Kill'em Fly Killer II (Bomyl) plus Lure'em II attractant produced the best results in the field although laboratory testing indicated Kill'em Fly Killer II had a relatively long knockdown period of ca. 4 hr. Kill'em Fly Killer (Vapona + ronnel) had the fastest knockdown, but the lowest fly catches of all baits tested with

the exception of Atroban. Atroban was unattractive to flies in the field even though laboratory tests indicated it had fly-killing ability comparable to that of Golden Malrin with Muscamone.

Atroban and Golden Malrin with Muscamone were producing mortality rates of 87.5 and 100.0% respectively after 40 days of laboratory residual testing. Baits field-tested by Bailey et al. (1970) provided 75% control for half as long. Baits for use on poultry farms do not necessarily need long residuals. Poultrymen routinely sprinkle baits along walkways in poultry houses. In the late afternoon while feed is being distributed to the chickens, walks are swept by a mechanically operated broom mounted beneath the feed cart.

Wilson and Mulla (1975) observed that bait stations near the perimeters of poultry houses caught flies in nearly a one-to-one ratio and that bait stations dominated by one sex had catches significantly lower than those stations conducive to both sexes. Results from field testing conducted at the tiling site were not always in agreement with the above observations. The bait catching the most flies attracted significantly greater numbers of females than males.

The addition of Lure'em II attractant to baits resulted in fly catches significantly higher than those produced by Golden Malrin with Muscamone. This was not unexpected since other attractants have been formulated which produce larger catches of flies than Muscamone (Mulla et al., 1977).

At the tilling site, flies killed by Muscamone baits were in a 2:1 female to male ratio instead of the 1:1 ratio reported by Carlson and Beroza (1973). When Lure'em II attractant was added to baits, flies killed were also in a 2:1 female to male ratio.

Granular baits could be used in conjunction with a tilling program to reduce the numbers in existing fly populations and prevent an increase in fly numbers due to migrating adults. Attractiveness of baits could conceivably increase as manure is dried by tilling and becomes less attractive to flies.

Sugar baits are routinely used by poultrymen to kill adult house flies. They are easy to use and results are visible in a short period of time.

Efficacy of Synthetic Pyrethroids as Contact Residuals

Although the house fly LD_{50} for permethrin (JFU 5819) is above the acceptable level of 1 ppm, the slope of the dosage-mortality curve indicates that resistance may be building within the population (Hoskins and Gordon, 1956). Such resistance has already been reported in mosquitoes (Priester and Georghiou, 1978). The regression equation presented in the results section was used to arrive at a tentative LD_{90} of 45 ppm. This is below the concentration rates of 500 ppm and 50 ppm tested on wood, tin, and cement block templates in the laboratory.

Permethrin (JFU 5021A) decomposed in less than 24 and 72 hr on cement and tin surfaces respectively, but persisted up to 23 months on wood. This was much longer than could be expected for organophosphorous compounds (Hansens and Bartley, 1953).

The wooden rafters in poultry houses were logical field application sites because they are favorite overnight resting places for house flies (Anderson and Poorbaugh, 1964a). Since synthetic pyrethroids were not labeled for use in or around poultry houses at the time of the experiment, selected compounds were sprayed on plywood panels which were hung in poultry houses at the tilling site after the panels had dried.

After flies had been exposed to the panels for ca. 15 min, they exhibited signs of incoordination and other characteristic symptomology described by Wouters and van den Bercken (1978). Had they not been caged, the spasmodic wing fluttering prior to death could have carried flies far from the panels' surfaces and thus given the impression that the panels were not efficacious. This may have been what was actually occurring in the field since dead flies were never seen near the panels.

The synthetic pyrethroid compounds probably persisted at toxic levels for more than 121 days in the field. Breakdown was attributed to the disinfectants used to clean the poultry houses during between-flock sanitation procedures. Once approved for use, synthetic pyrethroids applied to rafters prior to the arrival of a new flock of chickens should provide adequate fly control for as long as the flock is housed.

Long term contact residuals like the synthetic pyrethroids would be a good addition to any fly control program. If applied between flocks as suggested above, hens, eggs, feed, and water would not be contaminated. The pyrethroids could be applied very thoroughly since the poultryman would not be concerned with other activities such as daily flock management. Fly resistance would be of foremost concern. Shono et al. (1978) stated that ease of permethrin detoxification by insects may limit its use.

<u>Susceptibility of Endemic Florida Strains of Norther Fowl Mites to Carbaryl, Malathion, Ravap, and Synthetic Pyrethroids</u>

Laboratory testing showed that the LC_{50}'s for SD 43775, SBP-1382, BW 21Z, and Ectiban were higher than those of carbaryl and malathion. Hall et al. (1978) had similar results except their LC_{50}'s for carbaryl and permethrin, and malathion were greater than mine by approximate

factors of 10 and 86 respectively. The mite strains tested by Hall et al. (1978) were more resistant than the Florida strains, especially to malathion.

Slopes for all compounds tested were less than one (Table 55). Carbaryl and malathion had the flattest slopes indicating that concentrations necessary to achieve 100% kill in the field would have to be much higher than concentrations of the permethrins required to elicit the same rate of response. The slopes of compounds tested by Hall et al. (1978) were slightly steeper indicating that the susceptibilities of their mite strains to the acaricides were more homogeneous than the Florida strains (Hoskins and Gordon, 1956). In summary, Florida strains of mites are not as resistant as the strains tested by Hall et al. (1978), but the shallow slopes of the regression equations indicate that resistance may be building within the Florida strains.

In field tests, Ravap gave practically 100% control of mites in 2 weeks. Carbaryl and malathion produced the same results in 4 and 6 weeks respectively. Control with Ravap was probably due in part to the vaporizing action of Vapona. Loomis et al. (1970) got poor mite control on hens with a 0.5% carbaryl solution, but Hall et al. (1978) got 100% control within 24 hr using the same concentration. Several reports of suspected malathion resistance (Foulk and Matthysse, 1963; Rodriguez and Riehl, 1963) preceded the confirmation of resistance by Hall et al. (1978). Many resistance claims are a result of poor application methods (Eleazer, 1978) for which the poultry industry is notorious. This was not the case with the experiments at the tilling site, since hens were sprayed more thoroughly than could be expected under normal conditions.

Mite control of 100% was achieved for 36 days with SD 43775 and BW 21Z on floor-housed hens. Hall et al. (1978) had similar results for 57 days with much lower concentrations of SD 43775. The reason for the extended mite control by synthetic pyrehtrins may be due to their ability to persist on the chickens' feathers (Hall et al., 1978).

Since the floor litter was not sprayed, the results of the synthetic pyrethroid test were particularly interesting. Mites can exist off the host for 2 to 4 weeks (Cameron, 1938; Baker et al., 1956; Kirkwood, 1963; Loomis, 1978), and the litter at Chipley, which was compacted and moderately damp beneath the surface, provided conditions necessary for mite survival. After short-term acaricides have broken down, mites hiding in the litter are available to reinfest the birds, but synthetic pyrethroids persist long enough to break this cycle.

The Effects of Northern Fowl Mites on Egg Production

When 12 strains of laying hens were treated for control of northern fowl mites, significant differences in production could not be detected when combined production means were compared by treatment. Although investigators have claimed that decreases in egg production were due to northern fowl mites (Cameron, 1938; Metcalf et al., 1962), recent studies have not been able to corroborate these claims (Loomis et al., 1970; Eleazer, 1978; DeVaney, 1979). One author suggested that mites be controlled solely to prevent worker discomfort (Bramhall, 1972).

Comparison of production means of each strain individually revealed a significant increase in production in one strain due to mite removal. Five other strains had nonsignificant improvements in production which compared favorably to the results of Combs et al. (1976).

Egg production levels could not be correlated with mite infestation levels (Figure 45). Perhaps hen performance during the experiment was not related to mite infestation levels that existed at the time of the experiment, but to mite infestation levels that existed at some time prior to the experimental period. Hens infested with mites by DeVaney (1978) at different points in the laying cycle produced eggs at significantly lower levels 6 to 8 weeks following the onset of infestation. Hen performance during the experiment at Chipley may also have been due to the effect of mite infestation levels over time.

During the experiment, mite populations on untreated hens fluctuated as they must have done prior to and following the end of the experiment. Therefore, inferences made about egg production and mite infestation rates may not be entirely correct since the entire picture is not known. Studies providing for long-term monitoring of egg production and mite infestation rates should provide the data necessary to determine whether or not a correlation does exist between the two.

Whether it is statistically significant or not, a 1% decrease in egg production can be costly to a poultry farm owner (R. H. Harms, *personal communication*). If a hen lays an average of 240 eggs per year with a current wholesale value of 5 cents per dozen, a 1% drop in production for just 1 day can cost the owner of a 35,000-bird flock $350. The first week after birds were treated in Chipley, the production level of the treated birds was 1% lower than the control birds. One week later, however, treated birds had increased production by 1.5% and were again laying at a numerically higher rate than the controls. After the second treatment, production in the treated birds dropped by 0.64%.

This time the production rate was not recovered and after 1 week, treated birds were laying at a rate which was less than 1% higher than the control birds. If spraying for mites results in a 1% drop in egg production for at least 1 week, but cannot guarantee a 1% increase over and above the original production level, northern fowl mites should be sprayed, as suggested by Bramhall (1972), only to prevent discomfort to the labor force.

Long-term compounds like the synthetic pyrethroids could help reduce the number of times per year that hens must be subjected to the pesticide application stress that occurs when miticides are applied. A well timed application of a long-term miticide when available, could break northern fowl mite reproductive cycles and prevent unnecessary retreatment of hens.

Evaluation of the Mite Rating Systems

The laboratory estimate of northern fowl mite populations was an attempt to correlate field-estimated mite populations with the actual numbers of mites on the hens. However, the data obtained by washing birds are not considered accurate because of the small sample sizes and the high variability within the samples. Since birds were only washed once to remove the mite populations, it is not known if all mites were removed. Also, just one aliquot from each sample was counted.

Since the laboratory estimate was designed to be used only as a guide, sampling methods that would give statistical confidence were not employed. The devised conversion factor of 8.13 was also used only as a guide. Statistically valid conversion factors would require enough sample numbers to plot a regression line correlating field estimates and mite numbers for actual mite populations.

No attempt to correlate field estimates of mite populations with actual mite numbers present on birds by use of laboratory counting procedures was found in the literature. The results of all northern fowl mite field research performed in the past are based on some type of mite rating system. Hence, the field estimates of northern fowl mite populations on hens (p. 70) were considered to be the better estimate of mite numbers and were the values used for statistical analyses of mite data presented in this dissertation.

CONCLUSIONS

Major conclusions formulated as a result of this research are as follows:

1. Rototilling was found to be a satisfactory method for drying wet manure although more time and effort are involved than when a tiller is used in a routine manure management program.

2. When a tiller is properly incorporated into a routine manure management program, benefits include fewer manure clean-outs per year, the increased value of tilled manure as a fertilizer, and reduced costs for fly control when compared to the use of pesticides.

3. A tiller is not necessary in order to have dry manure on a poultry farm. Likewise, owning a tiller will not compensate for overall poor management practices.

4. Liquid manure cannot be tilled to a drier state unless a stabilizer is added. Wood chips aid in the drying process by creating increased surface area and adding consistency. Builders' sand makes manure heavy and difficult to work with.

5. Larvae of *Ophyra aenescens* are predaceous on house fly larvae and can kill more than 20 first-instar larvae per day. Large populations of *Ophyra* adults are pestiferous and would be an unwanted nuisance on a poultry farm.

6. Larvae of *Hermetia illucens* and *Musca domestica* can coexist in larval media in the laboratory and in the field by developing at different depths in the media. Drying manure would eliminate both of these flies.

7. Methoprene and dimilin added to poultry feed are not effective as oral larvicides. CGA 72662 in drinking water of hens is an effective oral larvicide with activity continuing 3 days after cessation of treatment.

8. Methoprene is effective for up to 3 days as a topically applied larvicide, and would be ideal for use in wet manure since it is applied in a granular form.

9. CGA 72662 is effective up to 35 days when applied as a topical larvicide. Fly resurgence can occur within 2 weeks with the use of commercially labeled organophosphorus larvicides. CGA 72662 persists long enough to give manure a chance to dry out with no immediate chance of additional manure liquification due to fly activity.

10. Granular baits may be effective for reducing adult house fly populations, but probably not as effective as contact residuals. Baits kill only the flies attracted to them. A contact residual applied to the rafters of a poultry house would kill all flies coming in to rest for the night.

11. Northern fowl mites can be controlled with currently labeled compounds available to poultrymen. Resistance is developing to some compounds in some areas of the country, but many resistance problems are due to faulty application methods. Much research is needed in the area of acaricide application to poultry.

12. Synthetic pyrethroid compounds are effective acaricides with long residuals. Compounds such as these are necessary to break the mite life cycle especially when dealing with floor-housed chickens.

13. Although northern fowl mites do not significantly affect egg production, nonsignificant decreases in production due to mites may result in significant reductions in farm profits.

14. Application of acaricides can cause temporary nonsignificant drops in egg production that result in monetary losses. Hens in the latter part of the laying cycle may not be able to regain a rate-of-lay high enough to recover these losses. More research is needed in this area.

15. Mite populations do not affect the immediate production level of hens. However, the affects of present mite populations may be reflected by nonsignificant changes in egg production 6 to 8 weeks in the future.

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APPENDICES

Appendix 1A. Raw data from first *Ophyra aenescens* adult longevity study.

Day of Age	Male Mortality No.	Σ	%	Female Mortality No.	Σ	%	Total Mortality No.	Σ	%
2	67	-	87	38	-	48.7	105	-	67.7
3	10	77	100	17	55	70.5	27	132	85.2
4	-	-	-	6	61	78.2	6	138	89.0
19	-	-	-	1	62	79.5	1	139	89.7
20	-	-	-	1	63	80.8	1	140	90.3
29	-	-	-	4	67	85.9	4	144	92.9
30	-	-	-	7	74	94.9	7	151	97.4
31	-	-	-	4	78	100.0	4	155	100.0

Appendix 1B. Raw data from second *Ophyra aenescens* adult longevity study.

Day of Age	Male Mortality			Female Mortality			Total Mortality		
	No.	Σ	%	No.	Σ	%	No.	Σ	%
6	-	-	-	1	1	9	1	1	3
10	2	2	10	-	-	-	2	3	10
11	1	3	15	1	2	18	2	5	16
16	3	6	30	-	-	-	3	8	26
17	-	-	-	1	3	27	1	9	29
18	3	9	45	-	-	-	3	12	39
20	4	13	65	2	5	45	6	18	58
21	1	14	70	-	-	-	1	19	61
22	2	16	80	1	6	55	3	22	71
23	1	17	85	-	-	-	1	23	74
24	-	-	-	1	7	64	1	24	77
25	-	-	-	1	8	73	1	25	81
27	1	18	90	1	9	82	2	27	87
28	1	19	95	1	10	91	2	29	94
30	1	20	100	-	-	-	1	30	97
31	-	-	-	1	11	100	1	31	100

Appendix 1C. Raw data from third *Ophyra aenescens* adult longevity study.

Day of Age	Male Mortality			Female Mortality			Total Mortality		
	No.	Σ	%	No.	Σ	%	No.	Σ	%
3	-	-	-	1	1	0.5	1	1	0.3
4	1	1	0.6	1	2	0.9	2	3	0.8
6	1	2	1.1	-	-	-	1	4	1.0
8	1	3	1.7	1	3	1.4	2	6	1.5
9	2	5	2.8	-	-	-	2	8	2.0
12	1	6	3.3	-	-	-	1	9	2.2
13	6	12	6.6	-	-	-	6	15	3.8
15	9	21	11.6	3	6	2.8	12	27	6.8
16	28	49	27.2	5	11	5.1	33	60	15.1
17	25	74	41.1	8	19	8.8	33	93	23.4
18	21	95	52.8	13	32	14.7	34	127	32.0
19	21	116	64.4	8	40	18.4	29	156	39.3
20	4	120	66.7	12	52	24.0	16	172	43.3
21	12	132	72.3	20	72	33.2	32	204	51.4
22	11	143	79.4	36	108	49.8	47	251	63.2
23	8	151	83.9	30	138	63.6	38	289	72.8
24	11	162	90.0	20	158	72.8	31	320	80.6
25	1	163	90.6	21	179	82.5	22	342	86.1
26	4	167	92.8	8	187	86.2	12	354	89.2
27	4	171	95.0	6	193	88.9	10	364	91.7
28	2	173	96.1	3	196	90.3	5	369	92.9
29	2	175	97.2	1	197	90.8	3	372	93.7
30	1	176	97.8	4	201	92.6	5	377	95.0

Appendix 1C. Continued.

Day of Age	Male Mortality			Female Mortality			Total Mortality		
	No.	Σ	%	No.	Σ	%	No.	Σ	%
31	1	177	98.3	-	-	-	1	378	95.2
32	-	-	-	4	205	94.5	4	382	96.2
33	1	178	98.9	1	206	94.9	2	384	96.7
34	1	179	99.4	3	209	96.3	4	388	97.7
35	1	180	100.0	1	210	96.8	2	390	98.2
36	-	-	-	1	211	97.2	1	391	98.5
40	-	-	-	1	212	97.7	1	392	98.7
42	-	-	-	1	213	98.2	1	393	99.0
45	-	-	-	1	214	98.6	1	394	99.2
47	-	-	-	3	217	100.0	3	397	100.0

Appendix 1D. Raw data from fourth *Ophyra aenescens* adult longevity study.

Day of Age	Male Mortality			Female Mortality			Total Mortality		
	No.	Σ	%	No.	Σ	%	No.	Σ	%
3	2	2	0.4	-	-	-	2	2	0.2
5	12	14	3.2	34	34	8.0	46	48	5.6
6	32	46	10.5	43	77	18.1	75	123	14.2
7	2	48	11.0	7	84	19.7	9	132	15.3
8	-	-	-	5	89	21.0	5	137	15.9
9	10	58	13.2	19	108	25.4	29	166	19.2
10	5	63	14.4	3	111	26.1	8	174	20.1
11	7	70	16.0	1	112	26.3	8	182	21.1
12	20	90	20.5	3	115	27.0	23	205	23.7
13	27	117	26.7	7	122	28.6	34	239	27.7
14	49	166	37.9	8	130	30.5	57	296	34.3
15	87	253	57.8	23	153	35.9	110	406	47.0
16	72	325	74.2	32	185	43.4	104	510	59.0
17	27	352	80.4	33	218	51.1	60	570	66.0
18	37	389	88.8	39	257	60.3	76	646	74.8
19	17	406	92.7	24	281	66.0	41	687	79.5
20	13	419	95.7	37	318	74.6	50	737	85.3
21	5	424	96.8	15	333	78.2	20	757	87.6
22	7	431	98.4	33	366	85.9	40	797	92.2
24	-	-	-	17	383	89.9	17	814	94.2
25	-	-	-	14	397	93.2	14	828	95.8
26	-	-	-	7	404	94.8	7	835	96.6
27	-	-	-	6	410	96.2	6	841	97.3

Appendix 1D. Continued.

Day of Age	Male Mortality			Female Mortality			Total Mortality		
	No.	Σ	%	No.	Σ	%	No.	Σ	%
28	3	434	99.1	4	414	97.2	7	848	98.1
29	1	435	99.3	2	416	97.7	3	851	98.5
30	-	-	-	2	418	98.1	2	853	98.7
32	2	437	99.7	3	421	98.9	5	858	99.3
36	1	438	100.0	3	424	99.5	4	862	99.8
39	-	-	-	2	426	100.0	2	864	100.0

Appendix 2. Raw data from CGA 72662 laboratory studies.

Fly Species		CGA 72662 Concentrations(%)				
M. domestica		control	0.06	0.05	0.04	0.03
		7/10[a]	0/10	0/10	0/10	0/10
		7/10	0/10	0/10	0/10	0/10
		9/10	0/10	0/10	0/10	0/10
		6/10	0/10	0/10	0/10	0/10
	Σ	29/40	0/40	0/40	0/40	0/40
	% Mortality	27.5	100.0	100.0	100.0	100.0
H. illucens[b]		control	0.10	0.075	0.050	0.025
		10/10	0/10	2/10	0/10	6/10
		10/10	0/10	1/10	3/10	5/10
		10/10	0/10	0/10	3/10	6/10
		10/10	0/10	2/10	0/10	2/10
	Σ	40/40	0/40	5/40	6/40	19/40
	% Mortality	0.0	100.0	87.5	85.0	52.5
S. robusta		control	0.10	0.075	0.050	0.025
		8/10	0/10	0/10	0/10	0/10
		9/10	0/10	0/10	0/10	0/10
		10/10	0/10	0/10	0/10	0/10
		10/10	0/10	0/10	0/10	0/10
	Σ	37/40	0/40	0/40	0/40	0/40
	% Mortality	7.5	100.0	100.0	100.0	100.0
P. regina		control	0.10	0.075	0.050	0.025
		10/10	0/10	0/10	0/10	0/10
		8/10	0/10	0/10	0/10	0/10
		10/10	0/10	0/10	0/10	0/10
		10/10	0/10	0/10	0/10	0/10
	Σ	38/40	0/40	0/40	0/40	0/40
	% Mortality	5.0	100.0	100.0	100.0	100.0

Appendix 2. Continued.

Fly Species		CGA 72662 Concentrations(%)			
O. aenescens	control	0.10	0.075	0.050	0.025
	8/10	0/10	0/10	0/10	0/10
	8/10	0/10	0/10	0/10	0/10
	8/10	0/10	0/10	0/10	0/10
	8/10	0/10	0/10	0/10	0/10
Σ	32/40	0/40	0/40	0/40	0/40
% Mortality	20.0	100.0	100.0	100.0	100.0
F. canicularis	control	0.10	0.075	0.050	0.025
	9/10	4/10*	2/10*	0/10†	5/10†
	6/10	1/10*	3/10*	5/10†	6/10†
	5/10	1/10*	7/10*	2/10†	5/10†
	10/10	3/10*	5/10*	3/10†	3/10†
Σ	30/40	9/40	17/40	10/40	19/40
% Mortality	25.0	77.5	57.5	75.0	52.5

[a] Represents the number of pupae that eclosed unless otherwise stated.

[b] Values are the number of pupae that formed. Pupal mortality was 100% in all cases except the controls.

* Indicates the presence of dead larvae.

† Indicates the presence of larviform pupae.

Appendix 3. House flies killed in Farnam bait field trial.

House No.	1 Kill'em Fly Killer II		2 Kill'em Fly Bait		3 Kill'em Fly Killer	
	o	o	o	o	o	o
1	6	11	33	38	1	0
3	8	8	7	10	1	2
1	6	8	0	1	0	0
3	7	9	2	0	0	0
Total by Sex	27	37	42	49	2	2
Grand Total	64		91		4	

House No.	1+5 Kill'em Fly Killer II + Lure'em II		2+5 Kill'em Fly Bait + Lure'em II		3+5 Kill'em Fly Killer + Lure'em II	
	o	o	o	o	o	o
2	92	280	172	288	17	52
4	72	144	63	157	12	62
2	97	285	43	81	0	10
4	81	215	21	104	1	3
Total by Sex	342	924	299	630	30	127
Grand Total	1266		929		157	

Appendix 3. Continued.

House No.	4 SX-70 Fly Bait		6 Golden Malrin™ w/o Muscamone		7 Golden Malrin™ with Muscamone	
	o	o	o	o	o	o
1	53	45	31	50	5	14
3	39	63	17	20	6	21
1	2	1	3	2	1	0
3	8	18	1	2	0	2
Total by Sex	102	127	52	74	12	37
Grand Total	229		126		49	

House No.	4+5 SX-70 Fly Bait + Lure'em II		6+5 Golden Malrin™ w/o Muscamone + Lure'em II		7 Golden Malrin™ with Muscamone	
	o	o	o	o	o	o
2	12	26	17	49	15	18
4	17	44	17	61	8	16
2	5	20	3	8	2	1
4	5	32	0	5	1	2
Total by Sex	39	122	37	123	26	37
Grand Total	161		160		63	

Appendix 4. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of SBP-1382 dosage-mortality data.

OBS	PROB	PROBIT	LDOSE	LOWER	UPPER
1	0.01	2.67	-11.02	-17.62	-9.65
2	0.02	2.95	-10.67	-16.61	-9.44
3	0.03	3.12	-10.46	-15.96	-9.31
4	0.04	3.25	-10.29	-15.47	-9.21
5	0.05	3.36	-10.16	-15.08	-9.12
6	0.06	3.45	-10.05	-14.75	-9.05
7	0.07	3.52	-9.95	-14.45	-8.99
8	0.08	3.59	-9.86	-14.19	-8.93
9	0.09	3.66	-9.78	-13.95	-8.88
10	0.10	3.72	-9.71	-13.73	-8.84
11	0.15	3.96	-9.40	-12.82	-8.64
12	0.20	4.16	-9.15	-12.11	-8.43
13	0.25	4.33	-8.94	-11.50	-8.33
14	0.30	4.48	-8.76	-10.95	-8.20
15	0.35	4.61	-8.58	-10.46	-8.07
16	0.40	4.75	-8.42	-9.99	-7.94
17	0.45	4.87	-8.26	-9.56	-7.79
18	0.50	5.00	-8.10	-9.16	-7.63
19	0.55	5.13	-7.94	-8.78	-7.44
20	0.60	5.25	-7.78	-8.44	-7.20
21	0.65	5.39	-7.62	-8.16	-6.90
22	0.70	5.52	-7.44	-7.92	-6.51
23	0.75	5.67	-7.25	-7.71	-6.04
24	0.80	5.84	-7.04	-7.52	-5.47
25	0.85	6.04	-6.80	-7.33	-4.79
26	0.90	6.28	-6.49	-7.11	-3.90
27	0.91	6.34	-6.42	-7.06	-3.69
28	0.92	6.41	-6.34	-7.00	-3.45
29	0.93	6.48	-6.25	-6.94	-3.19
30	0.94	6.55	-6.15	-6.88	-2.90
31	0.95	6.64	-6.04	-6.80	-2.57
32	0.96	6.75	-5.90	-6.72	-2.15
33	0.97	6.88	-5.74	-6.61	-1.73
34	0.98	7.05	-5.52	-6.48	-1.06
35	0.99	7.33	-5.13	-6.26	-0.04

Appendix 5. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of BW 21Z dosage-mortality data.

OBS	PROB	PROBIT	LDOSE	LOWER	UPPER
1	0.01	2.67	-10.71	-11.87	-10.13
2	0.02	2.95	-10.43	-11.44	-9.91
3	0.03	3.12	-10.25	-11.17	-9.78
4	0.04	3.25	-10.11	-10.97	-9.67
5	0.05	3.36	-10.00	-10.81	-9.59
6	0.06	3.45	-9.91	-10.67	-9.52
7	0.07	3.52	-9.83	-10.55	-9.46
8	0.08	3.59	-9.75	-10.44	-9.40
9	0.09	3.66	-9.69	-10.34	-9.35
10	0.10	3.72	-9.63	-10.25	-9.30
11	0.15	3.96	-9.37	-9.87	-9.10
12	0.20	4.16	-9.17	-9.58	-8.94
13	0.25	4.33	-9.00	-9.33	-8.80
14	0.30	4.48	-8.84	-9.12	-8.66
15	0.35	4.61	-8.70	-8.93	-8.53
16	0.40	4.75	-8.56	-8.75	-8.39
17	0.45	4.87	-8.43	-8.61	-8.24
18	0.50	5.00	-8.30	-8.47	-8.08
19	0.55	5.13	-8.17	-8.35	-7.91
20	0.60	5.25	-8.04	-8.23	-7.73
21	0.65	5.39	-7.90	-8.11	-7.54
22	0.70	5.52	-7.76	-8.00	-7.33
23	0.75	5.67	-7.60	-7.87	-7.10
24	0.80	5.84	-7.43	-7.74	-6.85
25	0.85	6.04	-7.23	-7.58	-6.55
26	0.90	6.28	-6.97	-7.38	-6.17
27	0.91	6.34	-6.91	-7.34	-6.08
28	0.92	6.41	-6.85	-7.29	-5.98
29	0.93	6.48	-6.77	-7.23	-5.87
30	0.94	6.55	-6.69	-7.17	-5.75
31	0.95	6.64	-6.60	-7.10	-5.61
32	0.96	6.75	-6.49	-7.01	-5.44
33	0.97	6.88	-6.35	-6.91	-5.24
34	0.98	7.05	-6.17	-6.78	-4.97
35	0.99	7.33	-5.89	-6.56	-4.55

Appendix 6. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of SD 43775 dosage-mortality data.

OBS	PROB	PROBIT	LOGDO	LOWER	UPPER
1	0.01	2.67	-10.07	-11.08	-9.49
2	0.02	2.95	-9.79	-10.70	-9.23
3	0.03	3.12	-9.62	-10.46	-9.14
4	0.04	3.25	-9.49	-10.23	-9.04
5	0.05	3.36	-9.38	-10.13	-8.95
6	0.06	3.45	-9.29	-10.01	-8.88
7	0.07	3.52	-9.21	-9.90	-8.82
8	0.08	3.59	-9.14	-9.80	-8.76
9	0.09	3.66	-9.08	-9.71	-8.71
10	0.10	3.72	-9.02	-9.63	-8.66
11	0.15	3.96	-8.77	-9.29	-8.46
12	0.20	4.16	-8.58	-9.02	-8.31
13	0.25	4.33	-8.41	-8.80	-8.17
14	0.30	4.48	-8.26	-8.60	-8.04
15	0.35	4.61	-8.12	-8.41	-7.92
16	0.40	4.75	-7.98	-8.24	-7.80
17	0.45	4.87	-7.86	-8.08	-7.68
18	0.50	5.00	-7.73	-7.93	-7.56
19	0.55	5.13	-7.60	-7.78	-7.43
20	0.60	5.25	-7.48	-7.65	-7.29
21	0.65	5.39	-7.34	-7.51	-7.13
22	0.70	5.52	-7.20	-7.38	-6.96
23	0.75	5.67	-7.05	-7.25	-6.77
24	0.80	5.84	-6.88	-7.10	-6.55
25	0.85	6.04	-6.69	-6.94	-6.29
26	0.90	6.28	-6.44	-6.73	-5.95
27	0.91	6.34	-6.38	-6.69	-5.87
28	0.92	6.41	-6.32	-6.63	-5.79
29	0.93	6.48	-6.25	-6.58	-5.69
30	0.94	6.55	-6.17	-6.51	-5.58
31	0.95	6.64	-6.08	-6.44	-5.45
32	0.96	6.75	-5.97	-6.36	-5.31
33	0.97	6.88	-5.84	-6.25	-5.13
34	0.98	7.05	-5.67	-6.11	-4.89
35	0.99	7.33	-5.39	-5.90	-4.51

Appendix 7. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of ICI Ectiban™ dosage-mortality data.

OBS	PROB	PROBIT	LOGDOS	LOWER	UPPER
1	0.01	2.67	-11.73	-12.73	-11.03
2	0.02	2.95	-11.37	-12.31	-10.79
3	0.03	3.12	-11.14	-12.00	-10.60
4	0.04	3.25	-10.97	-11.78	-10.46
5	0.05	3.36	-10.82	-11.59	-10.35
6	0.06	3.45	-10.71	-11.43	-10.25
7	0.07	3.52	-10.60	-11.30	-10.16
8	0.08	3.59	-10.51	-11.17	-10.09
9	0.09	3.66	-10.42	-11.06	-10.02
10	0.10	3.72	-10.34	-10.90	-9.95
11	0.15	3.96	-10.02	-10.54	-9.68
12	0.20	4.16	-9.76	-10.20	-9.46
13	0.25	4.33	-9.54	-9.92	-9.27
14	0.30	4.48	-9.34	-9.67	-9.10
15	0.35	4.61	-9.15	-9.45	-8.93
16	0.40	4.75	-8.98	-9.24	-8.76
17	0.45	4.87	-8.81	-9.04	-8.60
18	0.50	5.00	-8.64	-8.86	-8.42
19	0.55	5.13	-8.47	-8.69	-8.24
20	0.60	5.25	-8.31	-8.52	-8.05
21	0.65	5.39	-8.13	-8.35	-7.84
22	0.70	5.52	-7.95	-8.18	-7.61
23	0.75	5.67	-7.75	-8.01	-7.37
24	0.80	5.84	-7.52	-7.82	-7.08
25	0.85	6.04	-7.27	-7.60	-6.75
26	0.90	6.28	-6.94	-7.33	-6.33
27	0.91	6.34	-6.85	-7.25	-6.23
28	0.92	6.41	-6.78	-7.19	-6.11
29	0.93	6.48	-6.68	-7.12	-5.99
30	0.94	6.55	-6.58	-7.03	-5.85
31	0.95	6.64	-6.46	-6.93	-5.70
32	0.96	6.75	-6.32	-6.82	-5.51
33	0.97	6.88	-6.15	-6.66	-5.28
34	0.98	7.05	-5.92	-6.49	-4.94
35	0.99	7.33	-5.55	-6.20	-4.50

Appendix 8. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of carbaryl dosage-mortality data.

OBS	PROB	PROBIT	LOGDOSE	LOWER	UPPER
1	0.01	2.67	-18.19	-23.54	-15.78
2	0.02	2.95	-17.23	-22.01	-15.09
3	0.03	3.12	-16.64	-21.04	-14.65
4	0.04	3.25	-16.19	-20.31	-14.32
5	0.05	3.36	-15.82	-19.71	-14.05
6	0.06	3.45	-15.51	-19.21	-13.82
7	0.07	3.52	-15.23	-18.77	-13.61
8	0.08	3.59	-14.96	-18.37	-13.43
9	0.09	3.66	-14.76	-18.01	-13.27
10	0.10	3.72	-14.56	-17.68	-13.11
11	0.15	3.96	-13.70	-16.31	-12.48
12	0.20	4.16	-13.02	-15.23	-11.97
13	0.25	4.33	-12.44	-14.31	-11.53
14	0.30	4.48	-11.92	-13.50	-11.12
15	0.35	4.61	-11.44	-12.75	-10.73
16	0.40	4.75	-10.98	-12.06	-10.34
17	0.45	4.87	-10.53	-11.41	-9.95
18	0.50	5.00	-10.09	-10.82	-9.51
19	0.55	5.13	-9.66	-10.28	-9.03
20	0.60	5.25	-9.21	-9.79	-8.48
21	0.65	5.39	-8.75	-9.34	-7.85
22	0.70	5.52	-8.27	-8.91	-7.14
23	0.75	5.67	-7.75	-8.48	-6.35
24	0.80	5.84	-7.16	-8.02	-5.44
25	0.85	6.04	-6.49	-7.50	-4.38
26	0.90	6.28	-5.83	-6.86	-3.02
27	0.91	6.34	-5.43	-6.70	-2.69
28	0.92	6.41	-5.20	-6.54	-2.33
29	0.93	6.48	-4.96	-6.35	-1.94
30	0.94	6.55	-4.68	-6.15	-1.49
31	0.95	6.64	-4.37	-5.92	-0.99
32	0.96	6.75	-4.00	-5.65	-0.40
33	0.97	6.88	-3.55	-5.31	0.33
34	0.98	7.05	-2.95	-4.87	1.30
35	0.99	7.33	-1.99	-4.18	2.83

Appendix 9. The probabilities, probits, log doses, and lower and upper fiducial limits from the probit analysis of malathion dosage-mortality data.

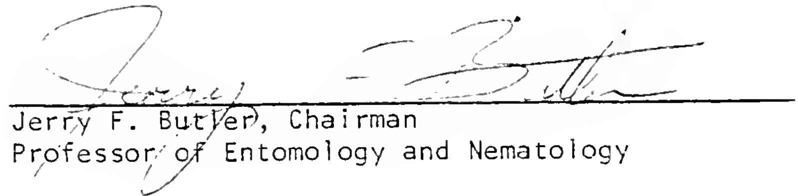
OBS	PROB	PROBIT	LDOSE	LOWER	UPPER
1	0.01	2.67	-13.52	-15.35	-12.43
2	0.02	2.95	-12.95	-14.59	-11.97
3	0.03	3.12	-12.59	-14.12	-11.68
4	0.04	3.25	-12.32	-13.74	-11.46
5	0.05	3.36	-12.10	-13.45	-11.28
6	0.06	3.45	-11.91	-13.20	-11.13
7	0.07	3.52	-11.75	-12.98	-10.99
8	0.08	3.59	-11.60	-12.78	-10.83
9	0.09	3.66	-11.47	-12.61	-10.77
10	0.10	3.72	-11.34	-12.44	-10.67
11	0.15	3.96	-10.83	-11.77	-10.25
12	0.20	4.15	-10.42	-11.23	-9.91
13	0.25	4.33	-10.08	-10.78	-9.61
14	0.30	4.48	-9.76	-10.38	-9.35
15	0.35	4.61	-9.47	-10.01	-9.09
16	0.40	4.75	-9.20	-9.67	-8.84
17	0.45	4.87	-8.93	-9.35	-8.59
18	0.50	5.00	-8.67	-9.04	-8.33
19	0.55	5.13	-8.41	-8.75	-8.06
20	0.60	5.25	-8.14	-8.47	-7.77
21	0.65	5.39	-7.86	-8.20	-7.45
22	0.70	5.52	-7.57	-7.93	-7.11
23	0.75	5.67	-7.25	-7.64	-6.72
24	0.80	5.84	-6.91	-7.34	-6.27
25	0.85	6.04	-6.50	-6.99	-5.75
26	0.90	6.28	-6.00	-6.57	-5.03
27	0.91	6.34	-5.87	-6.47	-4.92
28	0.92	6.41	-5.74	-6.36	-4.74
29	0.93	6.48	-5.59	-6.23	-4.54
30	0.94	6.55	-5.42	-6.10	-4.33
31	0.95	6.64	-5.24	-5.95	-4.08
32	0.96	6.75	-5.01	-5.77	-3.78
33	0.97	6.88	-4.74	-5.55	-3.42
34	0.98	7.05	-4.38	-5.25	-2.94
35	0.99	7.33	-3.31	-4.80	-2.13

BIOGRAPHICAL SKETCH

Jerome Adkins Hogsette, Jr., was born on March 5, 1945, in Miami, Florida. He graduated from North Miami Senior High School in June of 1963, and received an A.A. from Miami-Dade Junior College in April of 1966. In September of 1966, he entered the University of Florida, but joined the U.S.A.F. in May of 1968 prior to the completion of his bachelor's program. During a 4-year tour of duty in the Air Force, the author was involved with sanitary inspections of food and food service facilities, and health care programs for sentry dogs and animals belonging to military personnel.

After separating from the Air Force in March of 1972, Jerome re-entered the University of Florida and received a Bachelor of Science in Poultry Science in December of 1973. In August of 1975, he received a Master of Science in Agriculture with a major in poultry science and a minor in entomology. In January of 1976, the author began working on a Ph.D. program in entomology, which he is presently completing. Jerome is a member of the Entomological Society of America.

I certify that I have read this study and that in my opinion it conforms to acceptable standards of scholarly presentation and is fully adequate, in scope and quality, as a dissertation for the degree of Doctor of Philosophy.



Jerry F. Butler, Chairman
Professor of Entomology and Nematology

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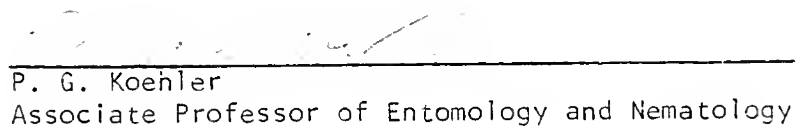

P. G. Koehler
Associate Professor of Entomology and Nematology

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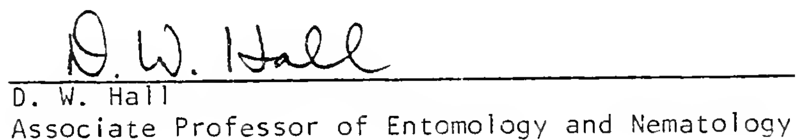

D. W. Hall
Associate Professor of Entomology and Nematology

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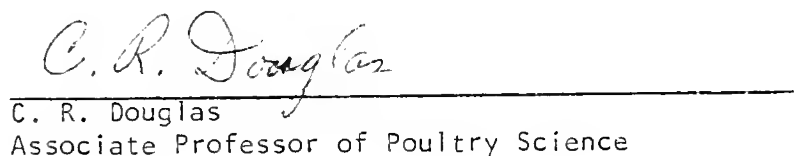
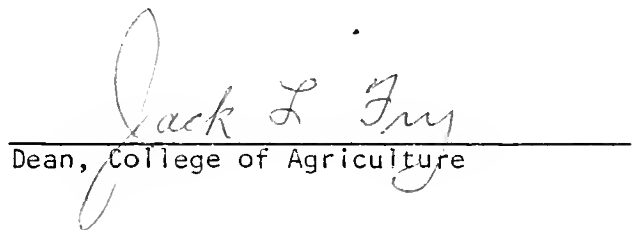


C. R. Douglas
Associate Professor of Poultry Science

This dissertation was submitted to the Graduate Faculty of the College of Agriculture and to the Graduate Council, and was accepted as partial fulfillment of the requirements for the degree of Doctor of Philosophy.

December, 1979



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