RELATIVE EFFECTIVENESS OF COMMERCIAL FLY TRAPS AND AMMONIUM CARBONATE/YEAST IN CATCHING HOUSE FLIES (DIPTERA: MUSCIDAE)

By

RYAN MERRILL WELCH

A THESIS PRESENTED TO THE GRADUATE SCHOOL
OF THE UNIVERSITY OF FLORIDA IN PARTIAL FULFILLMENT
OF THE REQUIREMENTS FOR THE DEGREE OF
MASTER OF SCIENCE

UNIVERSITY OF FLORIDA

2006

ACKNOWLEDGMENTS

I express deep gratitude to Dr. Phil Koehler for his direction in this thesis and in my graduate study as a whole. His guidance and concern have been extraordinary. I also acknowledge my graduate committee—Dr. Chris Geden, Dr. Phil Kaufmann, and Dr. Richard Patterson—for their help in making this thesis possible with revisions and general support. Also, thanks go to Dr. Phil Kaufmann for allowing space in his laboratory to rear house flies for my experiments. Ricky Vazquez, Jeff Hertz, and Matt Aubuchon offered valuable help in rearing house flies. Special thanks go to my friends and co-workers in the urban entomology lab, especially Gil Marshall and Osborne "Tiny" Willis, for their help in all things related to the urban laboratory. Debbie Hall was an invaluable help with so many aspects of my graduate experience from registering for classes to general advice. Finally, I would like to thank my family for their lifelong support of my academic endeavors.

TABLE OF CONTENTS

page

LIST OF TABLES

LIST OF FIGURES

Abstract of Thesis Presented to the Graduate School
of the University of Florida in Partial Fulfillment of the
Requirements for the Degree of Master of Science

RELATIVE EFFECTIVENESS OF COMMERCIAL FLY TRAPS AND AMMONIUM CARBONATE/YEAST IN CATCHING HOUSE FLIES (DIPTERA: MUSCIDAE)

By

Ryan Merrill Welch

December 2006

Chair: P.G. Koehler
Major Department: Entomology and Nematology

House flies are important pests that are best controlled by using integrated pest management (IPM) because of the house fly's ability to develop insecticide resistance and its considerable reproduction potential. Commonly used IPM tactics include sanitation, insecticides, biological control agents, and fly traps.

My studies deal with the ability of commercial fly traps to catch house flies and the evaluation of house fly behavior in relation to fly trap catches. In a series of experiments, house flies were released into screened cages fitted with commercial fly trap funnels. The released house flies escaped the cage either into the laboratory room or into fly traps. However, despite the seemingly random escape from the release cages, air movement may also affect house fly dispersal behavior. In my experiments, more house flies scattered into the laboratory room than were captured in the un-baited CDC net trap, and more house flies were captured in the un-baited CDC net trap than in the un-baited Trap n' Toss™. The behavior of the house flies in my experiments showed the house flies' tendency to fly upwind. Six commercial fly traps were compared for their ability to capture house flies released into screened cages. The Trap n' Toss™ fly trap caught significantly more house flies than the Fly Terminator® Pro fly trap, and this was probably due to a number of design factors including a large entrance hole area, a small

exit hole area, a large ratio of entrance to exit hole, the cone slope being close to 60^{o}, and the color of the entrance cone.

Two commonly used materials in commercial fly trap attractants, yeast and ammonium carbonate, were tested to identify their attractancy to adult house flies released into screen cages. In my studies, yeast was more attractive than water at 0, 2, and 4 days old by ~30-35%. My studies also found ammonium carbonate was never more attractive than water, and when added to yeast did not increase attractancy. Therefore, ammonium carbonate was not a chemical attractant for the adult house fly when used alone or in conjunction with the other attractants tested. Yeast, being a living organism, had a life cycle that was subject to availability of nutrition. The attractant combination (yeast and ammonium carbonate in a 41.67:1 ratio) in my study increased linearly from day 1 to 4. My results suggest that when using yeast to attract house flies, as long as the yeast is protected from temperature extremes, house fly attraction will increase linearly for at least 4 days.

My studies have shown that commercial fly traps have great potential as house fly IPM tools. They are versatile devices to be used in IPM programs, and can be used both as control devices and for monitoring house fly populations. According to my laboratory studies, and with the further exploration with field studies, commercial fly traps represent a viable tool for managing house flies in a wide variety of settings, including agriculture, urban, and military.

CHAPTER 1
REVIEW OF LITERATURE

Distribution and Classification

The house fly, *Musca domestica* L., belongs to the kingdom Animalia, phylum Arthropoda, class Insecta, order Diptera, suborder Cyclorrhapha, family Muscidae, subfamily Muscinae (West 1951). Linnaeus described the species in 1758. Other common names include typhoid fly, cholera fly, dysentery fly, and enteric fly after important diseases thought to be vectored by the fly at certain times and places (West 1951).

The house fly is for all practical purposes considered worldwide in its distribution, although it may not be found in arctic regions or places of high altitude (West 1951). The fact remains that it is a nearly ubiquitous, synanthropic insect that is found from sub-polar regions to the tropics (Graham-Smith 1914, West 1951).

Morphology/Identification

The house fly is a gray insect that is usually 4 to 9 mm long depending on available resources. The wings are held directed posteriorly when at rest. There is a sharp bend in the fourth longitudinal wing vein. There are four darkened, parallel stripes running vertically down the thorax (West 1951). The abdomen is a gray to yellow color with a dark midline and dark markings set irregularly. Females are usually slightly larger than males and are easily distinguished by noting the space between the eyes, in which the females' is almost twice as large (West 1951).

Life Cycle

The house fly, like all Diptera, is holometabolous, with distinct egg, larva, pupa, and adult stages (Chapman 1998). The first stage in any insect development, including *M. domestica*, is the egg. The eggs are white and appear polished, due to a clear, viscous coating (Newstead

1908, West 1951). They are about one millimeter in length and have a pattern of small hexagonal markings due to the shape of the egg tube in which the ovum developed (West 1951). The egg may hatch in as little as 7 hours and 35 minutes when the temperature is an optimum 34 to 38^{o}C and in as long as three days if the temperature is as low as 10^{o}C (Hewitt 1914, Larsen and Thomsen 1940). The usual time to hatch is between eight and 20 hours. Eggs that become too dry will not hatch.

Larvae, or maggots, emerge from the egg by way of a slit that appears on the dorsal side of the egg (West 1951). The larvae are white to cream-colored and possess no eyes, legs, antennae, or other appendages (West 1951). The larva must pass through three instars reaching 7-12 mm before pupating (Newstead 1908, West 1951). The third instar migrates from the food source to a dry, cool place to pupate. The larval stage can take anywhere from five days to eight weeks, depending on the temperature and food quality (Newstead 1908).

The fly reaches the pupal stage when the movements of the contracted larvae have ceased and it is still a cream color, and ends when the adult fly emerges (Larsen and Thomsen 1940). The puparium color quickly changes from the cream color darkening to a reddish brown or black (Newstead 1908, West 1951). It is usually around 6 to 7 mm long and weighs about 21 mg (Newstead 1908, West 1951). The pupal stage typically accounts for more than half of the immature development time of the fly. It may last anywhere from 3.75 to 28 days, depending on the food of the larval stage and the temperature (Newstead 1908, Larsen and Thomsen 1940). Development time is inversely proportionate to temperature to a certain threshold. The upper temperature range in which retardation becomes apparent is between 36 and 40^{o}C (Larsen and Thomsen 1940). If the medium reaches temperature below 10^{o}C, there is a considerable level of mortality (Sacca 1963).

The adult, or imagio, emerges from the puparium by breaking away the anterior end with the ptilinum found on the frontal region of the head (Newstead 1908, West 1951). The fly escapes the puparium and escapes the pupal medium such as manure, debris, sand, etc. by contracting the ptilinum (West 1951). This important structure is expanded and contracted by changes in blood pressure and retraction of muscles (Laing 1935, West 1951).

The newly emerged adult crawls about rapidly as the wings unfold and the exoskeleton becomes hard. The wings become flat, thin, and transparent by withdrawal of fluids and are supported by the veins. At last, the ptilinum is withdrawn and only the frontal "lunule" that marks its former location is left behind (Newstead 1908, West 1951). Overall, the whole process from egg to adult, like every life stage, varies with temperature. However, when the medium is 33.22^{o}C, the process takes about 6.5 days (Hogsette 1995).

Flight and Dispersal

House flies are able to fly considerable distances. The extent of this distance is different according to different authors. Bishopp and Laake (1921) found that within 24 hours of release, house flies average a distance of about 9.66 km, with a maximum distance of about 21.15 km. Schoof and Siverly (1954) conducted a study that suggests that flies can travel as far as 32.19 km by way of flight. They also found that large numbers of flies might migrate up to 6.44 km. However, most flies are not recovered at distances greater than 3.22 km. This is because house flies have been shown to fly in a more or less random pattern, until a suitable place is located (Sacca 1963). The house fly tends to travel shorter distances in urban areas where structures, sources of food, and sources of oviposition are more readily available, whereas in rural environments, the fly will have to disperse further to find suitable sites (Hindle 1914, Murvosh and Thaggard 1966). Additionally, air temperature limits the flight ability of the house fly. As temperature increases, so does the motility of the house fly (Sacca 1963).

Arguably the most important means of dispersal for the house fly, and almost certainly the reason for its worldwide distribution, is hitchhiking. Flies can travel great distances by using large mammals or human-assisted transportation (Sacca 1963).

Mating Behavior

The mating behavior of the house fly is a complicated process that, once sexual maturity is reached, is always initiated by the male. Many researchers have investigated this issue. However, the temperatures used in their studies were not always the same. Males require at least 16 hours to mature, whereas females require at least 24 hours (Murvosh et al.. 1964a). Chang (1965) determined a male maturation time of at least 20 hours and female maturation time of at least 40 hours. Despite the fact that female flies may not be ready to copulate, males persist. This may lead to a female fending off and possible damaging of the male, especially his wings, with her posterior legs that are held near the tip of the abdomen (Bishopp et al. 1915).

Copulation may occur much later even though males aggressively try. Bishopp et al. (1915) found flies mating as late as 16 days after emergence. This large range of time may be, at least in part, the result of nutrition. The nutrition may affect either the sexual development or the desire to mate. Bishopp et al. (1915) observed mating within four days of emergence on flies fed with milk and horse manure, while a group fed partially ripened peaches and horse manure required 16 days. Copulation was not observed among the unfed flies.

Sexually determined males seek females using visual cues. The male is sexually aggressive, and will attempt to mate with anything resembling a female house fly, including male house flies, fly cadavers, and inanimate objects (Rogoff et al. 1963). However, visual cues are not necessary for a male to locate a mate. In fact, mating can occur in complete darkness (Basden 1947, Rogoff 1965).

The male fly also uses chemical cues to identify a partner (Patterson 1957). Both male and female house flies have gender-specific hydrocarbons, as well as hydrocarbons common to both sexes (Nelson et al. 1981). The major pheromone of the house fly is found on the female. It is a C_{23} hydrocarbon called (Z)-9-tricosene (Carlson et al. 1971). The activity of this pheromone is enhanced by other hydrocarbons including methylalkanes, (Z)-9,10-epoxytricosane, and (Z)-14-tricosen-10-one (Uebel et al. 1976, Nelson et al. 1981, Bloomquist et al. 1984). It works as a short-range attractant, sex-recognition factor, arrestant, and copulatory stimulant of males (Mayer and James 1971, Mansingh et al. 1972, Carlson et al. 1974, Adams and Holt 1987, Hanley et al. 2004).

When the male does locate an appropriate partner, both flies begin seemingly random movements. The male will stop movement when he gets within a few centimeters of the female, wherein he leaps on her back (Rogoff et al. 1963). The mount may also be attempted in the air, but the couple quickly comes to a rest, as copulation never happens in flight. If disturbed, however, the couple may fly to another surface together (Murvosh et al. 1964a). Mating occurs in the superimposed position, with the male above the female and both facing the same direction. If the male is too small or disturbed, it may face at a near 90^{o} angle or even face the opposite direction (Hardy 1944, West 1951). After mounting, the male caresses the head of the female with the tarsi of his front legs. This probably has a reflex effect on the female as it usually results in ovipositor extension until it contacts the male genital atrium. The aedeagus extends as does the accessory copulatory vesicles of the female so that they contact each other. This is when the spermatozoa make their way into the spermethecae of the female (West 1951). The spermatozoa are transferred to the female in as little as three to five minutes however, the couple will usually stay in coitus for a much longer time (Murvosh et al. 1964a). Coitus has been

observed to last anywhere from 30 minutes to two hours (Hampton 1952, Murvosh et al. 1964a, Chang 1965).

After mating, the female usually remains monogamous, and only a few mate more than a few times (Riemann et al. 1967). In a study by Zimgrone et al. (1959), only two percent of the females mated multiple times. One fertilization episode is sufficient for the entire egg-laying period (Zimgrone et al. 1959). The male seminal fluid, and not mechanical stimulation or sperm, has been shown to cause the loss of female sexual receptivity (Riemann et al. 1967).

Oviposition

Many factors are involved in ovigenesis and oviposition. For instance, diet is of utmost importance. Protein is necessary for the production of mature eggs in the female house fly, as with many other dipterans (Sacca 1964). Whereas the diet is integral in the production of eggs, it is also important insofar as how quickly or slowly the eggs are oviposited. Bishopp et al.. (1915) provided one group of flies peaches for food as opposed to another lot being supplied with milk, peaches, and horse manure. Those fed peaches required 16 days to oviposit, whilst those fed the mixture of foods took five. This suggests that variety, or probably more specifically, the presence of protein, in the diet al.lows the female to oviposit more quickly.

Temperature also plays an important role in the rapidity of egg deposition. In general, within reasonable ranges, speed of egg deposition is positively correlated with temperature (Bishopp et al.. 1915). Oviposition will altogether cease if the atmospheric temperature drops to 10^{o} C or lower, regardless of the substratum temperature (Kalandadze and Chilingarova 1942, West 1951).

Humidity is also a factor in time to first oviposition. As observed with temperature, when humidity increases, the speed of egg deposition increases. The humidity may make the media is more moist, and therefore more appealing to the female flies (Bishopp et al.. 1915). However,

because liquid medium is hardly ever a suitable location, there appears to be an upper limit to this effect (Kalandadze and Chilingarova 1924).

Taking into account the variety of factors impacting egg deposition, a female house fly may require from five to twenty days post-eclosion to deposit her first batch of eggs (Griffith 1908, Bishopp et al.. 1915). Once she is ready to oviposit, she must find an appropriate medium. An appropriate medium is one that will provide ample nutrition to the larvae that will hatch from the eggs. The larvae develop most quickly in any site that is fermenting and vegetal in nature. Flies often oviposit in garbage and manure (Mellor, J.E.M. 1919, Kalandadze and Chilingarova 1942, West 1951, Sacca 1963, Cosse and Baker 1996). Another factor influencing oviposition is the presence of other flies. Jiang et al. (2002) documented increased oviposition at sites where other female flies were present, possibly due to the semiochemicals n-tricosane and (Z)-9-tricosene. When a suitable spot is located, a female fly will orient to cracks and crevices in where she either inserts her ovipositor, or crawls into the crevice so that the eggs are protected from desiccation and actinic light (Newstead 1908, West 1951).

House fly reproductive capacity is considerable. Hodge (1911) calculated that if one pair of flies, unchecked, began reproducing in April, by August there would be 191,010,000,000,000,000,000 flies. It is estimated that this would cover the surface of the earth to a depth of 47 feet. One female fly usually oviposits anywhere from 75 to 159 eggs in batches (Newstead 1908, Dunn 1923, Herms and James 1961). House flies have been reported to produce between two and twenty-one egg batches at intervals of eight to fourteen days, though usually only two to four batches are deposited (Griffith 1908, Bishopp et al. 1915, Dunn 1923).

Feeding

House flies, like all animals, require nutrition in order to maintain body processes, such as flight, movement, feeding, etc. and to provide for the building blocks of the organs, especially

the reproductive system. For the house fly, sugars or assimilable starch is required, while proteins or byproducts of protein hydrolysis are necessary for organ formation (Kobayashi 1934, West 1951).

Certain substances and chemicals are feeding attractants. Although house flies are omnivorous, there are suitable, but unattractive, foods that require stimulus for consumption. The sugars that are particularly attractive to adult flies for feeding purposes include lactose and dextrin. Sugars that are not necessarily attractive despite their nutritive potential are glucose, maltose, starch, or sucrose (Richardson 1917). Richardson (1917) also found that succinic and lactic acid were feeding stimulants. Awati and Swaminath (1920) found ammonia, hydrogen sulfide, and phosphorous compounds to be adequate to induce feeding in the house fly adult. Robbins et al. (1965) found casein, more specifically, its amino acid components (leucine, lysine, isoleucine, and methionine) and the guanonsine monophosphate of yeast hydroloyzate to be feeding stimulants.

Regardless of how attractive or nutritive a food substance is to the house fly, it can only be consumed if it is in a liquid or semi-liquid form, or is capable of being rendered so. Some solid foods are, if soluble, put into solution by the house fly's use of vomit from the crop. There are some instances where non-soluble solids will be taken up, then broken down further with prestomal teeth. After the food is ingested, it will make its way through the alimentary tract (Sacca 1963).

Longevity

The longevity of house flies, no doubt, is directly related to the temperature and humidity. West (1951) states that in the summer flies live no longer than a few weeks, whilst in the colder months, three months is realistic. Many studies have examined house fly longevity. Herms (1928) recorded a mean of 30 days. Murvosh et al. (1964b), found that at a mean of 23.9^{o} C,

males live an average of 33 days, and females 43 days, with a range of one to 99 days. Rockstein (1957) found that when the temperature was 26.7^{o}C and relative humidity was 45%, male house flies lived from 15 to 40 days, and female house flies lived from 20 to 60 days.

The age of the parents affects the longevity of the offspring. The average life span of the succeeding generations decreases if only eggs from young flies are used. Flies developing from females aged 20 to 30 days at oviposition resulted in an increase in the average lifespan (Callahan 1962).

Sex Ratio

Studies with lab colonies suggest that the sex ratio of house flies is about 1:1. Murvosh et al. (1964a) reported a sex ratio of 50.6 to 49.4 ratio based on 8700 individuals, and a ratio of 53.5 to 46.5, based on 5233 individuals. Rogoff (1965) determined his sex ratio using 16 different samples of 100 pupae each, and reported similar results.

West (1951) observed that when the average adult size is smaller than 6-7 mm in length, then males would predominate, while when the average adult size is larger, the males never predominate the population. Herms (1928) showed, while working with *Lucilia sericata* (Meigen), that larval females require more nourishment than their male counterparts to survive to the pupal stage. This being the case, an abundance of males may signify a lack of larval nutrition in the area.

Economic Importance

House flies gather at garbage, compost, sewage, manure, livestock facilities, and other unsanitary sites, and then readily enter buildings making them a considerable nuisance to people and animals (Chapman et al. 1998). Although the house fly is not as much of a problem in many advanced industrial societies due to personal transportation being automobiles instead of horses, it is still one of, if not the most, important pests in markets, stores, and animal sites due mostly to

annoyance (Cosse and Baker 1996, Robinson 1996, Chapman et al. 1998, Hogsette 2003). They become most annoying when they land on people and animals causing irritation. When they crawl on food the potential for pathogen transmission is raised. Whilst resting on walls and windows they leave behind specks from vomit and feces. Mere annoyance has turned into litigation. When flies from livestock facilities find their way into the spreading suburban and urban areas, lawsuits follow (Campbell 1993). The annoyance that the flies cause livestock may also reduce production yields (Cosse and Baker 1996, Chapman et al. 1998).

House flies are economically important as disease vectors. House flies have been long suspected of disease transmission (Howard 1911). However, it was not until the studies carried out by Watt and Lindsay (1948) and Lindsay et al. (1953), that there was convincing evidence for the house fly's involvement. After a fly has become infected with a pathogen, it can transmit the disease one of four ways. These include contact with the body setae, setae of the feet, regurgitation, and passage through the digestive tract (West 1951). While West (1951) and Greenberg (1973) indicate that of these four, the passage through the digestive tract is probably the most important because it allows for multiplication of the disease organism within the alimentary canal, Zurek (2001) shows that the potential for a pathogen to be spread via vomitus or feces is dependent on the specific pathogen.

Greenberg (1971) lists over 30 viruses, hundreds of bacteria species, as well as a number of fungi, protozoa, and nematodes that are associated with *M. domestica*. A far from exhaustive list of some of the disease organisms that house flies are known to transmit are *Escherichia coli* (including O157:H7), *Shigella* spp., *Salmonella* spp., *Helicobacter pylori*, *Yersinia pseudotuberculosis*, and the organisms responsible for typhoid fever, cholera, summer diarrhea,

dysentery, tuberculosis, anthrax, opthalmia, and parasitic worms (West 1951, Greenberg 1971, Grubel et al. 1997, Iwasa et al. 1999, Zurek et al. 2000, Geden 2005).

Control

The management of the house fly is an exercise in problem solving. West (1951) broke the control of house flies into two categories: planned control and emergency control. This approach is better known as integrated pest management (IPM) today. West's apparent foresight into pest management is due to the fact that managing house flies offers a classic opportunity for IPM. An effective house fly management strategy includes sanitation and other cultural methods, mechanical control, insecticidal control, and, more recently, biological control (West 1951, Kolbe 2004). Greene and Breisch (2002) documented the success of IPM in urban environments by comparing the number of pest control service requests. From 1988 to 1999, the requests for the group that included house flies were decreased to half its original number. Also, the amount of insecticide used was drastically decreased to about 5% of its original use.

Sanitation is the primary means of managing house fly problems by eliminating breeding sites and larval food, which also attract adult flies (Clymer 1973, Gojmerac 1979, Meyer and Shultz 1990). Thus, by eliminating trash and manure, all stages of the fly life cycle are disrupted.

In the urban setting, the main breeding site for the house fly is garbage. Kolbe (2004) offers insight into the management of garbage for house fly control. Food in trash cans and dumpsters should be picked up frequently. Twice a week is ideal. Trash cans and dumpsters should have tight-fitting lids, and should be kept closed. The garbage placed in trash cans and dumpsters should be sealed off in plastic bags to minimize attractive odors. Dumpsters should be kept as far away from building entrances as possible. Also, keeping the dumpster and the ground underneath it clean will help reduce house fly numbers.

In the agriculture, the main breeding site is manure (West 1951). Agricultural settings that have accumulated manure have higher house fly populations (Lysyk and Axtell 1986). Feedlots should allow for proper drainage that minimizes wet spots and prevents the build up of waste. Manure and spilled feed should be removed twice a week (Clymer 1973). Sanitation, especially the removal of manure, of poultry houses and dairies is also an essential tool in an integrated pest management strategy against house flies (Lysyk and Axtell 1986, Lazarus et al. 1989). Manure removal is most effective in all instances when it is done in the late spring (Axtell 1970a). Also the bedding material that is routinely excreted upon by livestock may be changed or added to-sawdust or sand-in order to make the manure less enticing as a breeding site for house flies (MacCreary and Haenlein 1962, Schmidtmann 1988, Lazarus et al. 1989).

Another cultural method to prevent house fly problems is exclusion. House flies are anthropomorphic and move to human structures through temperature and odor attractants. A way to combat this invasion is to make sure that all windows and doors have screening, that the screens lack holes, and are properly fitted (West 1951). House flies can also be excluded by fans and air doors between rooms (West 1951, Hedges 1994).

Mechanical control is another method critical to an effective house fly management regimen. Most likely the oldest control measure for house flies falls under this category - flyswatting (West 1951). More modern methods of mechanical control are the plethora of available fly traps. The different types of fly traps include baited traps, light traps, and sticky traps (West 1951, Pickens et al. 1975, Skovmand and Mourier 1986, Pickens 1989, Kaufman et al. 2005b). Of these types of traps there are a variety of different designs, attractants, and killing methods. These are effective supplements to other control methods because they are able to trap flies up to 24 hours a day, seven days a week with minimal effort (Kolbe 2004).

Insecticides can be used to treat a house fly problem. Chemicals have been used to control flies since before World War II (West 1951). But, this should not be done indiscriminately, but rather after other methods have failed to reduce the fly populations (Hedges 1993). This is due to the house fly's ability to develop resistance to almost every insecticide used against it (Learmount et al. 2002, Scott et al. 2000, Gao and Scott 2006). Not only does overuse of insecticides contribute to house fly resistance, but there are also the concerns of public perception, increased cost, safety, and environmental considerations (Carson 1962, Appling 1988, Stinson 1989). Different formulations should be considered for house fly management based on the situation. They are residual insecticides applied to adult resting sites and larval breeding sites, baits, and aerosols and ultra-low-volume (ULV) applications used as space treatments (Kolbe 2004). Insecticide treatment should be used with caution as it may hinder a proper IPM program i.e. affect natural predators that could be used as biological control agents (Wills et al. 1990).

Augmentative biological control is the newest form of house fly control and can be used as part of a successful IPM program. Fly egg predators represent one type of biological control, the two major species being *Carcinops pumilio* (Erichson) and *Machrocheles muscadomesticae* (Scopoli) (Axtell 1970b, Geden and Stoffolano 1987). *Carcinops pumilio* is capable of causing ~95% mortality of house flies, whereas *M. muscadomesticae* can cause ~80% mortality in the lab, with somewhat lower results in the field due to abiotic factors and the presence of alternate prey (Geden and Axtell 1988, Geden et al. 1988). Fly larval predators also exist as biological control agents. The black dump fly, *Ophyra aenescens* (Wiedemann), is a predator of house fly larvae and is capable of out-competing the house fly (Nolan and Kissam 1985). Although *O. aenescens* is a filth fly, it remains a suitable candidate for biological control because it is not a

nuisance. This is due to the adult habit of staying close to the breeding medium and its preference for woods and shade as opposed to human structures (Legner and Dietrick 1974, Nolan and Kissam 1987). Muscovy ducks are predators of adult house flies that may be used as a biocontrol agent. The ducks ingest about 25 house flies per 15 minutes, and controlled 90% of the house flies in a much shorter time than fly traps or baits (Glofcheskie and Surgeoner 1990).

Another group of biological control agents are the Pteromalid parasitoids that attack house fly pupae. The most common are *Muscidifurax raptor* Girault and Sanders, *Muscidifurax zaraptor* Kogan and Legner, *Muscidifurax raptorellus* Kogan and Legner, *Spalangia cameroni* Perkins, *Spalangia endius* Walker, and *Spalangia nigroaenea* Curtis (Rueda and Axtell 1985, Havron and Margalit 1991, Geden and Hogsette 2006). These are naturally occurring species that hardly ever reach peak numbers and activity before or during the peak house fly nuisance levels, therefore, the parasitoid populations must be augmented either by inoculative or inundative release (Legner and Brydon 1966, Skovard and Nachman 2004). Skovard and Nachman (2004) reported that the inundative release of *S. cameroni* usually caused house fly populations to remain under nuisance levels (13-25 house flies per animal) on both dairy and pig farms. Single species releases of *Muscidifurax spp.* and *Spalangia spp*. have had mixed results (Geden et al. 1992, Geden and Hogsette 2006). Combined releases of the two genera are preferred due to *Muscidifurax spp*. searching for hosts on the surface of manure, and *Spalangia spp*. searching deeper (Rueda and Axtell 1985). *Muscidifurax raptorellus* and *S. cameroni* simultaneously released accounted for up to ~80% pupal mortality (Geden and Hogsette 2006).

Microbes can also provide biological control of the house fly. One example is the entomopathogenic fungus, *Entomophthora muscae* Cohn. The fungus causes adult house fly death in about 5-8 days after infection (Zurek et al. 2002). Once infected, house flies have a

100% mortality rate (Steinkraus and Kramer 1987). Infection of other individuals is likely due to the males' preference to mate with infected females (Zurek et al. 2002). Another aspect that makes *E. muscae* a good biological control agent is its ability to discharge conidiophores from the cadaver of the dead fly (Zurek et al. 2002). A second entomopathogenic fungus that offers promise is *Beauveria bassiana* (Balsamo). In laboratory studies, up to 98% mortality of house flies after 15 days of exposure has been reported (Lecuona et al. 2005). In the field, *B. bassiana* treated facilities had lower house fly populations than facilities treated with pyrethrins (Kaufman et al. 2005a).

Nematodes in the genus *Heterorhabditis*, *Steinernema*, and *Paraiotonchium* have been tested against house flies in the laboratory. While some show the ability to infect house flies and show promise as control agents in the lab (*Paraiotonchium muscadomesticae* Coler caused 98% mortality with a huge concentration of nematodes), the nematodes' infectivity is highly reduced in manure, requiring larger application concentrations (Mullens et al. 1987, Geden 1997).

Fly Traps

There are three common types of house fly traps: light traps, sticky traps, and baited traps. Light traps consist of an ultraviolet lamp that attracts the house fly into an electrocuting grid (ELT) or a glue board (GLT) (Pickens 1994). Although ELTs are successful, GLTs are becoming increasingly popular due to the noise emitted every time an insect is killed, and because when electrocuted, house flies can scatter body parts and bacteria throughout the air (Pickens 1989). Though Kolbe (2004) reports that low-voltage ELTs do not introduce fly fragments into the air, Pickens (1989) observed a scatter of parts up to 1.5 m from the trap. Light traps should be placed 0.3 to 0.9 m from the ground about 15 m apart around common fly gathering places to maximize their effectiveness (Driggers 1971, Gilbert 1984, Pickens 1994). Pickens et al. (1969) reported almost 100% capture of house flies in a 7.6 m^2 room in only seven

hours. However, Thimijan et al. (1970) reported 0 to 16.3% catch of released house flies. It is not surprising that some authors suggest that light traps are not sufficient control devices and should only be used for monitoring (Schiefer 1970, Skovand and Mourier 1986, Rambo 1995).

Another type of trap that may be employed in an IPM program for house fly management is sticky traps. One common type of sticky trap is a roll of paper with glue adhesive, usually made attractive by color or printing patterns such as other flies on it. Kaufman et al. (2005b) found that these traps caught over 80,000 house flies on each of five dairy calf greenhouse facilities in only four days. They concluded that the sticky trap was responsible for a decline in house fly populations and in each case, the producer noticed a reduction in house fly numbers and was pleased. However, the efficacy of this type of trap declined in dusty environments, such as occurs on many poultry farms (Kaufman et al. 2001). Another common type of sticky trap is a pyramid sticky trap. Given the numbers of house flies trapped, these seem to be better suited for monitoring instead of control. Pickens and Miller (1987) caught 5.6 to 14.0% of released house flies with pyramid traps. They also point out that after about two days, the adhesive became saturated with flies and was unable to catch significant numbers. Pickens et al. (1986) and Johnson and Campbell (1987) describe pyramid traps as being most useful as monitoring devices, not control devices, for *Musca vetripennis* (Say) and *Musca autumnalis* De Geer respectively. Sticky tapes are also used as monitoring devices (Pickens et al. 1972, Turner Jr. and Ruszler 1989).

Baited traps are probably the oldest of the fly traps, having been in use since about 1911 (Pickens 1995). The baited fly trap consists of an odiferous attractant inside of the trap that entices the house flies to enter through an inverted cone (Bishopp and Henderson 1946, Pickens 1995). Similar to the other trap types, there is debate whether baited traps should be used as

monitoring or control devices. Dadour and Cook (1992) point out that to reduce a fly population by 50-90%, a trapping regimen must remove 24-58% of the house flies. Pickens and Miller (1987) report catching only 4.4 to 20.0% of released flies in baited traps. Smallegange (2004) states that although baited traps show promise, they are not yet effective enough to reduce populations to acceptable levels. Das (1994) reported that using Redtop Flycatcher baited with ready made protein meal was easier and less expensive to use than a Baygon bait, but admitted that it did not catch as many house flies as the Baygon bait killed. Burg and Axtell (1984) and Geden (2005) also refer to baited traps as monitoring devices. Despite Dadour and Cook's (1992) pessimism, Cohen et al. (1991) attained 64% reduction in house fly density, and attributed a 42% reduction in diarrhea visits and 85% reduction in shigellosis visits of Israeli soldiers to the trapping. Perhaps some of the low baited trapping numbers can be attributed to how the baited traps are placed, after all, daily catches of up 40,000 to 129,000 have been reported (Blair 1945, Pickens 1995). Also, seasonal catches of 1.1 to 9.7 million have been recorded (Fenton and Bieberdorf 1936, Mer and Paz 1960). Baited traps should be placed outside every 6-9 m and placed as close as possible to common fly-breeding materials (Pickens 1994). Traps are most effective when placed within 0.3 m of the ground (Mitchell and Tingle 1975).

Objectives

The first objective of these experiments was to discover the relative effectiveness of each of six commercial fly trap designs in capturing house flies. The second objective was to determine the attractiveness of yeast and ammonium carbonate to house flies, and whether combining and/or aging these ingredients would affect their attractiveness. Using this information, I would be able to make a few general conclusions as to where commercial fly traps and the tested attractants fit into an IPM program for house flies.

CHAPTER 2
MATERIALS AND METHODS

Laboratory Rearing of the House Fly

House fly larvae, *M. domestica*, Horse Teaching Unit (HTU) strain established in 2004, from Gainesville, FL, were reared in plastic tubs (33.50 by 28.41 cm) on medium containing 250 ml of Calf Manna® (Manna Pro Corp., St. Louis, MO), 1.5 liters of water, and 3 liters of wheat bran. The pupae were separated from the medium by water floatation on the seventh day and dried on paper toweling. The pupae were placed in screened rearing cages (40.64 by 26.67 by 26.67 cm) for emergence. The screened rearing cages were screened on five sides with the sixth side used as an entryway that was covered with surgical stocking net that could be tied off to prevent house fly escape. Adult house flies were provided granulated sugar, powdered milk, and water *ad libitum* (Hogsette et al. 2002). Both larvae and adults were held on a 12:12 (L:D) photoperiod, 26.2 $\pm$ 0.5° C, and 51.2 $\pm$ 3.5% RH. Adults emerged in the cage following three days as pupae. Oviposition was induced with spent larval medium at around ten days post eclosion. The eggs collected were used to sustain the colony. Males and females were held together in the same cages.

Adult house flies were aspirated from screen cages with a modified hand-held vacuum. Aspirated house flies were placed into a freezer (-30° C) until inactive (~1-5 min). After removal from the freezer, the house flies were placed on a chilled aluminum tray. House flies were counted and sexed (25 males, 25 females) and held in a deli cup (236.58 ml) for about one hour. House flies used in the design experiment and the aged attractant combination test were between three and seven days post-eclosion. House flies used in scatter and attractant component experiments were between three and five days post-eclosion.

Fly Trap Design Experiments

Scatter Experiment

An open release cage study was developed to evaluate the house flies' propensity to escape from an enclosed space in a 24 hour period. Two different trap designs were utilized - no trap, designated the laboratory room, and a CDC (Center for Disease Control) net trap (Fig. 2-1).

Each release cage (28.8 by 26.1 by 39.1 cm high) consisted of a screen cage fitted with a stocking net opening. A cotton-filled plastic cup (88.72 ml) soaked with 50 ml of 10% aqueous sugar solution was placed in release cage. House flies were then released from the deli cup into each release cage. A continuous channel was created by fitting a yellow funnel that was removed from a Trap n' Toss^{TM} (Farnam Companies, Inc., Phoenix, AZ) fly trap. The stocking net opening of the release cage was made continuous with the cone. One half of the release cages were oriented so that the cone opening was below the cage, while the other half were made so that the cone opening was above the release cage (Fig. 2-2 and 2-3). For the release cages that were left open to the laboratory room, after 24 h, the sugar water was removed, the cone removed, and the release cage was tied off for 24 h to kill the house flies remaining in the release cage. The dead house flies were counted and recorded to determine the percentage of house flies that escaped into the laboratory room by taking the difference of how many house flies were left in the release cage as opposed to how many were initially released. For those release cages fitted with CDC net traps, the CDC net traps were taken from the release cage, tied off and left to sit for 24 h to kill the flies. The flies were then counted, sexed, and recorded to determine the percentage of male, female, and total house flies that were trapped in the CDC net traps. A replication consisted of a cage for top-entry or bottom-entry treatments. Four replications were of laboratory room catch at the same time in the laboratory (22.8 ± 0.02^{o} C) on a 24:0 (L:D) photoperiod. The same procedure was done for the experiments with CDC net traps.

Commercial Fly Trap Experiment

Fly traps

A comparison of six commercial fly traps was conducted in the laboratory. Fly traps used were categorized as either bottom-entry or top-entry. Bottom-entry fly traps were designed with an inverted funnel leading into a collection container. Bottom-entry fly traps included Trap n' Toss™, The Advantage Fly Trap™ (Advantage Traps, Inc., Columbia, SC), and BC 1752 Dome (McPhail) Trap (Agrisence™ Agriculture, Pontypridd, UK) (Figs. 2-4, 2-5, and 2-6).

Top-entry fly traps were containers fitted with entrance(s) on the lid. Rescue!® Reusable Fly Trap (Sterling International, Inc., Liberty Lake, WA) and Victor Fly Magnet® Trap (Woodstream, Lititz, PA) each had four entrance holes, while the Fly Terminator® Pro (Farnam Companies, Inc., Phoenix, AZ) had only one entrance hole (Figs. 2-7, 2-8, and 2-9).

The diameter for each entrance/exit hole were measured and used to calculate area. The areas were used to calculate the entrance:exit ratios for each fly trap. The slope of the funnel entrance and the volume of each fly trap's capacity were also measured. Each entrance color was observed and recorded.

Bioassay

A fly attractant mixture consisting of 5 g yeast, 0.12 g ammonium carbonate, and 75 ml of water was placed into each fly trap immediately after mixing. A cotton-filled plastic cup soaked with 50 ml of 10% aqueous sugar solution was placed in release cage. House flies were released from the deli cup into release cage. The stocking net opening of the release cage was made continuous with the opening of the fly trap, which were either placed on top of the release cage (bottom-entry fly traps) or suspended below the release cage (top-entry fly traps) (Figs. 2-10 and 2-11). The stocking net was pulled over the set fly trap and tied off to prevent house flies from escaping between the release cage and the fly trap.

After 24 h, each fly trap was removed from the release cage, placed into a sealable plastic bag, then refrigerated at 3° C for ~ 24 h. Upon removal from the refrigerator, captured house flies were extracted from the attractant mixture using a colander, placed on a chilled aluminum tray, counted, and sexed.

The experiment was a completely randomized design comparing each fly trap's capture efficacy. Each trial was run in the laboratory (22.8 ± 0.02° C) on a 24:0 (L:D) photoperiod. The locations of the fly traps were randomized to minimize position effect. Testing included one of each fly trap (bottom-entry and top-entry) examined five times, except Fly Terminator® Pro which was left out once due to availability. On two additional occasions, bottom-entry traps were tested using two Trap n' Toss™ fly traps, one Advantage Fly Trap™, and one BC 1752 Dome Trap.

Attractant Experiments

Aged Attractant Experiment

The Trap n' Toss™ fly trap was used as a standardized fly trap to evaluate the effect of attractant age on fly capture. Sets of three groups of fly attractants were prepared separately using 5 g yeast, 0.12 g ammonium carbonate, and 75 ml of water. These mixtures were held for 0, 1, 2, 3, or 4 d at 22.8 ± 0.02° C yielding five age classes of attractant. Following the aging of each attractant, the attractants were introduced into individual fly traps. A cotton-filled plastic cup containing 10% aqueous sugar solution and the 50 house flies were introduced as previously described. The fly trap was secured on the cage, as before. Following the 24 h experiment, house flies were collected, counted, and sexed as in commercial trap experiments. Three cages of each attractant age mixture were run simultaneously (22.8 ± 0.02° C) on a 24:0 (L:D) photoperiod. The experiment was run twice. Due to contamination of release cage with insecticide, one cage of 1-d aged attractant was discarded.

Attractant Component Experiment

This study consisted of four treatments including 75 ml water control, 0.12 g ammonium carbonate, 5 g of yeast, or a combination of the ammonium carbonate and yeast, each in 75 ml of water. Each of the attractants was aged 0, 2, or 4 d prior to placement into a Trap n' Toss^{TM} fly trap. A cotton-filled plastic cup containing 10% aqueous sugar solution and the 50 house flies were introduced, then the stocking net used to secure the fly trap on the cage, as before. House flies were collected, counted, and sexed as in commercial trap experiments. Each combination was run simultaneously and was run independently six times, for a total of six replications. Experiment was run in the lab (22.8 ± 0.02^{o} C) on a 24:0 (L:D) photoperiod.

Data Analysis

Actual means are presented in tables. Data for percentage catch of males, females, and total for laboratory room, CDC net trap, each commercial trap, and each aged attractant were arcsine square root transformed and analyzed using one-way analysis of variance. Male, female, and total percent house fly catch was analyzed by linear regression for the attractant mixture aged 1-4 days. A one-way analysis of variance was run on each attractant component by days aged (0, 2, 4). Means for total, male, and female percent catch were separated using Student Newman-Keuls (SNK) Test (P = 0.05) (SAS 2001). A Student's t-test (P = 0.05) was used to compare the entrance position variable for laboratory room versus CDC net trap, the catch variable for laboratory room versus CDC net trap catch, and the sex variable for the different ages and combinations of attractants.

Figure 2-1. CDC Net Trap used to capture house flies escaping from screen cages.

Figure 2-2. Bottom-entry release cage design fitted with a Trap n' Toss™ cone and open to laboratory room.

Figure 2-3. Top-entry release cage design fitted with modified Trap n' Toss™ and open to laboratory room

Figure 2-4. Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap

Figure 2-5. The Advantage Fly Trap™ (Advantage Traps, Inc., Columbia, SC)

Figure 2-6. BC 1752 Dome (McPhail) Trap (AgrisenceTM Agriculture, Pontypridd, UK)

Figure 2-7. Rescue!® Reusable Fly Trap (Sterling International, Inc., Liberty Lake, WA)

Figure 2-8. Victor Fly Magnet® Trap (Woodstream, Lititz, PA)

Figure 2-9. Fly Terminator® Pro (Farnam Companies, Inc., Phoenix, AZ)

Figure 2-10. Bottom-entry study design with Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap attached.

Figure 2-11. Top-entry study design with Victor Fly Magnet® Trap (Woodstream, Lititz, PA) attached.

CHAPTER 3
RESULTS

Fly Trap Design Experiments

Scatter Experiment

When the house flies were released from the deli cup into the release cage, they exhibited resting behavior preceded by a short time of flying or walking. They did not show any initial observational attraction to the trap, sugar water, or opening of the release cage. However, over half (53.38 ± 4.18%) were eventually captured. The cone allowed house flies to easily leave the release cage, but made it difficult to reenter.

The percentage of house flies both captured in CDC net traps and escaped into the laboratory room after 24 h was determined (Table 3-1). For the released male house flies, there was no significant difference between bottom-entry and top-entry treatments (df = 9.11, t = 2.08, P = 0.0669). There was no significant difference in house fly escape between bottom-entry (82.00 ± 6.63%) and top-entry (90.00 ± 2.58) treatments for the laboratory room. However, significantly more males escaped into the laboratory room than escaped into CDC net traps (df = 9.34, t = 4.39, P = 0.0016). Significantly more males escaped through top-entry (67.00 ± 2.52%) treatments than into the bottom-entry (22.00 ± 3.46%) treatments into CDC net traps (df = 3, F = 22.13, P < 0.0001). Within the bottom-entry treatment, significantly more males escaped into the laboratory room (82.00 ± 6.63%) than were caught in the CDC net trap (22.00 ± 3.46%) (Table 3-1). Significantly more male house flies escaped into the laboratory room (90.00 ± 2.58%) than into the CDC net trap (67.00 ± 2.52%) within the top entry treatment.

For the released female house flies, there was no significant difference between bottom-entry and top-entry treatments (df = 14, t = 0.20, P = 0.8457). Regardless of treatment, significantly more female house flies escaped into the laboratory room than were captured in

CDC net traps (df = 8.15, t = 6.43, P = 0.0002). There was no significant difference in house fly escape between bottom-entry (67.00 ± 11.12%) and top-entry (65.00 ± 11.24%) treatments for the laboratory room escape option. Overall, few female house flies were captured in the CDC net traps (17.00 ± 2.97%) and there was no significant difference between bottom-entry (19.00 ± 2.52%) and top-entry (15.00 ± 3.42%) treatments. Within the bottom-entry treatment, significantly more females escaped into the laboratory room (67.00 ± 11.12%) than were captured in the CDC net trap (19.00 ± 2.52%). Significantly more female house flies also escaped into the laboratory room (65.00 ± 11.24%) than into the CDC net trap (15.00 ± 3.42%) for the top-entry treatment (df = 3, F = 11.50, P = 0.0008).

There was no significant difference between the total numbers of house flies that escaped through top entry and bottom entry treatments (df = 14, t = 0.90, P = 0.3844). Overall, house flies were not caught in CDC net traps as often as they escaped into the laboratory room (df = 14, t = 7.67, P < 0.0001). About three quarters of the released house flies escaped into the laboratory room (76.00 ± 4.04%). Top-entry CDC net traps (41.00 ± 2.89%) caught significantly more house flies than the bottom-entry CDC net traps (20.50 ± 2.87%) (df = 3, F = 27.49, P < 0.0001). There was no significant difference between bottom-entry (74.50 ± 6.99 %) and top-entry (77.50 ± 5.06%) traps. The mean trap catch for the CDC net traps was only one-third (30.75 ± 4.31%) of the total released house flies. However, significantly more house flies escaped through bottom-entry traps into laboratory room (74.50 ± 6.99%) than were captured in the bottom-entry CDC net trap (20.50 ± 2.87%) (df = 3, F = 27.49, P < 0.0001). This pattern was also observed with top-entry traps where significantly more house flies escaped into the laboratory room (77.50 ± 5.06%) than into the CDC net trap (41.00 ± 2.89%) (df = 3, F = 27.49, P < 0.0001).

Commercial Fly Trap Dimensions

Six commercial fly traps were compared for their ability to capture house flies released into screened cages (Table 3-2). Each fly trap consisted of a plastic container that held both the attractant and the captured house flies. All fly traps had funnels that had both entrance and exit holes that facilitated house fly capture but limited escape. Every fly trap, except the Terminator® Pro, had entrance holes that were larger than the exit holes. The Trap n' Toss™, a bottom-entry trap, had the greatest entrance area of all the fly traps (176.24 cm^2), whereas the Fly Terminator® Pro had the largest entrance area for the top-entry fly traps (20.51 cm^2). The smallest entrance area of the top-entry fly traps was the Rescue!® Reusable Fly Trap (3.16 cm^2), whereas the smallest bottom-entry area was The Advantage™ Fly Trap (46.57 cm^2). The smallest exit area of the top-entry fly traps was the Rescue!® Reusable Fly Trap (2.00 cm^2), whereas the bottom-entry fly trap with the smallest exit area was The Advantage Fly Trap™ (9.08 cm^2). The largest exit area of the top-entry fly traps was the Fly Terminator® Pro (20.51 cm^2), whereas the Trap n' Toss™ had the largest exit area (11.95 cm^2) of bottom-entry traps.

The fly trap entrance to exit ratio is a measure of the entrance area divided by the exit area. This is an important measurement because it provides the best relative combination of allowing house flies into the fly trap without releasing them. The bottom-entry Trap n' Toss™ had the greatest entrance:exit ratio of the fly traps (14.75), whereas the Fly Magnet® had the greatest entrance:exit ratio of the top-entry fly traps (2.05). The top-entry Fly Terminator® Pro had the smallest entrance:exit ratio of all the fly traps (1.00), whereas BC 1752 Dome had the smallest entrance:exit ratio of the bottom-entry fly traps (4.64).

The slope of the entrance cone directs the house flies into the holding container and the range of slopes observed by these traps was considerable. The BC 1752 Dome had the greatest slope of the bottom-entry fly traps (75.57^o), whereas the Terminator® Pro, a top-entry fly trap,

had the greatest slope of all the fly traps (90.00^{o}). The Trap n' Toss^{TM}, a bottom-entry fly trap, had the least slope of all the fly traps (61.67^{o}), whereas the Rescue!® had the least slope of the top-entry fly traps (85.07^{o}).

The bottom-entry fly trap with the largest fly holding volume was Trap n' Toss^{TM} (1936 ml), whereas the fly trap with the largest volume was the top-entry Fly Terminator® Pro (3786 ml). The fly trap with the smallest volume was the BC 1752 Dome, whereas the top-entry fly trap with the smallest volume was the Fly Magnet® (983 ml).

Each fly trap's entrance was one of several colors. The Trap n' Toss^{TM} and BC 1752 Dome's entrances were colored yellow. The Advantage Fly Trap^{TM}'s entrance was white. The Fly Magnet® and Fly Terminator® Pro had black entrances, while the Rescue!® had a green entrance.

Commercial Fly Trap Experiment

When house flies were released from deli cups into the release cage, they showed the previously described initial behavior. At the completion of the 24 h experiment, some of the house flies were attracted to or wandered into the fly traps. However, it is likely that some may have escaped back into the release cage. House flies would continue to show this basic behavior throughout the experiments.

The percentage of house flies captured in six different commercial fly traps after 24 h was recorded (Table 3-3). For this experiment, the mean catch of released males was 36.67 ± 3.17% (n = 42). The highest capture was with the The Advantage Fly Trap^{TM} (51.43 ± 7.33%), while the lowest catch was with the Fly Terminator® Pro (22.00 ± 13.71%). These were the only significantly different catch means with respect to released male house flies (df = 5, F = 2.87, P = 0.0303). There was no significantly superior bottom-entry fly trap when compared only to the other bottom-entry traps or top-entry fly trap when compared only to other top-entry traps.

The mean catch of released females for this experiment was 41.90 ± 3.56% (n = 42). The Trap n' Toss™ caught significantly more released females (66.22 ± 5.89%) than the Advantage (35.43 ± 6.32%), BC 1752 Dome (34.86 ± 6.97%), Fly Magnet® (32.80 ± 7.31%), and Fly Terminator® Pro (28.00 ± 5.42%) (df = 5, F = 4.58, P = 0.0031). Rescue!® (58.40 ± 12.24%) was not significantly different than the best or the worst fly traps when catching female house flies. There was no significant difference between the top-entry fly traps; however, the Trap n' Toss™ caught significantly more house flies than the other two bottom-entry fly traps (df = 5, F = 4.58, P = 0.0031).

The mean capture for all fly traps in the experiment was 39.33 ± 2.58% of the total released house flies (n = 42). The fly trap that captured the most house flies was the Trap n' Toss™ (52.22 ± 5.13%), while the Fly Terminator® Pro captured the fewest (25.00 ± 8.10%). These were the only significantly different catch means with respect to total house flies released (df = 5, F = 3.46, P = 0.0134). There were no significantly superior fly traps when comparing only top-entry fly traps with each other and bottom-entry fly traps with each other.

Attractant Experiments

Aged Attractant Experiment

The percentage of house flies after captured after 24 h in fly traps with a yeast and ammonium carbonate mixture aged 0 - 4 days (by one day intervals) was determined (Table 3-4). A significant difference between aged attractants was observed with male house flies (df = 4, F = 3.44, P = 0.0233). Attractants aged for four days (62.00 ± 5.54%) and three days (60.67 ± 7.04%) caught significantly more house flies than 1-day aged attractant (35.20 ± 5.43%). The freshly mixed attractant (38.22 ± 3.49%) and 2-day aged attractant (42.00 ± 7.85%) were not significantly different from the mixtures that captured the most and least house flies. Regression

analysis demonstrated that as attractant aged from one to four days, male house fly catch increased linearly ($y = 9.907x + 25.2$, $R^2 = 0.9073$) (Fig. 3-1).

As with males, a significant difference was also observed between female house fly capture among the attractants ($df = 4$, $F = 3.63$, $P = 0.0188$). The 4-day aged attractant (78.00 ± 5.91%) caught significantly more females than attractant aged 1 day (47.20 ± 4.80%). Significant differences were not observed among other treatments. Regression analysis demonstrated that as attractant aged from 1 to 4 days, female house fly catch increased linearly ($y = 11.173x + 31.2$, $R^2 = 0.8801$) (Fig. 3-1).

Overall, there was a significant difference between the numbers of house flies captured using variously aged attractants ($df = 4$, $F = 4.62$, $P = 0.0065$). The 4 day aged attractant (70.00 ± 4.73%) captured significantly more house flies than attractants aged two days (49.00 ± 6.65%) and one day (41.20 ± 4.08%). Attractant aged three days (63.00 ± 5.99%) caught significantly more house flies than attractant aged one day (41.20 ± 4.08%). Freshly mixed attractant (55.33 ± 3.13%) was not significantly different from any of the other mixtures. Regression analysis demonstrated that as attractant aged from 1 to 4 days, total house fly catch increased linearly ($y = 10.04x + 30.7$, $R^2 = 0.9827$) (Fig. 3-1).

Attractant Component Experiment

To determine the importance of attractant age and attractant components on house fly capture, a study was conducted examining individual and combinations of components at 0, 2, and 4 days post-mixing (Table 3-5). The freshly mixed combination treatment (56.00 ± 7.93%) captured significantly more male house flies than the ammonium carbonate (20.00 ± 6.20%) and the control treatments (9.33 ± 3.04%) ($df = 3$, $F = 9.31$, $P = 0.0005$). For attractants aged both 2 and 4 days, the combination and yeast treatments captured significantly more house flies than the

ammonium carbonate and control treatments, respectively (df = 3, F = 10.94, P = 0.0002; df = 3, F = 8.45, P = 0.0008).

The freshly mixed and 4-day-old attractant, combination and yeast treatments captured significantly more female house flies than the ammonium carbonate and control treatments, respectively (df = 3, F = 12.97, P < 0.0001; df = 3, F = 16.98, P < 0.0001). For attractants aged 2 days, the combination treatment (69.33 ± 8.56%) caught significantly more house flies than the yeast treatment (40.67 ± 8.48%), which caught significantly more house flies than ammonium carbonate (18.67 ± 6.25%) and the control treatments (12.67 ± 7.26%) (df = 3, F = 10.21, P = 0.0003).

The freshly mixed combination treatment (61.00 ± 5.48%) captured significantly more total house flies than the yeast treatment (41.67 ± 5.55%), which captured significantly more house flies than the ammonium carbonate (22.00 ± 6.85%) and the control treatments (9.67 ± 3.32%). With both 2-day and 4-day aged attractants, the combination and yeast treatments captured significantly more house flies than the ammonium carbonate and control treatments, respectively (df = 3, F = 11.77, P = 0.0001; df = 3, 24.27, P < 0.0001).

The combination (df = 10, *t* = 2.91, P = 0.0155) and yeast (df = 10, *t* = 2.56, P = 0.0283) treatments captured significantly more female house flies than males when aged for 4 days. All other combinations of attractant components and ages did not show significant differences between the sexes.

Table 3-1. Percent escape of male, female, and total house flies into CDC net traps or laboratory room.

Trap[b]	n[c]	% (SE)[a] Males	Females	Total
Bottom Entry				
Room	4	82.00 (6.63)a	67.00 (11.12)a	74.50 (6.99)a
Net	4	22.00 (3.46)c	19.00 (2.52)b	20.50 (2.87)c
Top Entry				
Room	4	90.00 (2.58)a	65.00 (11.24)a	77.50 (5.06)a
Net	4	67.00 (2.52)b	15.00 (3.42)b	41.00 (2.89)b
		df = 3, F = 22.13, P < 0.0001	df = 3, F = 11.50, P = 0.0008	df = 3, F = 27.49, P < 0.0001

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different (α = 0.05, Student Newman-Keuls test; SAS 2001).

[a] 25 male and 25 female, 3-5 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Fly trap removed from release cage following 24 h exposure.

[b] Room = house flies allowed to escape into laboratory room. Net = house flies captured in CDC net trap affixed to cone opening.

[c] n = replications

Table 3-2. Characteristics of six commercially available fly traps used to evaluate house fly capture efficacy.

Trap[a]	Entrance (cm^2)	Exit (cm^2)	Ratio[b]	Slope (deg)	Volume (ml)[c]	Color
Trap n' Toss™	176.24	11.95	14.75	61.67	1936	yellow
Advantage™	46.57	9.08	5.13	75.33	1840	white
BC 1752 Dome	49.00	10.56	4.64	75.57	748	yellow
Rescue!®	3.16	2.00	1.58	85.07	1060	green
Fly Magnet®	6.16	3.00	2.05	89.97	983	black
Terminator® Pro	20.51	20.51	1.00	90.00	3786	black

[a] Bottom-entry traps were Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ), The Advantage Fly Trap™ (Advantage Traps, Inc., Columbia, SC), and BC 1752 Dome McPhail) Trap (Agrisence™ Agriculture, Pontypridd, UK), and were placed on top of the release cage. Top-entry traps were Rescue!® Reusable Fly Trap (Sterling International, Inc., Liberty Lake, WA), Victor Fly Magnet® Trap (Woodstream, Lititz, PA), and Fly Terminator® Pro (Farnam Companies, Inc., Phoenix, AZ), and were placed under the release cage.
[b] Ratio = entrance ÷ exit
[c] Volume = number of ml holding capacity of fly trap

Table 3-3. Percentage of male, female, and total house flies caught in six commercial traps.

		% Catch (SE) [a]		
Trap[b]	n	Males	Females	Total
Trap n' Toss™	9	38.22 (5.21)ab	66.22 (5.89)a	52.22 (5.13)a
Advantage™	7	51.43 (7.33)a	35.43 (6.32)b	43.71 (4.40)ab
BC 1752 Dome	7	22.86 (4.07)ab	34.86 (6.97)b	28.86 (5.03)ab
Rescue!®	5	36.00 (9.38)ab	58.40 (12.24)ab	47.20 (9.73)ab
Fly Magnet®	5	24.80 (5.57)ab	32.80 (7.31)b	28.80 (6.09)ab
Terminator® Pro	4	22.00 (13.71)b	28.00 (5.42)b	25.00 (8.10)b
		df = 5, F = 2.87, P = 0.0303	df = 5, F = 4.58, P = 0.0031	df = 5, F = 3.46, P = 0.0134

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different ($\alpha = 0.05$, Student Newman-Keuls test; SAS 2001).

[a] 25 male and 25 female, 3-7 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Fly trap removed from release cage following 24 h exposure. All traps contained a combination of yeast and ammonium carbonate (41.67 : 1) in 75 ml of water.

[b] Bottom-entry traps were Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ), The Advantage Fly Trap™ (Advantage Traps, nc., Columbia, SC), and BC 1752 Dome McPhail) Trap (Agrisence™ Agriculture, Pontypridd, UK), and were placed on top of the release cage. Top-entry traps were Rescue!® Reusable Fly Trap (Sterling International, Inc., Liberty Lake, WA), Victor Fly Magnet® Trap (Woodstream, Lititz, PA), and Fly Terminator® Pro (Farnam Companies, Inc., Phoenix, AZ), and were placed under the release cage

Table 3-4. Percentage of male, female, and total house flies captured after 24 h in Trap n' Toss™ fly traps baited with an attractant aged up to 4 d.

		% Catch (SE)[a]		
Attractant age (d)[b]	n[c]	Males	Females	Total
0	6	38.22 (3.49)ab	59.33 (4.78)ab	55.33 (3.13)abc
1	5	35.20 (5.43)b	47.20 (4.80)b	41.20 (4.08)c
2	6	42.00 (7.85)ab	56.00 (7.00)ab	49.00 (6.65)bc
3	6	60.67 (7.04)a	65.33 (7.06)ab	63.00 (5.99)ab
4	6	62.00 (5.54)a	78.00 (5.91)a	70.00 (4.73)a
		df = 4, F = 3.44, P = 0.0233	df = 4, F = 3.63, P = 0.0188	df = 4, F = 4.62, P = 0.0065

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different ($\alpha = 0.05$, Student Newman-Keuls test; SAS 2001).

[a] 25 male and 25 female, 3-7 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap removed from release cage following 24 h exposure.

[b] Combination of yeast and ammonium carbonate (41.67 : 1) in 75 ml of water

[c] Replications

Table 3-5. Percentage of house flies caught in baited Trap n' Toss™ fly traps with attractants aged up to 4 d.

	Male % Catch (SE) [a]		
	Days		
Attractant[b]	0	2	4
Control	9.33 (3.04)c	7.33 (2.40)b	10.67 (3.82)b
Yeast	34.00 (6.26)ab	45.33 (6.98)a	37.33 (5.81)a
Ammonium Carbonate	20.00 (6.20)bc	16.67 (5.97)b	17.33 (4.92)b
Combination	56.00 (7.93)a	46.00 (7.14)a	44.00 (7.08)a
	df = 3, F = 9.31, P = 0.0005	df = 3, F = 10.94, P = 0.0002	df = 3, F = 8.45, P = 0.0008

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different ($\alpha = 0.05$, Student Newman-Keuls test; SAS 2001). n = 6

[a] 25 male and 25 female, 3-5 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap removed from release cage following 24 h exposure.

[b] Control = 75 ml tap water, yeast = dried active baker's yeast in 75 ml of tap water, ammonium carbonate = ammonium carbonate in 75 ml of tap water, combination = yeast and ammonium carbonate (41.67 : 1) in 75 ml of tap water

Table 3-5 Continued.

Female % Catch (SE) [a]			
	Days		
Attractant[b]	0	2	4
Control	10.00 (4.70)b	12.67 (7.26)c	15.33 (3.78)b
Yeast	49.33 (6.67)a	40.67 (8.48)b	61.33 (7.35)a
Ammonium Carbonate	24.00 (8.20)b	18.67 (6.25)c	24.67 (5.79)b
Combination	66.00 (5.03)a	69.33 (8.56)a	77.33 (8.98)a
	df = 3, F = 12.97, P < 0.0001	df = 3, F = 10.21, P = 0.0003	df = 3, F = 16.98, P < 0.0001

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different ($\alpha = 0.05$, Student Newman-Keuls test; SAS 2001). n = 6

[a] 25 male and 25 female, 3-5 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap removed from release cage following 24 h exposure.

[b] Control = 75 ml tap water, yeast = dried active baker's yeast in 75 ml of tap water, ammonium carbonate = ammonium carbonate in 75 ml of tap water, combination = yeast and ammonium carbonate (41.67 : 1) in 75 ml of tap water

Table 3-5 Continued

	Total % Catch (SE)[a]		
	Days		
Attractant[b]	0	2	4
Control	9.67 (3.32)b	10.00 (4.41)b	13.00 (3.09)b
Yeast	41.67 (5.55)a	43.00 (7.30)a	49.33 (5.77)a
Ammonium Carbonate	22.00 (6.85)b	17.67 (5.85)b	17.67 (3.56)b
Combination	61.00 (5.48)a	57.67 (6.52)a	60.67 (5.90)a
	df = 3, F = 13.15, P < 0.0001	df = 3, F = 11.77, P = 0.0001	df = 3, F = 24.27, P < 0.0001

Data were arcsine square root transformed before analysis. Means within a column followed by the same letter are not significantly different ($\alpha = 0.05$, Student Newman-Keuls test; SAS 2001). n = 6

[a] 25 male and 25 female, 3-5 d post-eclosion, house flies released into 28.8 by 26.1 by 39.1 high cm release cage. Trap n' Toss™ (Farnam Companies, Inc., Phoenix, AZ) fly trap removed from release cage following 24 h exposure.

[b] Control = 75 ml tap water, yeast = dried active baker's yeast in 75 ml of tap water, ammonium carbonate = ammonium carbonate in 75 ml of tap water, combination = yeast and ammonium carbonate (41.67 : 1) in 75 ml of tap water

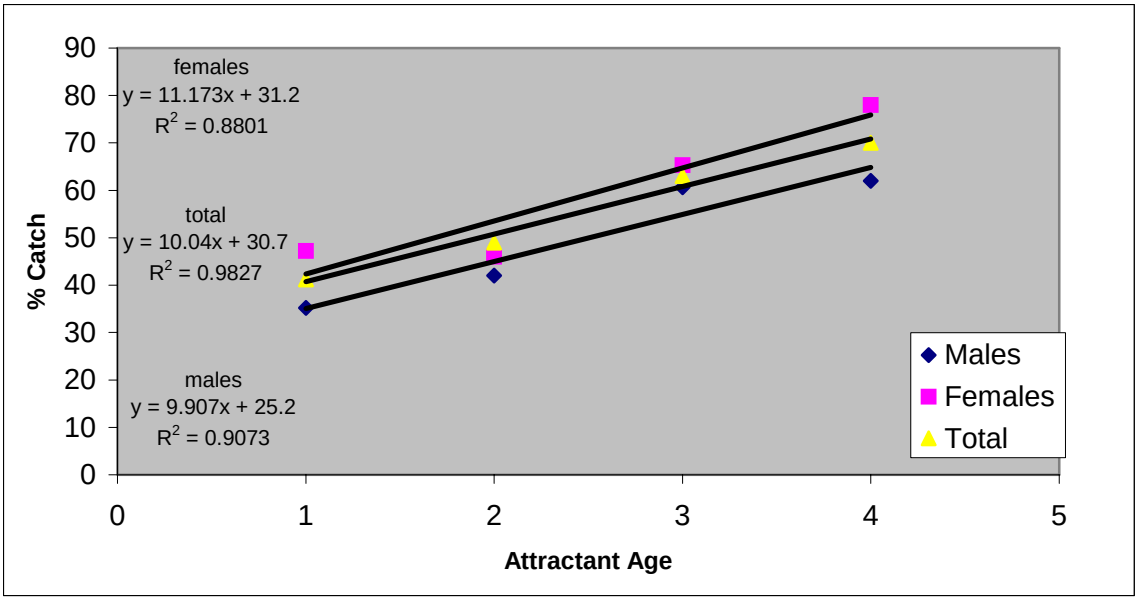

Figure 3-1. Effect of attractant age on percent catch of male, female, and total house flies.

CHAPTER 4
DISCUSSION AND CONCLUSIONS

Fly Trap Design Experiments

House flies tend to scatter from sites where they are released (Hindle 1914, Jones et al. 1999). Sacca (1963) concluded that house flies disperse in a more or less random pattern until suitable conditions are found. However, Bishopp and Laake (1919) found that house flies would pass up favorable feeding and breeding places in the course of dispersal. In my experiments, the house flies released into a cage scattered from the cage into the laboratory room and various fly traps. This scattering behavior was in accordance with the past literature. The house flies scattered from the release cage, which contained nutrition, water, and potential mates. Sacca's (1963) observation that house flies seem to move from one place to another in a random fashion was true of the observations in my experiments. However, Sacca (1963) stated that these random movement episodes end when suitable conditions are found i.e. food and mates, but the behavior of the house flies in my experiments to forgo suitable conditions contradicted this. Instead the house flies in my experiments behaved more in accordance with the findings of Bishopp and Laake (1919). This behavior may account for why more house flies escaped from the release cage into the laboratory room than the CDC net trap and the un-baited Trap n' Toss™, because there is a small chance that the house flies would move back towards the opening of the release cage and go back in, but as the house flies scatter into one of the fly traps, they can just as well scatter back into the release cage.

However, despite the seemingly random scatter from the release cages, air movement may also affect the house fly dispersal behavior. Several authors have found house flies to be positively anemotropic (Hindle 1914, Hindle and Merriman 1914, Ball 1918, Carment 1922, Uvarov 1931). In my experiments, more house flies scattered into the laboratory room than

ended up in the CDC net trap, and more were in the CDC net trap than in the un-baited Trap n' Toss™. There is a direct correlation with allowed air movement in this situation. The bottom-entry CDC net trap versus the top-entry CDC net trap difference in house fly catch suggest the position of the trap was very important to return dispersal. For each treatment, there was the same entrance/exit cone from the Trap n' Toss™, but the Trap n' Toss™ trap allowed less air flow than did the CDC net trap, and the CDC net trap allowed less air flow than did the open laboratory room. The behavior of the house flies in my experiments was in accordance with previous authors' findings, that house flies will seek and fly upwind.

Most of the commercial fly traps were not significantly different in their abilities to capture house flies. However, the Trap n' Toss™ caught significantly more house flies than the Terminator® Pro, and this was probably due to a number of design factors including large entrance hole area, small exit hole area, the ratio of entrance to exit hole ratio, the slope being close to 60^{o}, and the color of the entrance. This is not surprising when considering the overall design of each fly trap. Pickens (1995) stated that the larger the entrance hole, the more house flies that would be captured with a fly trap. His optimum was an entrance of 506.45 cm^2. I found the Trap n' Toss™ to be only ~35% of that area, but the Fly Terminator® Pro was a mere 4% of Pickens' (1995) ideal entrance area. So, it can be said that a large entrance may be partially responsible for the Trap n' Toss™ having a higher percent catch of the house flies than Terminator® Pro. Pickens (1995) also states that a small exit will work best to keep captured house flies in the fly trap. His ideal exit area is 0.28 to 1.33 cm^2. I found the Trap n' Toss™ to be almost nine times larger than 1.33 cm^2, but Fly Terminator® Pro is over 15 times larger. The smaller exit area of the Trap n' Toss™ may have contributed to its higher house fly capture through retention of captured house flies. If a large entrance and a small exit are best for

capturing house flies, then, within reason, the higher the ratio of entrance to exit, the more house flies a fly trap should catch. Again the Trap n' Toss™ ratio was almost 15 times larger than that of the Terminator® Pro. The slope may have been a factor in the Trap n' Toss™ capturing more house flies than Terminator® Pro. According to Pickens (1995) the slope of the cone of the fly trap should be 60°. The Trap n' Toss™ had a slope barely above 60°, while Fly Terminator® Pro was 90°.

There have been many studies documenting which color makes a surface most attractive to house flies, and there have been varied conclusions. Awati and Swaminath (1920) used colored paper and found yellow to be most attractive. Burg and Axtell (1984) painted plastic jug traps and found yellow to be the most attractive to house flies. Pickens (1995) stated that white or yellow were preferred in cool weather, while red, black and blue were the preferred color surfaces when the weather was hot, while Muniz (1967) found that color preference was independent of temperature. Hecht et al. (1968) using colored pieces of cardboard found that black was most attractive in the laboratory, while yellow was most attractive outdoors. Nava (1967) found that house flies were attracted to black cardboard more than any other color. The studies of Awati and Swaminath (1920), Burg and Axtell (1984), and to some extent Hecht et al. (1968) and Pickens (1995) substantiate that the yellow color may have played a role in the yellow entrance of Trap n' Toss™ capturing more house flies than the black entrance of Fly Terminator® Pro.

The commercial fly traps had many different characteristics, but only two fly traps were significantly different from each other. The Trap n' Toss™ entrance area was almost nine times larger and the exit area was about half as large as the Fly Terminator® Pro. The ratio then of entrance area to exit area for the Trap n' Toss™ was almost 15 times more than Terminator®

Pro. The slope of the entrance of Trap n' Toss™ was nearly the ideal 60°, while Fly Terminator® Pro was ~30° steeper. The color of the Trap n' Toss™ entrance was yellow, whereas the Fly Terminator® Pro entrance was black. Although there was no one apparent overriding characteristic that allowed the Trap n' Toss™ to capture more house flies than the Terminator® Pro, these studies document the variety of issues impacting house fly trapping.

Attractant Experiments

Two commonly used materials in commercial fly trap attractants are yeast and ammonium carbonate. Pickens (1995) devised a categorizing system for the different odor attractants for house flies. They are sugars and fermentation products, proteins, and animal excretions. Yeast is in the sugar and fermentation products category, while ammonium carbonate is in both the sugar and fermentation products as well as animal excretions category (Pickens 1995).

Cohen et al. (1991) used fly traps baited with yeast around a military camp and reported a 64% decrease in house fly density that accounted for a 42% decrease in clinic visits by the soldiers for diarrhea and an 85% reduction in Shigellosis visits. Chapman et al. (1998) found that yeast provided slightly increased house fly attraction over the control, while adding yeast to (Z)-9-tricosene-impregnated targets produced a significant reduction in the number of male house flies attracted. In my studies, I found yeast to be more attractive than the water control at 0, 2, and 4 days old by ~30-35%. My studies do agree with both authors that yeast attracts more house flies than a control, but my studies put yeast's attractiveness somewhere between the attractancy observed by either Cohen et al. (1991) or Chapman et al. (1998).

Ammonium salts have been shown to be attractive to various filth flies most likely due to the salts' release of ammonia. Ammonium carbonate is an ammonium salt and releases both ammonium carbonate and carbon dioxide (Loeser et al. 2004). Mulla et al. (1977) lists ammonium chloride and ammonium sulfate, as highly attractive to house flies. Wieting and

Hoskins (1939) found ammonia attractive and carbon dioxide not attractive to house flies. Hobson (1935, 1936) found ammonium carbonate in conjunction with indole was attractive to *Lucilia sericata* (Meigen). Mulla and Ridsdill-Smith (1986) found that when ammonium carbonate was added to an attractant mixture, attractancy was not significantly increased for *Musca vetustissima*. My studies demonstrated that ammonium carbonate was never more attractive than the control, and when added to yeast (an attractant) did not increase attractancy. The results that I obtained were never directly comparable with the aforementioned studies due to the difference in attractant components used and/or species tested against. However, my studies agree most with Mulla and Ridsdill-Smith's (1986) experiments with *M. vetustissima*, in that ammonium carbonate does not represent an attractant chemical for the house fly when used alone or in conjunction with other attractants.

Initially, there was reason to believe that a combination of yeast and ammonium carbonate would be more attractive than the yeast or ammonium carbonate alone. Cosse and Baker (1996) used Eletctoantennagram (EAG) to determine a number of chemicals that illicit a response in house flies, but many required combination with other attractants to stimulate a behavioral response i.e. attraction. Geden (2005) combined two commercial attractant mixtures and reported synergistic effects. When yeast and ammonia carbonate were mixed, the combination did not catch significantly more house flies than the yeast alone, but did catch more than ammonium carbonate alone. There was certainly no synergistic effect. Because the assumed attractive ammonium carbonate turned out to be not attractive, my results do not agree or disagree with Cosse and Baker (1996) or Geden (2005).

Given the nature of the presumed attractant combination used (yeast and ammonium carbonate components), it is logical to assume that the attractant combination would change over

time. Ammonium carbonate quickly released ammonia and carbon dioxide when added to water. The yeast was a living organism and its life cycle was subject to availability of nutrition. In my experiment, there was not an ample carbon supply for the yeast i.e. sugar, meaning the yeast either arrested in G0 phase, progressed slowly through the life cycle, or died (Dickinson and Schwiezer 1999). How the yeast changed is unknown, but it was changing. Pickens et al. (1973) stated that baits of fermented grains and yeasts took about three to four days to become attractive. Geden (2005) observed commercial attractants to become less attractive by the fourth day. The attractant combination in my study increased linearly from day 1 to 4 and agrees more with Pickens et al. (1973), most likely due to the similarity of fermentation product attractants that we each used. The components of the attractant mixtures used by Geden (2005) were not reported; therefore it is difficult to discuss the similarities and differences. Also the experiments of Geden (2005) were run outdoors at temperatures as high as 37^{o} C, which may have affected volatility or stability of the attractants, while my attractants were aged in less extreme temperatures indoors (22.8 ± 0.02^{o} C) which may have kept the yeast viable.

Attractants increase the effectiveness of fly traps considerably. In fact, Kuzina (1940) stated that odor was the most important mechanism used by house flies to locate food sources. In my studies, yeast proved to be much better at attracting house flies than ammonium carbonate, and ammonium carbonate never attracted significant numbers of house flies. The fact that ammonium carbonate was not attractive, resulted in the combination of yeast and ammonium carbonate being no better at attracting flies than the yeast alone. When using a live organism such as yeast in order to attract house flies, as long as it is sustained, the attraction will increase linearly for at least 4 days.

Conclusions

House flies are important pests that are best controlled by using IPM practices. This is particularly true given the house fly's ability to develop insecticide resistance and its considerable population potential (Hodge 1911, Scott et al. 2000, Learmount et al. 2002, Gao and Scott 2006). Commercial fly traps have two potential niches in a house fly IPM program, surveillance and control.

Field trials must still be run in order to obtain a better understanding on how well the tested commercial fly traps perform in non-laboratory conditions. However my laboratory studies demonstrated that fly traps can be viable devices for catching house flies. Although the Fly Terminator® Pro with an un-aged attractant mixture of yeast and ammonium carbonate only captured ~25% of the total house flies, the Trap n' Toss™ with the same attractant mixture aged 4 days, caught ~70% of the total house flies, and almost 80% of the female house flies. A capture of ~70% rivals the effectiveness of some biological control agents and insecticides. Aside from comparing with other control measures, ~70% capture exceeds Dadour and Cook's (1992) requirement that in order to reduce a house fly population by 50-90%, 24-58% of the present house flies should be captured. According to my laboratory studies, the proper design (e.g. at least a 176.24cm^2 entrance area, at most an 11.95 cm^2 exit area, a ratio of at least 14.75, a slope around 60^o, and a yellow entrance) and an aged attractant can capture high numbers of house flies and should be considered for use as a control measure in an IPM program.

In addition to controlling fly populations, commercial fly traps may serve as monitoring tools in a house fly IPM program. Burg and Axtell (1984) suggested that fly traps represented a simple, practical way to monitor house fly populations, thus giving a better idea of when to use additional control measures. Therefore, the commercial fly traps that did not catch the most house flies in these studies could still be important tools in house fly IPM.

In conclusion, my studies have shown that commercial fly traps have potential as house fly IPM tools. They are versatile devices to be used in IPM programs, and can be used either as control devices or monitoring house fly populations. They are compatible with both insecticides and biological control measures. Also, if there exists a situation in which insecticides or biological controls cannot be used, they are safe, easy, and effective enough to be used as stand-alone devices. According to my laboratory studies, and with the further exploration with field studies, commercial fly traps represent a viable tool for managing house flies in a wide variety of settings including agriculture, urban, and military

LIST OF REFERENCES

Adams, T. S., and G. G. Holt. 1987. Effect of pheromone components when applied to different models on male sexual behavior in the housefly *Musca domestica*. Journal of Insect Physiology 33:9-18.

Appling, J. 1988. Toxicology on tap: Uncertain health significance of contaminated; drinking water. New York's Food & Life Sciences Quarterly 18:24-26.

Awati, P. R., and C. S. Swaminath. 1920. Bionomics of houseflies. III. A preliminary note on attraction of houseflies to certain fermenting and putrefying substances. Indian Journal of Medical Research 7:560-567.

Axtell, R. 1970a. Fly control in caged-poultry houses: Comparison of larviciding and integrated control programs. Journal of Economic Entomology 63:1734-1737.

Axtell, R. 1970b. Integrated fly-control program for caged-poultry houses. Journal of Economic Entomology 63:400-405.

Ball, S. 1918. Migrations of insects to Rebecca Shoal Light-Station and the Tortugas Islands, with special reference to mosquitoes and flies. Papers of the Department of Marine Biology Carnegie Institute Washington 12:193-212.

Basden, E. B. 1947. Breeding the house-fly (*Musca domestica* L.) in the laboratory. I. Introduction and technique. Bulletin of Entomological Research 37:381-387.

Bishopp, F., and L. Henderson. 1946. Housefly control. Usda Leaflet 182.

Bishopp, F., and E. Laake. 1919. The dispersion of flies by flight. Journal of Economic Entomology. 12: 210-211.

Bishopp, F., and E. Laake. 1921. Dispersion of flies by flight. Journal of Agricultural Research 21:729-766.

Bishopp, F. C., W. E. Dove, and D. C. Parman. 1915. Notes on certain points of economic importance in the biology of the house fly. Journal of Economic Entomology 8:54-71.

Blair, D. M. 1945. Baited fly traps. Rhodesia Agricultural Journal 42:488-492.

Bloomquist, G. J., T. S. Adams, and J. W. Dillwith. 1984. Induction of female sex pheromone production in male houseflies by ovary implants or 20-hydroxyecdysone. Journal of Insect Physiology 30:295-302.

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

Callahan, R. F. 1962. Effects of parental age on life cycle of the house fly, *Musca domestica* Linnaeus (Diptera: Muscidae). Journal of the New York Entomological Society 70:150-158.

Campbell, J. 1993. The economics of the fly problem, Pages 34-39 *in* G. S. Thomas, SR, ed. Proceedings of a symposium: Rural flies in the urban environment. Lincoln, NE, University of Nebraska-Lincoln.

Carlson, D. A., M. S. Mayer, D. L. Silhacek, L. D. James, M. Beroza, and B. A. Bierl. 1971. Sex attractant pheromone of the house fly: Isolation, identification, and sythesis. Science 174:76-77.

Carment, A. 1922. Report on experiment of fly breeding from stable manure with a short account of the finding of a parasite. Agricultural Circulation Department Agriculture of Fiji 3:1-5.

Carson, R. 1962, Silent spring. Boston, MA, Houghton Mifflin.

Chang, S. C. 1965. Chemosterilization and mating behavior of male house flies. Journal of Economic Entomology 58.

Chapman, J., J. Knapp, P. Howse, and D. Goulson. 1998. An evaluation of (Z)-9-tricosene and food odours for attracting house flies, *Musca domestica*, to baited targets in deep-pit poultry units. Entomologia Experimentalis et Applicata 89:183-192.

Chapman, J. W. 1998. Evaluation of three (Z)-9-tricosene formulations for control of *Musca domestica* (Diptera: Muscidae) in caged-layer poultry units. Journal of Economic Entomology 91:915-922.

Chapman, R. F. 1998, The insects: Structure and function. Cambridge, Cambridge University Press.

Clymer, B. 1973. Control of flies around feedlots. Great plains cattle feeding handbook. College Station, TX, Texas A&M University.

Cohen, D., M. Green, C. Block, R. Slepon, R. Ambar, S. Wasserman, and M. Levine. 1991. Reduction of transmission of shigellosis by control of houseflies (*Musca domestica*). Lancet 337:416-420.

Cosse, A.A., and T.C. Baker. 1996. House flies and pig manure volatiles: Wind tunnel behavioral studies and electrophysiological evaluations. Journal of Agricultural Entomology 13:301-317.

Dadour, I., and D. Cook. 1992. The effectiveness of four commercial fly traps at catching insects. Journal of the Australian Entomological Society 31:205-208.

Das, B. 1994. Field trial with redtop flycatcher baited with ready made protein meal for the control of houseflies in Gazipur Dairy Farm, Delhi. Journal of Communicable Disease 26:19-25.

Dickinson, J. R., and M. Schweizer. 1999, The metabolism and molecular physiology of *Saccharomyces cerevisiae*. Philadelphia, PA, Taylor & Francis Inc.

Driggers, D. 1971. Field evaluation of blacklight electrocutor grid traps for the control of flies associated with poultry. M.S. thesis, University of Florida, Gainesville, FL.

Dunn, L. H. 1923. Observations on the oviposition of the house fly, *Musca domestica* in Panama. Bulletin of Entomological Research 13:301-305.

Fenton, F., and G. Bieberdorf. 1936. Fly control on A & M farms, Stillwater, OK. Journal of Economic Entomology 29:1003-1008.

Gao, J., and J. Scott. 2006. Role of the transcriptional repressor mdGfi-1 in CYP6D1v1-mediated insecticide resistance in the house fly, *Musca domestica*. Insect Biochemistry and Molecular Biology 36:387-395.

Geden, C. 1997. Evaluation of *Paraiotonchium muscadomesticae* (Nematoda: Tylenchida: Iotonchiidae), a potential biological control agent of the housefly (Diptera: Muscidae). Biological Control 10:42-47.

Geden, C. 2005. Methods of monitoring outdoor populations of house flies, *Musca domestica* L. (Diptera: Muscidae). Journal of Vector Ecology 30.

Geden, C., and R. Axtell. 1988. Predation by *Carcinops pumilio* (Coleoptera: Histeridae) and *Machrocheles muscaedomesticae* (Acarina: Macrochelidae) on the house fly (Diptera: Muscidae): Functional response, effects of temperature, and availability of alternative prey. Environmental Entomology 17:739-744.

Geden, C., and J. Hogsette. 2006. Suppression of house flies (Diptera: Muscidae) in Florida poultry houses by sustained releases of *Muscidifurax raptorellus* and *Spalangia cameroni*. Environmental Entomology 35:75-82.

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

Geden, C., R. Stinner, and R. Axtell. 1988. Predation by predators of the house fly in poultry manure: Effects of predator density, feeding history, interspecific interference, and field conditions. Environmental Entomology 17:320-329.

Geden, C., and J. Stoffolano Jr. 1987. Succession of manure arthropods at a poultry farm in Massachusetts, USA, with observations on *Carcinops pumilio* (Coleoptera: Histeridae) sex ratios, ovarian condition, and body size. Journal of Entomological Science 23:136-148.

Gilbert, D. 1984. Insect electrocutor light traps *in* F. Baur, ed. Insect Management for Food Storage and Processing. St. Paul, MN, American Association of Cereal Chemists.

Gojmerac, W. 1979. Sanitation still is first fly control defense. Hoard's Dairyman 124:789, 825.

Golfcheskie, B. and G. Surgeoner. 1990. Muscovy ducks as an adjunct for the control of the house fly (Diptera: Muscidae). Journal of Economic Entomology 83:788-791.

Goudy, C. 1973, Flies and disease. Biology and disease transmission., v. 2. Princeton, NJ, Princeton University Press.

Graham-Smith, G. 1914. Flies in relation to disease: Nonbloodsucking flies. Cambridge, England.

Greenberg, B. 1971. Flies and disease, volume 1. Ecology, classification and biotic association. Princeton, NJ, Princeton University.

Greenberg, B. 1973. Flies and disease, volume 2. Biology and disease transmission. Princeton, NJ, Princeton University Press.

Greene, A., and N. Breisch. 2002. Optimizing IPM for public buildings. Pest Control Technology 30:32, 34, 36, 38, 40, 42.

Griffith, A. 1908. The life history of house flies. Public Health 21:122-127.

Grubel, P., J. Hoffman, F. Chong, N. Burstein, C. Mepani, and D. Cave. 1997. Vector potential of houseflies (*Musca domestica*) for *Heliobacter pylori*. Journal of Clinical Microbiology 35:1300-1303.

Hampton, U. M. 1952. Reproduction in house fly (*Musca domestica* L.). Proceedings of the Royal Entomological Society of London (A) 27:29-32.

Hanley, M., D. Dunn, S. Abolins, and D. Goulson. 2004. Evaluation of (Z)-9-tricosene baited targets for control of the housefly (*Musca domestica*) in outdoor situations. Journal of Applied Entomology 128:478-482.

Hardy, G. H. 1944. The copulation and the terminal segments of diptera. Proceedings of the Royal Entomoligical Society of London (A) 19:52-65.

Havron, A., and J. Margalit. 1991. Pupal parasitoids (Hymenoptera: Pteromalidae) of muscoid filth flies in Israel. Medical and Veterinary Entomology 5:267-275.

Hecht, O., R. Muniz, and A. Nava. 1968. Contrary responses of *Musca domestica* L. concerning their selection of different shades and hues. Entomologia Experimentalis et Applicata 11:1-14.

Hedges, S. 1993. Controlling flying insects. Pest Control Technology 22:40-44.

Hedges, S. 1994, Field guide for choosing the management of structure-infesting flies. Cleveland, OH, Franzak and Foster.

Herms, W. B. 1928. The effect of different quantities of food during the larval period on the sex ratio and size of *Lucilia sericata* Meigen and *Theobaldia incidens* Thom. Journal of Economic Entomology 21:720-729.

Herms, W. B., and M. T. James. 1961, Medical entomology. New York, Macmillan.

Hewitt, C. G. 1914, The housefly *Musca domestica* Linn. Its structure, habits, development, relation to disease and control. Cambridge, MA, Cambridge University Press.

Hindle, E. 1914. The flight of the house fly. Proceedings of the Cambridge Philosophical Society 17:310-313.

Hindle, E., and G. Merriman. 1914. The range of flight of *Musca domestica*. Journal of Hygiene 14:23-45.

Hobson, R. 1935. Sheep blowfly investigations. II. Substances which induce *Lucilia sericata* (Mg.) to oviposit on sheep. Annals of Applied Biology 22:294-300.

Hobson, R. 1936. Sheep blowfly investigations. III. Observation on the chemotropism of *Lucilia sericata* (Mg.). Annals of Applied Biology 23:846-851.

Hodge, C. F. 1911. How can you make your home, town or city flyless. Nature and Culture 3:9-23.

Hogsette, J.A 1995. The house fly: Basic biology and ecology. USDA Medical and Veterinary Entomology Research Laboratory 10:71-76.

Hogsette, J. A. 2003. United States Department of Agriculture—Agricultural Research Service research on veterinary pests. Pest Management Science 59:835-841.

Hogsette, J.A., D.A. Carlson, and A.S. Nejame. 2002. Development of granular boric acid sugar baits for house flies (Diptera: Muscidae). Journal of Economic Entomology. 95: 1110-1112.

Howard, L. O. 1911, The house fly—Disease carrier. New York, Frederick A. Stokes.

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

Jiang, Y., C. Lei, C. Niu, Y. Fang, C. Xiao, and Z. Zhang. 2002. Semiochemicals from ovaries of gravid females attract ovipositing female houseflies, *Musca domestica*. Journal of Insect Physiology 48:945-950.

Johnson, G., and J. Campbell. 1987. Evaluation of sticky traps for collecting face flies (Diptera: Muscidae) from different locations in an irrigated pasture. Journal of Medical Entomology 16:190-194.

Jones, C., S. Isard, and M. Cortinas. 1999. Dispersal of synanthropic Diptera: Lessons from the past and technology for the future. Annals of the Entomological Society of America 92:829-839.

Kalandadze, L. P., and S. V. Chilingarova. 1942. The role of the substrates in the oviposition and pre-imaginal development of *Musca vicina* Macq. (In Russian.). Medical Parasitology 11:105-112.

Kaufman, P., C. Reasor, D. Rutz, J. Ketzis, and J. Arends. 2005a. Evaluation of *Beauveria bassiana* applications against adult house fly, *Musca domestica*, in commercial caged-layer poultry facilities in New York state. Biological Control 33:360-367.

Kaufman, P., D. Rutz, and S. Frisch. 2001. Sticky traps for large scale house fly (Diptera: Muscidae) trapping in New York poultry facilities. Journal of Agricultural and Urban Entomology 18:43-49.

Kaufman, P., D. Rutz, and S. Frisch. 2005b. Large sticky traps for capturing house flies and stable flies in dairy calf greenhouse facilities. Journal of Dairy Science 88:176-181.

Kobayashi, H. 1934. The influence of foods on the fecundity of *Musca domestica*. Keijo Journal of Medicine 5: 36-67.

Kolbe, W. 2004. Flies, Gnats, & Midges, Pages 1397 *in* A. Mallis, ed. Handbook of Pest Control, GIE Media, Inc.

Kuzina, O. 1940. Role des organes sensitifs chez la *Musca domestica* L. dans la recherche du fumier et dans la ponte des oeufs. (In Russian). Medical Parasitology 9:340-349.

Laing, J. 1935. On the ptilinum of the blow-fly (*Calliphora erythrocephala*). Quarterly Journal of Microscopical Science 77:497-521.

Larsen, E. B., and M. Thomsen. 1940. The influence of temperature on the development of some species of Diptera. Videnskabelige Meddelel ser fra Dansk Naturhistorisk Foreninz: Kobenhaun 104:1-75.

Lazarus, W., D. Rutz, R. Miller, and D. Brown. 1989. Costs of existing and recommended manure management practices for house fly and stable fly (Diptera: Muscidae) control on dairy farms. Journal of Economic Entomology 82:1145-1151.

Learmount, J., P. Chapman, and A. Macnicoll. 2002. Impact of an insecticide resistance strategy for house fly (Diptera: Muscidae) control in intensive animal units in the United Kingdom. Journal of Economic Entomology 35:1245-1250.

Lecuona, R., M. Turica, F. Tarocco, and D. Crespo. 2005. Microbial control of *Musca domestica* (Diptera: Muscidae) with selected strains of *Beauveria bassiana*. Journal of Medical Entomology 42:332-336.

Legner, E., and H. Brydon. 1966. Suppression of dung-inhabiting fly populations by pupal parasites. Annals of the Entomological Society of America 59:638-651.

Legner, E., and E. Dietrick. 1974. Effectiveness of supervised control practices in lowering population densities of synanthropic flies on poultry ranches. Entomophaga 19:467-478.

Lindsay, D. R., W. H. Stewart, and J. Watt. 1953. Effect of fly control on diarrheal disease in an area of moderate morbidity. Public Health Reports 68:361-367.

Loeser, C., H. Ulbricht, and H. Seidel. 2004. Degradation of Polycyclic Aromatic Hydrocarbons (PAHs) in Waste Wood. Compost Science & Utilization 12:335-341.

Lysyk, T., and R. Axtell. 1986. Movement and distribution of house flies (Diptera: Muscidae) between habitats in two livestock farms. Journal of Economic Entomology 79:993-998.

MacCreary, D., and G. Haenlein. 1962. House fly breeding in oak sawdust and peanut hulls used as bedding in calf pens. Journal of Economic Entomology 55:419.

Mansingh, A., R. W. Steele, and B. N. Smallman. 1972. Pheromone effects of cis-9 long chain alkenes on the common house fly—An improved sex attractant combination. Canadian Entomology 104.

Mayer, M. S., and J. D. James. 1971. Response of male *Musca domestica* to a specific olfactory attractant and its initial chemical purification. Journal of Insect Physiology 17:833-842.

Mellor, J. E. M. 1919. Observations on the habits of certain flies, especially of those breeding in manure. Annals of Applied Biology 6:53-88.

Mer, G., and M. Paz. 1960. Control of house flies in rural areas by means of traps. Rivista di Parassitologia (Rome) 21:143-150.

Meyer, J., and T. Shultz. 1990. Stable fly and house fly breeding sites on dairies. California Agriculture 44:28-29.

Mitchell, E., and F. Tingle. 1975. Effect of Muscalure on house fly traps on different color and location in poultry houses. Journal of Georgia Entomological Society 10:165-168.

Mulla, M., and J. Ridsdill-Smith. 1986. Chemical attractants tested against the Australian bush fly *Musca vetusitissima* (Diptera: Muscidae). Journal of Chemical Ecology 12:261-270.

Mulla, M. S., Y.-S. Hwang, and H. Axelrod. 1977. Attractants for synanthropic flies: Chemical attractants for domestic flies. Journal of Economic Entomology 70:644-648.

Mullens, B., J. Meyer, and T. Cyr. 1987. Infectivity of insect-parasitic nematodes (Rhabditida: Steirnemeatidae, Heterohabditidae) for larvae of some manure-breeding flies (Diptera: Muscidae). Environmental Entomology 16:769-773.

Muniz, R. 1967. Observaciones sobre la busqueda de sitios de reposo y estudio experimental sobre la seleccion de colores por *Musca domestica* en el ambiente natural. M.S. thesis, Institución Politecnico Nacional, Mexico, D.F.

Murvosh, C., and C. Thaggard. 1966. Ecological studies of the house fly. Annals of the Entomological Society of America 59:533-547.

Murvosh, C. M., R. L. Fye, and G. C. LaBrecque. 1964a. Studies on the mating behavior of the house fly, *Musca domestica* L. Ohio Journal of Science 64:264-271.

Murvosh, C. M., G. C. LaBrecque, and C. N. Smith. 1964b. Effect of three chemosterilants on houe fly longevity and sterility. Journal of Economic Entomology 51:89-93.

Nava, A. 1967. Preferencias de *Musca domestica* en la seleccion de superficies de color. Professional thesis, Institucion Politecnico Nacional, Mexico, D.F.

Nelson, D. R., J. W. Dillwith, and G. J. Bloomquist. 1981. Cuticular hydrocarbons of the house fly, *Musca domestica*. Insect Biochemistry 11:187-197.

Newstead, R. 1908. On the habits, life-cycle and breeding places of the common house-fly (*Musca domestica*, Linn.). Annals of Tropical Medical Parasitology 1:507-520.

Nolan, M. I., and J. Kissam. 1985. *Ophyra aenescens*: A potential bio-control alternative for house fly control in poultry houses. Journal of Agricultural Entomology 2:192-195.

Nolan, M. I., and J. Kissam. 1987. Nuisance potential of a dump fly, *Ophyra aenescens* (Diptera: Muscidae), breeding at poultry farms. Environmental Entomology 16:828-831.

Patterson, R. S. 1957. On the causes of broken wings of the house fly. Journal of Economic Entomology 50:104-105.

Pickens, L. 1989. Factors affecting the distance of scatter of house flies (Diptera: Muscidae) form electrocuting traps. Journal of Economic Entomology 82:149-151.

Pickens, L. 1994. Bring filth flies under control. Pest Management 13:14-18.

Pickens, L. 1995. Baited fly traps 1900 to 1995. The IPM practitioner: The newsletter of integrated pest management 17:1-6.

Pickens, L., D. Hayes, N. Morgan, E. Schmidtmann, and R. Miller. 1986. Pyramid traps for monitoring *Musca vitripennis* (Diptera: Muscidae). Journal of Medical Entomology 23:112-113.

Pickens, L., and R. Miller. 1987. Techniques for trapping flies on dairy farms. Journal of Agricultural Entomology 4:305-313.

Pickens, L., R. Miller, and G. R. Mowry. 1973. An improved bait for flies (Diptera: Muscidae, Calliphoridae). Journal of Medical Entomology 10:84-88.

Pickens, L., N. Morgan, and R. Miller. 1972. Comparison of traps and other methods for surveying density of populations of flies in dairy barns. Journal of Economic Entomology 65:144-145.

Pickens, L., N. Morgan, and R. Thimijan. 1969. House fly response to fluorescent lamps: Influenced by fly age and nutrition, air temperature, and position of lamps. Journal of Economic Entomology 62:536-539.

Pickens, L., N. Morgan, and R. Thimijan. 1975. Bait light combinations evaluated as attractants for house flies and stable flies. Journal of Medical Entomology 11:749-751.

Rambo, G. 1995. Light traps: Control devices or monitoring techniques? Pest Control Technology 23:106-108.

Richardson, C. 1917. The response of the house-fly to certain foods and their fermentation products. Journal of Economic Entomology 10:102-109.

Riemann, J. G., D. J. Moen, and B. J. Thorson. 1967. Female monogamy and its control in house flies. Insect Physiology 13:407-418.

Robbins, W., M. Thompson, R. Yamamoto, and T. Shortino. 1965. Feeding stimulants for the female house fly, *Musca domestica* Linnaeus. Science 147:628-630.

Robinson, W. 1996, Urban entomology: Insect and mite pests in the human environment. London, Chapman and Hall.

Rockstein, M. 1957. Longevity of male and female house flies. Journal of Gerontology 12:253-256.

Rogoff, W., A. D. Beltz, J. Johnsen, and F. Plapp. 1963. A sex pheromone in the housefly, *Musca domestica* L. Journal of Insect Physiology 10:239-246.

Rogoff, W. M. 1965. Mating of the house fly, *Musca domestica* L., in monitored darkness. Journal of Medical Entomology 2:54-56.

Rueda, L., and R. Axtell. 1985. Effect of depth of house fly pupae in poultry manure on parasitism by six species of Pteromalidae (Hymenoptera). Journal of Entomological Science 20:444-449.

Sacca, G. 1963. Comparitive bionomics in the genus *Musca*. Annals of the Entomological Society of America:341-358.

SAS. 2001. SAS stat user's guide version 8.01. Cary, NC, SAS Institute.

Schiefer, B. 1970. An evaluation of electrocutor grid traps as a means of housefly control. M.S. thesis, University of Arkansas.

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

Schoof, H., and R. Siverly. 1954. Privies as a source of fly production in an urban area. American Journal of Medical Tropical Hygeine 3:930-935.

Scott, J., T. Alefantis, P. Kaufman, and D. Rutz. 2000. Insecticide resistance in house flies from caged-layer poultry facilities. Pest Management Science 56:147-153.

Skovard, H., and G. Nachman. 2004. Biological control of house flies *Musca domestica* and stable flies *Stomoxys calcitrans* (Diptera: Muscidae) by means of inundative releases of *Spalangia cameroni* (Hymenoptera: Pteromalidae). Bulletin of Entomological Research 94:555-567.

Skovmand, O., and H. Mourier. 1986. Electrocuting light traps evaluated for the control of house flies. Journal of Applied Entomology 102:446-455.

Smallegange, R. 2004. Fatal attraction. Control of the housefly (Diptera: Muscidae). Entomologische Berichten (Amsterdam) 64:87-92.

Smallegange, R., and C. Otter. 1998. Attraction of the housefly (*Musca domestica* L.). Proceedings of the Section Experimental and Applied Entomology of the Netherlands Entomological Society (N.E.V.) Amseterdam 9:201-201.

Steinkraus, D., and J. Kramer. 1987. Susceptibility of sixteen species of Diptera to the fungal pathogen *Entomophthora muscae* (Zygomycetes: Entomophthoraceae). Mycopahologia 100:55-63.

Stinson, E. 1989. Pesticides: Killers in disguise. Virginia Wildlife 50:22-25.

Thimijan, R., L. Pickens, and N. Morgan. 1970. A trap for house flies. Journal of Economic Entomology 63:1030-1031.

Turner Jr., E., and P. Ruszler. 1989. Research Note: A quick and simple quantitative method to monitor house fly populations in caged layer houses. Poultry Science 68:833-835.

Uebel, E. C., P. E. Sonnet, and R. W. Miller. 1976. Housefly sex pheromone enhancement of mating strike activity combination of (Z)-9-tricosene with branched saturated hydrocarbons. Journal of Economic Entomology 5:905-909.

Uvarov, B. 1931. Insects and climate. Transactions of the Entomological Society, London 79:1-247.

Watt, J., and D. R. Lindsay. 1948. Diarrheal disease control studies. I. Effect of fly control in a high morbidity area. *Public Health Reports* 63:1319-1334.

West, L. 1951. The house fly. Its natural history, medical importance, and control. Ithaca, NY, Comstock.

Wieting, J., and W. Hoskins. 1939. The olfactory response of flies in a new type of insect olfactometer. II. Responses of the housefly to ammonia, carbon dioxide, and ethyl alcohol. Journal of Economic Entomology 32:24-29.

Wills, L., B. Mullens, and J. Mandeville. 1990. Effects of pesticides on filth fly predators (Coleoptera: Histeridae, Staphylinidae; Acarina: Macrochelicidae, Uropididae) in caged layer poultry manure. Journal of Economic Entomology 83:451-457.

Zimgrone, L. D., W. N. Bruce, and G. C. Decker. 1959. A mating study of female house fly. Journal of Economic Entomology 52:236.

Zurek, L., S. Denning, C. Schal, and D. Watson. 2001. Vector competence of *Musca domestica* (Diptera: Muscidae) for *Yersinia pseudotuberculosis*. Journal of Medical Entomology 38:333-335.

Zurek, L., D.W. Watson, S. B. Krasnoff, and C. Schal. 2002. Effect of the entomopathogenic fungus, *Entomophthora muscae* (Zygomycetes: Entomophthoraceae), on sex pheromone and other cuticular hydrocarbons of the house fly, *Musca domestica*. Journal of Invertebrate Pathology 80:171-176.

BIOGRAPHICAL SKETCH

Ryan Merrill Welch was born on June 23, 1982, to Dwight and Amy Welch, in Jacksonville, FL. There he attended Parkwood Heights Elementary School, Arlington Middle School, and Stanton College Preparatory School. He graduated from Stanton College Preparatory School with high school and international baccalaureate diplomas in 2000. He attended the University of Florida from 2000 to 2004 and graduated cum laude earning a Bachelor of Science in entomology and a Bachelor of Arts in political science. He attended the University of Florida from 2004 to 2006 and received his Master of Science in entomology under Dr. Phil Koehler's guidance.