Monitoring and management of thrips populations in vegetables, row crops, and greenhouse crops in Virginia

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ABSTRACT

Thrips are pests in a variety of crops and are responsible for millions of dollars in damage worldwide. In Virginia there are a few key thrips species that cause a large portion of damage to both vegetable and floricultural crops. Three prominent pests include *Frankliniella tritici* (Fitch), *Frankliniella fusca* (Hinds), and *Frankliniella occidentalis* (Pergande). Significant yield losses in row crops such as cotton, peanuts and vegetables have been attributed to feeding and oviposition of these insects in high densities. In addition, both *F. fusca* and *F. occidentalis* can transmit plant pathogenic tospoviruses, such as tomato spotted wilt virus (TSWV), in certain susceptible crops. While all of these thrips species are difficult to detect due to their cryptic lifestyles, *F. occidentalis* is a particularly challenging pest to manage due to its resistance to many insecticides commonly used for thrips treatment.

Early spring weeds were sampled for the presence of *F. occidentalis* in 2008 and 2009 in eastern Virginia. Weed samples consisted of mustard, henbit and wild radish and were collected from several different sites on the Eastern Shore of Virginia. During the summer of 2008, 2009 and 2010 various agroecosystems were sampled for the relative incidence of *F. occidentalis*. Overall, thrips numbers were very low in weed samples. *F. occidentalis* was detected in early spring weed samples in 2009 at a few of the sites sampled. In nearly every habitat, the species composition was dominated by *F. fusca* and *F. tritici*, with *F. occidentalis* occurring in very low numbers.

Two different lures were evaluated in their ability to attract *Frankliniella* spp. thrips. The lures included Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO), a floral kairomone lure composed of a proprietary floral compound mixture, and Thripline$_{AMS}$ (Syngenta Bioline Ltd., Oxnard, CA) pheromone lure, containing the aggregation pheromone of *F. occidentalis*. In spring 2009 and 2010 lure experiments were conducted in several different agroecosystems including: a tomato and potato field in Painter, VA, a cotton and peanut field in Suffolk, VA, and grass fields near a greenhouse in Virginia Beach, VA, and a high tunnel in Chesapeake, VA, as well as within these structures. Baited and non-baited sticky cards were arranged in a completely randomized design, with a pan trap located in the center of each plot. Traps were collected approximately twice weekly. *F. fusca* numbers were low and catches on sticky cards were not significantly affected by either lure. Sticky cards baited with the kairomone caught more flower thrips than traps baited with the pheromone, or the non-baited traps, especially when thrips numbers were high.

Several biologically derived insecticides including: essential oils, spinetoram, spinosad, pyrethrins, and azadirachtin, were evaluated in their efficacy against thrips in several different crops. Randomized complete block design experiments were carried out in: tomatoes, snap beans, collards, soybeans, cotton and peanuts grown in several locations in southeastern Virginia in 2009 and 2010. Both spinetoram and spinosad reduced thrips numbers the most effectively compared with the untreated control. Peanut and cotton treated with spinosad, and treatments containing spinetoram suffered less thrips injury compared with the control, and yield was higher in cotton plots treated with spinetoram.

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Table of Contents

List of Tables

List of Figures

Introduction

Thrips are tiny insects that belong to the order Thysanoptera. There are many species of thrips, some of which are serious pests of crops. Often, thrips that are highly adaptable and polyphagous tend to be serious crop pests, whereas species that only breed on a single species of plant tend to be minor pests (Mound 2005). Thrips feeding can cause injury to leaves, flowers, and fruit of a great diversity of crops (Pohronezny et al. 1986, Childers 1997). Feeding injury to fruit can result in discoloration, deformity and reduced marketability (Mound 1997). Row crops such as cotton, peanuts, beans and vegetables, as well as floral plants grown in greenhouses are the most commonly affected in the eastern United States. While direct injury alone can be devastating to crop yield, the ability of some species of thrips to transmit tospoviruses such as tomato spotted wilt virus (TSWV) and impatiens necrotic spot virus (INSV) has elevated this insect group to one of the most serious economic pests in agriculture. In addition, some of the main pest thrips have developed resistance to commonly used pesticides, which further complicates pest management.

A number of strategies have been explored to manage thrips, but most emphasize the use of pesticides. However, due to their short life spans and ability to produce many progeny, some species of thrips tend to rapidly become resistant to insecticides. It is therefore wise to employ other methods for thrips control using an integrated pest management (IPM) approach, which utilize several tactics to reduce pest populations without relying completely upon a single pesticide. Such tactics use information on the life cycles of pests and their interaction with their host plants and the environment (U. S. Environmental Protection Agency 2009). This knowledge is then used with available pest control methods to manage damage with the least amount of hazard to people and the environment.

One species of thrips that is particularly difficult to manage due to insecticide resistance is *Frankliniella occidentalis* (Pergande), the western flower thrips. In addition to being highly polyphagous and thus able to feed upon a variety of economically important crops, this species is also a vector of some important tospoviruses including TSWV and INSV. Flower thrips, *Frankliniella tritici* (Fitch), looks very similar to *F. occidentalis* and also can be problematic in crops such as tomato despite its inability to transmit tospoviruses. Another important species of thrips in Virginia is the tobacco thrips, *Frankliniella fusca* (Hinds). This species of thrips is also capable of transmitting tospoviruses and can be found in a variety of important crops. Until the mid-2000s, *F. occidentalis* was not a significant pest in Virginia, but caused serious damage to crops in eastern Virginia in 2007 (Kuhar and Herbert, personal communication). Therefore, it has become important to thoroughly study the current pest complex and management of these insects in Virginia. The purpose of this research was to:

1. Determine the relative incidence of *F. occidentalis* in thrips populations in selected agroecosystems in eastern Virginia

2. Evaluate the effects of two different semiochemical attractants on thrips catch on yellow sticky cards

3. Evaluate the efficacy of several biologically derived insecticides in their ability to control thrips on various crops

Literature Review

History and Significance

Thrips (Thysanoptera) were first described in 1744 by De Geer under the name *Physapus* (Lewis 1997a). Later in 1744, Linnaeus, ignoring this name, placed the four species that he knew of into the genus *Thrips*. An English entomologist named Haliday elevated this group to the rank of order in 1836, but the common name thrips is still applied to any insects within the order.

For years interest in thrips was lacking, with few entomologists specializing in the study of these insects. This is probably due to the fact that they are quite small and difficult to study, and specimens tend to require meticulous and careful attention when being prepared and mounted for microscopic identification. After about 1900, interest in thrips increased with descriptive activity peaking in the 1920s and 30s. Slightly more than 5000 species of the estimated 8000 extant species have been recognized and placed into two suborders and eight families (Gaston and Mound 1993) that exhibit a range of biology, behavior and morphology. Approximately 50% of the 5000 known species of thrips are phytophagous, with barely 10% of these posing a threat to man's economic activity, and out of these, a mere 1.0% cause severe crop damage (Mound and Teulon 1995). However, those few species that are known to cause damage can be very problematic in certain crops and quite difficult to control. Moreover, as a result of increased globalization of trade and transportation, many of the major pest thrips are cosmopolitan pests. Alien species have been introduced to new areas and endemic species have had the opportunity to transfer to new hosts (Mound 1983).

Focus upon certain species of thrips has not only been due to the movement of thrips themselves, but has also depended upon where thysanopterists have chosen to reside, and upon species which seemed most harmful at the time. Prior to the 1980s, *Thrips tabaci* Lindeman was given much attention whereupon emphasis shifted to other pests, particularly *F. occidentalis* (Mound and Teulon 1995). In other areas of the world, focus upon thrips shifted among thrips that were largely responsible for negative impacts upon large cash crops. As agrobusinesses were expanding during the middle of the century in North America, species of *Thrips*, *Frankliniella*, *Scirtothrips*, and *Hercinothrips* received a large amount of attention, which has continued due to the gradual failure of conventional control methods (Lewis 1997a).

The movement of many species, particularly *F. occidentalis* and *Thrips palmi* Karni has been largely facilitated by an increase in global transport of fresh flowers, fruit and vegetables, as well as large-scale plant propagation enterprises becoming established. As a result, focus has shifted again, and some of the most lucrative and productive horticultural and agricultural parts of the world have been negatively impacted.

Along with shifts in focus upon different crops and species of thrips have come drastic changes in control measures. Various methods of cultural control such as stubble burning and regard for planting and harvesting times accompanied the use of crude chemicals and plant extracts applied in large volumes early in the century. Sophisticated synthetic insecticides applied at lower volumes by more refined equipment have gradually replaced such control measures. However, even with the advancement of technology, many pests have developed resistance to these pesticides, which has led to the search for alternative methods for control, such as beneficial organisms including nematodes, mites and insects, as well as fungal and bacterial pathogens. Commonly such organisms are used as part of IPM systems where some of the older cultural control methods have been combined with modern narrower spectrum insecticides that have limited impact upon biological control agents. This form of control is both effective and environmentally sound.

Despite the constant advancement of technology, thrips continue to pose a serious threat to numerous crops worldwide due to their ability to inflict indirect as well as direct injury and spread plant diseases. In fact, the significant thysanopteran pests could be as important to world crop production as whiteflies (Aleurodidae), mealy bugs and scale insects (Coccoidea), or even aphids (Aphididae) (Lewis 1997a).

Thrips biology

Both life cycle and anatomy have allowed thrips to become very successful pests. Short generation time and high fecundity allow populations to reach staggering numbers within a relatively short amount of time (Georghiou and Taylor 1977). In addition, thrips can be fairly mobile, and because they are quite small and light, can be blown long distances by the wind. Particularly strong winds, such as those associated with the inter-tropical front are responsible for transporting thrips vast distances in Central America (Mound and Teulon 1995). Thrips can also travel vast distances by plane, ship or vehicle when they are hidden among vegetation which is being transported (Lewis 1997a).

Species of thrips that are opportunists have particular advantage in colonizing new habitats. Life-history strategies of opportunistic species including early reproduction, polyphagy, high fecundity, production of numerous offspring, short generation time, high vagility, search behavior and lack of diapause increase the likelihood of establishment and spread compared to species with different life-history strategies (Mound and Teulon 1995).

Although life cycle can differ among species, a typical terebrantian (Thripidae) pest species begins life as an egg, and will then go through two active feeding larval instars, and two relatively inactive non-feeding pupal instars known as the prepupa and pupa (Lewis 1997a). A few pestiferous thrips that are in the tubuliferan family, Phlaeothripidae, pass through an additional pupal instar. The females in the suborder Terebrantia insert their smooth shelled eggs into plant tissue using a saw-like ovipositor. Thrips which do not have an ovipositor, such as Phlaeothripids, stick their eggs onto the surface of vegetation.

Female thrips can produce fertilized eggs by mating with a male, or they can develop progeny parthenogenetically (Ananthakrishnan 1984). Such means of reproduction allows thrips to develop resistance to pesticides more rapidly. The ability to obtain resistance to pesticides can be further enhanced when female thrips survive a treatment and produce haploid males which they then mate with and produce more resistant female and male progeny (Moritz 1998). Until recently, it was thought that unmated females could only produce haploid males; however, it now appears that a small number of these are female (Kumm and Moritz 2010).

Thrips possess piercing-sucking mouthparts, which are used to pierce various plant parts such as: leaves, flowers, seeds, pollen grains and fruit. They can also be used to consume open liquids including nectar, water, or even insect secretions (Kirk 1995). Some thrips feed predatorily on mites and small insects, eggs laid by mites and lepidopterans, and on other thrips, even those of their own species. A feeding hole is created in the plant when the mandible is driven downwards by thrusting the head, and the mouth cone is retracted (Mound 2005). Two maxillary stylets contain a feeding/salivary channel which is used to inject saliva into the plant cell (Mound and Teulon 1995); the cibarial muscles provide sufficient suction to withdraw contents of the plant cell through the feeding tube, leaving behind necrotic scars (Chisholm and

Lewis 1984, Childers and Achor 1995, Mound 2005, Frank 2009). If they are present, tospoviruses are transmitted into the plant during the feeding process (Takacs et al. 2008).

Nutrition plays a very important role in thrips' lives and to a large extent the struggle to obtain critical components of the diet appears to determine habitat preference (Ananthakrishnan 1984). Some species of thrips tend to favor various parts of plants such as flowers, leaves or fruit. This preference could be due to differing nutrient levels in the various plant parts. Young or growing plant tissues tend to have higher concentrations of soluble amino acids and amides in the sap compared to mature parts of the plant, making them more nutritious.

Obtaining adequate nutrition can be a problem because insects have a higher content of protein nitrogen than the plants that they feed on (Kirk 1995). Increased nitrogen content has led to an increase in survival, growth and reproduction in several studies, and it is therefore thought that nitrogen is very important for survival. Insects, some seeds and leaf buds tend to be high in nitrogen content whereas pollen, fleshy fruits and mature leaves are low in nitrogen. Thrips' feeding patterns tend to be affected by nitrogen content, with many thrips feeding on foods high in nitrogen such as pollen and other insects, while thrips feeding on low nitrogen leaves can meet their nitrogen requirements by occasional predation. An example of this pattern can be seen in *F. occidentalis,* which can be both phytophagous and predatory. This tendency to consume an eclectic diet probably enhances its ability to inhabit a wide variety of habitats. Although habitat preference does appear to reflect nutritional requirements, it could also be due to other factors such as microclimate or morphological features of plants including those that attract (e.g., pollen and nectar) and deter (e.g., pubescence) insects.

Flower thrips, *Frankliniella tritici*

Frankliniella tritici is indigenous to North America and is generally distributed over most of the United States (Watts 1936). This species of thrips is known to cause damage to a wide variety of crops including: ornamental shrubs and flowers, strawberries, peaches, plums, apricots, oranges, cotton, small grains, alfalfa, beans and peas. It appears to exhibit a preference for plants belonging to the grass, legume and rose families, with a somewhat less prominent preference for the Compositae and Cruciferae during the summertime. During the winter, *F. tritici* tends to prefer small grains and plants belonging to the mustard family. All stages of *F. tritici* tends to prefer terminal buds and blossoms, and can be found in aggregations in these areas (Ananthakrishnan 1984).

Eggs are faint creamy white, kidney-shaped, and approximately 0.25 mm in length, and are deposited into plant tissue by means of the female's saw-like ovipositor. The length of time required for incubation varies greatly with temperature, with shorter incubation periods associated with warmer temperatures. According to Watts (1936), the average length of time required for incubation ranges from 2 days at approximately 27°C to 4.5 days at approximately 21°C.

After hatching, the nearly colorless larvae become a deeper yellow with age. The average duration of the first instar ranges from 1.7 days at 21°F to 3.3 days at 16°C when raised indoors on cotton, while the second instar lasts from 1.5 days at 27°C to 4.1 days at 18°C (Watts 1936). Outdoors, the average developmental time for the first instar is 5.4 days at 10°C, while the second instar requires 7.3 days at 4°C.

The prepupa is similar in appearance to the larva, although it is much more sedentary and has short wing pads. Although they may pupate on plants, mature second instars usually drop to the ground to complete the pupal stage. In the laboratory, the prepupal instar lasts on average between 1.0 and 2.1 days at 28°C and at 21°C, respectfully. The pupa has longer wing pads compared with the prepupa, and the antennae are fastened along their entire length to the upper surface of the head. Between 1.8 days at 28°C and 3.4 days at 22°C are required on average for the pupa to complete this stage when raised in the laboratory.

Adult females tend to be slightly more than a millimeter in length with the general body color being a brownish yellow with an orange region on the thorax. Males are much smaller, about 0.7 mm, and are usually paler than the female. In the laboratory the average amount of time required for males and females to complete development varies from 9 days at 28°C to 16 days at 21°C. The lifespan of an adult female can range on average from 11 days at 27°C to 61 days at 21°C when raised in the laboratory, while males only survive 3 days at 27°C to 37 days at 21°C. In the field, females survive on average 26 days at approximately 21°C while males only live approximately 15 days at that same temperature. According to Watts (1936), females produce on average 28.6 eggs, although individuals can produce many more, up to 119 eggs when fed seedling cotton.

Females can reproduce parthenogenically with unfertilized eggs resulting in male progeny. A recent study performed on *F. occidentalis* showed that a small proportion of young produced by unfertilized females were female (Kumm and Moritz 2010). Due to the fact that *F. tritici* is in the same genus, it may also be assumed that *F. tritici* also exhibits identical reproductive behavior. In South Carolina, there is a continuous overlapping of generations with 12-15 generations developing within a single year (Watts 1936). From early spring through most of the summer, females outnumber males, but the proportionate differences become reduced during the autumn. In South Carolina, *F. tritici* remain active during the wintertime, although development of immatures is slowed and populations are smaller (Watts 1936).

Tobacco thrips, *Frankliniella fusca*

As mentioned earlier, tobacco thrips, *F. fusca,* is another species of thrips that causes damage in several crops in the southern United States. Their current range spreads from eastern Canada and the United States, west to the Rocky Mountains (Capinera 2001). This species of thrips is capable of transmitting tospoviruses, including TSWV and INSV, and can be found in a wide variety of host plants including crops such as: tomato, peanut, pepper, cotton, tobacco, bean, beet, cantaloupe, carrot, corn, cowpea, cucumber, onion, pea, potato, watermelon, alfalfa, barley, various varieties of clover, lespedeza, rye, vetch, wheat, and occasionally corn and oats (Ananthakrishnan 1984, Frantz and Mellinger 1990, McPherson et al. 1999, Capinera 2001, Osekre et al. 2009). In the United States, this species of thrips is particularly problematic in early stage peanuts, and in cotton has been reported to cause a reduction of leaf surface of seedlings by 50% or more and height by approximately 20% by the time the plants are six weeks old (Ananthakrishnan 1984). McPherson et al. (1999) found *F. fusca* to be the most common foliage thrips observed most years during a 6-year study on tobacco grown in Georgia.

According to Capinera (2001), *F. fusca* also is supported by numerous hosts including: Bermudagrass, *Cynodon dactylon*; blue toadflax, *Linaria canadensis*; broomsedge, *Andropogon virginicus*; buttercup, *Ranunculus* sp.; cocklebur, *Xanthium* sp.; crabgrass, *Digitaria* sp.; cutleaf evening primrose, *Oenothera laciniata*; dandelion, *Taraxacum officinale*; dog fennel, *Eupatorium capillifolium*; false dandelion, *Pyrrhopappus carolinianus*; feathergrass, *Leptochloa filiformis*; Johnsongrass, *Sorghum halepense*; little barley, *Hodeum pusillum*; rabbit tobacco, *Gnaphalium obtusifolium*; sand blackberry, *Rubus cuneifolius*; shepherdspurse, *Capsella bursa-pastoris*; spiny sowthistle, *Sonchus asper*; wild lettuce, *Lactuca* sp.; wild radish, *Raphanus raphanistrum*; wood sorrel, *Oxalis* spp.; and a grass, *Brachiaria extensa*.

Frankliniella fusca requires approximately 15-21 days to complete a life cycle (Capinera 2001). Females insert a white, bean-shaped egg measuring 0.25 mm long into foliage with one end slightly protruding. After 3-10 days, depending on conditions in the southeastern United States, larvae emerge. Immature thrips must pass through two larval and two pupal instars. At 25°C, the first larval instar lasts on average 1.1 days, and the second instar lasts about 4.7 days. At 35°C only one day is required to complete the first larval instar, and the second lasts 2.6 days. Larvae are rarely found in exposed places and usually remain in cryptic habitats including terminal growth and blossoms. The larva moves to the soil to pupate after completing both larval instars. Both the prepupal and pupal instars are yellow, and have wing pads. At 25°C the prepupal and pupal stages last on average 1.1 and 1.4 days respectively, and at 35°C these phases last 0.8 and 1.4 days.

Adults tend to have elongated, slender bodies measuring 1.0-1.3mm long and often have two pairs of fringed wings. The abdomen is dark-brown and the head and thorax are light-brown or yellow-brown. Antennae are eight-segmented. During the winter months both long-winged (macropterous) and short-winged (brachypterous) forms occur. Both adults and larvae exhibit similar feeding behaviors. Longevity of adult females ranges between 6-10 days and females have been reported by Lowry et al. (1992) to produce 13-24 eggs per female. However, diet appears to have an effect on longevity and fecundity. In a study conducted by Eddy and Livingstone (1931), when fed cotton, mated female *F. fusca* lived an average of 34.5 days and produced an average of 29.8 eggs per female. *F. fusca* can reproduce by mating or parthenogenically, with mated females producing both males and females and unmated females producing haploid males.

The growth stage of a plant is very important in determining the amount of injury (Ananthakrishnan 1984). *F. fusca* tends to attack seedling cotton as soon as seedlings emerge and subsequent generations develop, with populations usually peaking about a month after the seedlings have emerged. Young leaves become curled and older leaves become silvery, speckled and crinkled when thrips feed and oviposit on plant tissue (Capinera 2001). The seedlings may also appear scorched or burned when young tissue is killed. When thrips attack in sufficiently high numbers during very early development of cotton, seedlings will become malformed and stunted, and yield can also be impacted (Watson 1965, Ananthakrishnan 1984, Herbert et al. 2007).

In addition to causing direct damage through feeding and oviposition, *F. fusca* can also transmit tospoviruses, including TSWV and INSV, which escalates this thrips as a pest. After contracting the virus as a larva, the thrips remains infective throughout its life. Weeds serve as overwintering habitat for tobacco thrips, and can also become a virus reservoir when susceptible plants harbor infected thrips (Durant et al. 1994, Cho et al. 1995).

As with other flower thrips, *F. fusca* also has a cryptic lifestyle, which makes it another difficult pest to manage. Unlike *F. occidentalis* and *F. tritici,* which often reside in flowers, *F. fusca* feeds mainly on leaves, although they may be found in flowers as well.

Western flower thrips, *Frankliniella occidentalis*

One species of particular interest is *F. occidentalis*, which until recently was not known to be present in Virginia in significant numbers (Nault et al. 2003). Originally restricted mostly to the western United States, this species of thrips has spread east to Georgia (Capinera 2001), and very recently has been found in Virginia (Kuhar unpublished data). It has become a world-renowned pest, spreading into southern Canada and to other continents. The expansion of its range has been assisted by movement of plants infested with these insects. Survival rates are highest in warm climates, with overwintering taking place outside on plants along the west coast and throughout the southeastern states. In very cold climates this species of thrips does not overwinter, but re-invades areas annually from greenhouses, or they may be introduced along with seedlings from warmer areas. Although *F. occidentalis* looks almost identical to *F. tritici,* this species of thrips tends to be more resistant to many of the current pesticide treatments commonly used to control thrips, and it is also capable of transmitting tospoviruses. These factors have elevated *F. occidentalis* to one of the most serious pests in many economically important crops worldwide.

Frankliniella occidentalis has a very wide host range including cucumber, onion, pepper, potato, lettuce, and tomato, with fruit crops such as plum, pear, peach, apple, blackberry and blueberry also serving as hosts. Although tomato is quite seriously injured directly by thrips, the ability of this insect to transmit TSWV increases the damage when the disease is present. *F. occidentalis* also occurs on a variety of field crops including alfalfa, canola, crimson and white clover, millet, peanut rye, vetch and wheat. Although myriad other species of weeds can serve as hosts, some of the best include black nightshade, *Solanum nigrum*; cheese weed, *Malva parviflora*; daisy fleabane, *Erigeron annuus*; dandelion, *Taraxacum officinale*; false dandelion, *Pyrrhopappus carolinianus*; jimson weed, *Datura stramonium*; galinsoga, *Galinsoga parviflora*; lambsquarters, *Amaranthus* spp.; prickly lettuce, *Lactuca serriola*; sorrel, *Oxalis* spp. sowthistle, *Sonchus oleraceus*; and wild radish, *Raphanus raphanistrum* (Capinera 2001).

In some species of plants, *F. occidentalis* tends to be abundant in flowers, but is not able to thrive on leaves or stems (Terry 1997). This could be due to chemicals within the leaves (e.g., tannins or flavenoids), or morphological features (e.g., pubescence) which cause some types of plants to adversely affect feeding and development. In addition, virus infected plants may also negatively affect feeding thrips.

Female *F. occidentalis* typically begin oviposition 72 hours after emergence and continue to produce young throughout almost the entire remaining lifecycle (Ananthakrishnan 1984). At optimum temperatures, which range between 20°C and 25°C, a female *F. occidentalis* can hatch, mature into an adult, and produce on average 95.5 eggs within 10-30 days, depending on the plant host (Lewis 1973, Zhang et al. 2007). Bean-shaped white eggs approximately 0.25 mm long are deposited into plant tissue, with one end protruding slightly (Capinera 2001). In the field in the southeastern United States, the egg stage lasts between 5-15 days, although at a constant 25°C, mean duration only lasts 2.6 days. Plant host and subsequent nutrition can affect the number of eggs produced. Gaum et al. (1994) reported that females produced 9-10 eggs during their lifetime when fed cucumber. This is very different from the study conducted by Trichilo and Leigh (1988) who cultured females in cotton and observed 130 eggs produced per female, and 190 eggs on cotton supplemented with pollen. Immature thrips must go through two larval instars (sometimes called nymphs), and a non-feeding prepupal and pupal stage during development (Capinera 2001). In the field in the southeastern United States, larvae require 9-12 days to develop, although this duration can increase to 60 days during the winter when it is much cooler. At relatively warm temperatures (25°C) 2.3 and 3.7 days are required to complete the two larval instars, respectively. The prepupa is identified by the presence of short wing pads and erect antennae, while the pupa has backward bent antennae running along the head and long wing pads that nearly reach the end of the abdomen. Under field conditions in the southeastern United States, the prepupal and pupal stages require 1-3 and 3-10 days, respectively. However, at a constant temperature of 25°C this duration is reduced to an average of 1.1 and 2.7 days, respectively. Both prepupae and pupae can be found on or under debris, or in cracks at a depth of 7-10 cm.

Adult *F. occidentalis* tend to have elongated, narrow yellow to brown bodies averaging 1.5 mm long, with fully-formed, fringed wings. Antennae consist of eight segments. On average, adults live for 20-30 days (Capinera 2001). Females can either mate and produce both male and female offspring, or reproduce parthenogenically, with unmated females producing mostly haploid males (Kumm and Moritz 2010). Mated females tend to produce a ratio of two females to every male (Capinera 2001). After emerging as adults, females mate over the course of their remaining lives. Adults tend to disperse when food sources become compromised, often due to drought, removal or maturity.

Many thrips, particularly *F. occidentalis*, have a broad host range, and are capable of detoxifying numerous plant toxins. Such ability to metabolize various plant toxins could allow them to be better suited compared with other species for the detoxification of xenobiotics, including pesticides (Krieger et al. 1974).

Both feeding and oviposition tend to result in deformation of plant tissues. As mentioned above, these thrips tend to prefer habitats such as floral interiors, or leaf clusters and are rarely seen in exposed locations (Capinera 2001). Feeding upon flower pollen and ovaries tends to result in fruit which is malformed and stunted. When *F. occidentalis* feeds on foliage, expanding leaves tend to become distorted and mature leaves develop mottling or speckling. Oviposition also results in deformities of fruits when the eggs are laid into fruit tissue. For example, when *F. occidentalis* lay eggs into tomatoes, an indentation or dimple encircled by a lighter circle results. In addition, *F. occidentalis* can inflict further injury on susceptible crops such as tomatoes by transmitting TSWV, resulting in wilted foliage and unmarketable, blemished fruit.

Diseases and tospoviruses transmitted by thrips

Throughout the world, thrips are responsible for severe outbreaks in a variety of crops which are estimated to cost growers billions of dollars in control costs and lost yield (Ullman et al. 1997). It is virtually impossible to control pathogens vectored by thrips because high rates of pathogen spread can be achieved by a small number of thrips. In addition, many species of thrips are not effectively controlled by pesticides due to resistance. Finally, thrips can inoculate plants very rapidly and therefore have often already spread disease by the time they are killed by treatments. Selecting for plants resistant to pathogens can reduce disease spread in some cases.

Ten species of thrips within the Thripidae are known to transmit viruses belonging to at least four groups of viruses, which include bunyaviruses (family Bunyaviridae, genus *Tospovirus*), ilarviruses, sobemoviruses and carmoviruses. Tospoviruses tend to have complex relationships with their vectors and are transmitted by thrips from leaf to leaf, while the other three classes of viruses are pollen-borne and tend to have a less complex relationship with thrips.

Interestingly, of the approximately 5500 known species of thrips, only nine species are known vectors of tospoviruses (International Symposium on Thysanoptera 2002). In addition, different species of thrips vary in their ability to transmit these viruses. Although most of these species of thrips which are known to transmit plant viruses are not closely related phylogenetically, they share some ecological characteristics including polyphagy and the ability to reproduce on a variety of host plants (Ullman et al. 1997). Another factor which allows these insects to be such effective vectors of tospoviruses is their piercing-sucking method of feeding.

Tospoviruses are a group of plant pathogens naturally transmitted by thrips, and can cause a wide range of symptoms in plants depending on the virus and the particular plant being infected. They belong to a large family of viruses called the Bunyaviridae*,* which commonly are transmitted by mosquitoes, ticks, phlebotomine flies, and other arthropods (Labuda 1991). Viruses within this family which infect plants seem to be some of the most aggressive emerging viruses, where at least five new species have been identified during the past five years and at least eight additional potentially distinct species have been proposed (Ullman et al. 1997).

TSWV is ranked as one of the top ten economically important plant viruses, making it a serious problem in a variety of crops (Goldbach and Peters 1994). Only adult thrips that acquired TSWV as larvae, often as first instars, are able to transmit the disease (Ullman et al. 1992, Tsuda et al. 1996). As the vector matures, the virus replicates in the salivary glands (Wijkamp et al. 1995b) and the viruliferous thrips spread the virus when they feed upon additional plants (Ullman et al. 1993, Kritzman et al. 2002). Adult thrips are able to disperse over long distances by wind because they are winged and very light weight (Lewis 1973), increasing their ability to spread over a wide range.

In the southern United States, *F. fusca*, and *F. occidentalis* are the two most commonly found to be associated with the occurrence of TSWV in tomato (Johnson et al. 1995, Eckel et al. 1996, Groves et al. 2001, 2002), with *F. fusca* being the most common cause of early season spread in the southeastern states (Barbour and Brandenburg 1994, Mound 1995, Groves et al.

2003, Nault et al. 2003, McPherson 2006). *F. occidentalis* may be important when locally abundant (Eckel et al. 1996), and until recently, was not a problem in the southeastern United States. Although *T. tabaci* often occurs in this region, studies indicate that most populations are not effective vectors of TSWV (Wijkamp et al. 1995a).

INSV is another tospovirus more commonly occurring in ornamental plants, particularly floral crops, which is transmitted by several species of thrips (Parrella 1995, Jones 2005). In the United States, INSV is rapidly becoming one of the most important viral pathogens (Windham et al. 2009). Included in the more than 300 plant species susceptible to INSV are African violet, ageratum, amaranth, anemone, aster, begonia, calceolaria, calendula, calla lily, Christmas pepper, chrysanthemum, cosmos, impatiens, marigold, petunia, primula, snapdragon, verbena and zinnia. Diagnosis of this disease can be variable depending upon the plant, and it may also be confused with other plant diseases, fungi, bacteria or nutritional disorders. Plants infected with this disease may be stunted, have ringspots (brown-to-purple spots on the leaves or stems), may develop brown stems, have flowers that break off of the stems, and finally may die.

Using conventional pesticides to control the spread of tospoviruses by thrips may actually assist the spread of *F. occidentalis* and subsequently the spread of TSWV (Broadbent and Allen 1995). In addition, biological control agents of thrips may also be eliminated, which could result in outbreaks. Many compounds act slowly, and/or have low efficacy, and the virus can be transmitted to additional plants by disturbed thrips before they die. Some pesticides can agitate insects initially, causing them to fly.

Thrips impact on crops in the eastern United States

Virginia is the third largest producer of fresh market tomatoes in the United States earning over 50 million dollars in the state (NASS 2008b). Very strict rules regulate tomato production and any direct insect injury or hint of disease is not acceptable.

Major crops including tomato, peanut, and greenhouse crops are commonly infected by TSWV in the southeast and mid-Atlantic states. In fact, 20% or more of tomato and peanut fields in North Carolina, Virginia and Georgia are commonly infected by this devastating disease. Between 1-12% crop loss has been estimated in peanuts in Georgia, Alabama and Virginia (University of Georgia College of Agricultural & Environmental Sciences 2003, 2006).

Peanuts are an important commodity grown in Virginia. In 2008 total peanut production in the state of Virginia amounted to 35,924,515 kg (NASS 2008a). Although they are sought after by customers due to their size and flavor, they require high input costs and can succumb to insects and diseases. Adult thrips are attracted to the seedlings where they promptly oviposit, and the resulting larvae concentrate their feeding on the delicate unopened leaflets. As in cotton, such feeding results in plant deformation as well as delayed maturity. In Suffolk, VA in 2007, three trials carried out at the Tidewater Agricultural Research and Extension Center (TAREC) showed that unprotected peanut plants lost a staggering 629 kg, 447 kg and 401 kg per hectare, respectively (D.A. Herbert, unpublished data).

Numerous floricultural crops grown in greenhouses are also damaged by thrips, particularly members of the genus *Frankliniella*, with *F. occidentalis* often causing the greatest amount of crop injury (Parrella 1995). Plants become scarred and distorted as a result of thrips feeding. Various diseases may also be spread among floriculture crops including the tospoviruses TSWV and INSV (McDonough et al. 2009). In 2005, floriculture sales in Virginia were worth $87,378,000 (USDA/NASS Virginia Field Office 2007).

Thrips are some of the most serious cotton pests in Virginia and North Carolina, even though thrips do not vector TSWV in cotton. Since 2001, field tests carried out in Suffolk, VA have shown thrips injury to unprotected cotton can reduce lint yields up to 882 kg per hectare, which is 50% of the total yield potential (Herbert et al. 2007). Although thrips emergence is usually delayed due to prolonged cool springs, as temperatures warm, adults migrate into the fields where they feed and lay eggs. The population surge, which results in hatching larvae does the greatest damage to seedlings. Feeding occurs primarily at the seedling bud, and growth can be stunted or plants may become deformed, resulting in compromised final structure of the plant. If damage is extensive enough, seedlings may even die.

Monitoring methods

Finding effective sampling methods for monitoring thrips is becoming increasingly necessary because these insects are responsible for extensive crop damage. In doing so, pesticides can be applied only when populations pose the greatest risk to crops, which will reduce unnecessary use of sprays and therefore excessive contact with the pest which rapidly leads to resistance. Some species of thrips have already developed resistance to a wide range of pest-control products including commonly applied organophosphates, carbamates and pyrethroids (Bielza 2008). They have also become resistant to a handful of insecticides outside of these classes including abamectin (Immaraju et al. 1992), DDT (Zhao et al. 1995), and spinosad (Bielza et al. 2007).

There are a myriad of reasons to monitor for thrips in crops including detection of their initial presence, locating areas in crops considered “hot spots”, predicting outbreaks of disease, determining the timing of control measures, and then to assess the effectiveness of the implemented control measures (Shipp 1995). For growers to be able to reduce their spray applications by using pesticides only when it is necessary to prevent thrips damage, effective and accurate sampling methods are essential. Currently, there are a variety of sampling methods available to growers including sticky cards, pan traps, plant tappings, collecting whole plants or various plant parts such as leaves or flowers. Such sampling methods reduce the amount of labor required for direct sampling methods such as counting individual thrips on plants.

When considering different monitoring and sampling strategies, it is also important to evaluate various aspects of the insect such as dispersion tendencies. For example, *Frankliniella* spp. often reside in flowers as opposed to the foliage or stems of plants, and therefore it is necessary to sample from these plant organs when they are present to obtain an accurate assessment (Lewis 1997c). Temperature can affect sampling as insects are more active at elevated temperatures. Insects could be more likely to escape before capture can take place. Time of day when samples are taken can also have an effect on counts of insects if they are more prone to diurnal activity.

There are several methods available for trapping flying thrips in order to monitor for presence and population fluctuations (Lewis 1997c). Both sticky cards and water traps are widely used to monitor thrips. Pan traps can be colored to increase thrips catch with polyphagous species responding positively to yellow, blue and white, probably because these are the colors commonly associated with their hosts. Sticky cards are advantageous for sampling because they are cheap and fairly easy to inspect. However, if identification to species is necessary, such determination can prove difficult if critical anatomical features are attached to the card and therefore not observable. In addition, a large amount of time must be invested in the counting of thrips captured by the cards (Shipp 1995). McPherson and Riley (2006) have shown that yellow sticky cards are an effective monitoring tool for thrips populations in newly planted tobacco fields. Yellow sticky cards have also been shown to accurately monitor *F. occidentalis* populations within greenhouses (Higgins 1992) and are commonly used (Shipp 1995). Higgins (1992) found trap catch in greenhouses correlates well with the number of adults and immature stages on plants.

Colored pan traps allow for the easy identification of thrips because their bodies can be maneuvered beneath a microscope without the inhibition of sticky glue. Detergent added to the collection water breaks the surface tension and prevents thrips from drifting to the edge of the pan and escaping (Lewis 1997c). The number of thrips collected by pan traps, in addition to the above mentioned sampling methods, are affected by different wind speeds, as well as the height at which they are placed (Lewis 1997c), and this needs to be taken into account when assessing data.

Thrips chemical ecology

Many thrips exhibit olfactory responses to several plant volatiles, as well as pheromones. Male *F. occidentalis* have been found to produce an aggregation pheromone, attracting both male and female thrips (Hamilton et al. 2005). The two main male-specific components of this pheromone were identified as (R)-lavandulyl acetate and neryl (*S*)-2-methylbutanoate, with the latter showing activity in field trials. In a previous study by Kirk and Hamilton (2004), this pheromone was identified as a sex pheromone, but it has since been shown to attract thrips of both sexes.

Recently, research has shown that many thrips are also attracted to various plant-produced volatiles (kairomones), particularly odors given off by flowers within the chemical class of benzenoids and monoterpenes (Koschier et al. 2000). Examples of these compounds include geraniol, linalool, germacrene, benzaldehyde and eugenol (Terry 1997). Attractive compounds such as methyl anthranilate and ethyl nicotinate have been recently synthesized into products including Thrips Lurem-TR (Koppert Biological Systems, the Netherlands) and $\text{Thripline}_{\text{AMS}}$ (Syngenta Bioline Ltd., United Kingdom). The latter contains an aromatic substance meant to attract a wide variety of thrips including *F. occidentalis,* and onion thrips, *Thrips tabaci* Lindeman. Coupling attractive lures with attractively colored yellow or blue sticky cards and pan traps can enhance sampling and allow for better detection of thrips.

Kirk (1985) found that anisaldehyde was particularly attractive to five species of flower thrips, whereas three species of cereal thrips and a predatory flower thrips, *Aeolothrips intermedius* (Bagnall), showed no response to the scent. These results support the hypothesis that there is a relationship between the type of host and the response to the volatile chemical anisaldehyde. The thrips that were found in the traps can be found in a variety of flowers because they are generalists. Such generalists may respond to numerous scents associated with flowers, and anisaldehyde could be one of these compounds generating a positive response. In a similar study, Teulon et al. (1993) found that the addition of anisaldehyde, benzaldehyde, and ethyl nicotinate to traps increased the capture of several generalist flower thrips including *Thrips fuscipennis, T. obscuratus, T. tabaci, Frankliniella intonsa,* and *F. occidentalis,* but not cereal and grass-inhabiting thrips *Anaphothrips obscurus* and *Limothrips cerealium.*

In a separate study, Hollister et al. (1995) also found that *p*-anisaldehyde increased yellow pan trap catches of *F. occidentalis* more than ten-fold over non-baited traps. There was also evidence of baited trap interference with the non-baited traps nearby.

A combination of both attractively colored sticky cards and attractive odors has been found to increase catches in certain settings (Frey et al. 1994). Combining cues increased attractiveness approximately 2-fold beyond that brought about by either cue alone in the laboratory. However, these laboratory results could not be reliably reproduced in a greenhouse setting and only a moderate increase of 26% in catches was reached. Possibly, the difference in efficacy could be due to differing release rates of the attractant geraniol due to temperature and/or humidity changes. Air movement can also be a factor affecting the buildup of odors surrounding the baited traps. Finally, odor concentrations could have been too low to elicit a response, or they could have been too high, causing them to act as repellents.

The concentration of various scents is an important factor determining attractiveness. Numerous chemicals have been found to improve catches at low concentrations, but decrease them at higher concentrations (Snapp and Swingle 1929). Kirk (1985) found that myrcene reduced thrips catches at the concentrations that were used for this experiment. However, at lower concentrations it could potentially improve catches.

Insecticides and resistance

Thrips infesting crops are quite responsive to cultural control methods. However, when these practices are neglected, or if crops have been drastically weakened by dry weather when they would otherwise be able to resist infestations, it may be necessary to resort to insecticides to reduce damage (Lewis 1997b). Treatment with insecticides also can become necessary when small populations spread viruses, or in an environment favoring rapid increase of thrips populations, such as greenhouses, where a population can reach astounding numbers within a short amount of time. It is very clear that when insecticides are used as part of an IPM program, multiple types should be applied when necessary. Relying on a single insecticide for control is becoming less reliable as thrips gain resistance against such treatments (Lewis 1997b, CAST 2003).

Developing sprays in the lab to be applied in the field can be very challenging for a variety of reasons. Although many insecticides can kill thrips when tested in the lab, it is much more difficult to treat large populations in the field due to additional factors not experienced in the lab. Large numbers of thrips can infest plants and because they tend to have cryptic habits, many will conceal themselves in flower crevices or leaf sheathes, thereby evading insecticide treatments. Due to such traits, non-persistent systemic or translaminar insecticides tend to be more effective against thrips than sprays, which would not reach such areas. Populations can also build extremely rapidly in the field under optimal conditions, and more thrips can immigrate into crops. Application methods, pesticide persistence, previous treatments, and weather can also affect pesticide efficacy (Parrella 1995). Vulnerability to treatments may be different for thrips that spend a portion of their lives in the ground compared to those remaining above ground (Lewis 1997b). It is important to coordinate spray treatments with thrips life cycles and to time sprays with the stages that are most vulnerable. Nonetheless, resistance is constantly developing against insecticides, and there is a constant struggle to develop new treatments as the old ones become ineffective.

Prior to the mid-1940s, a wide range of plant derivatives and inorganic and organic compounds were tested against thrips (Lewis 1997b). They included such compounds as kerosene, whale oil soap, crude carbolic and creosote, calcium cyanide, sulfur dust and lime sulfur, quicklime, and pyrethrum, among others. Persistent chlorinated hydrocarbons, primarily toxaphene, dieldrin, DDT, BHC and aldrin were used from 1945 to 1960. Such treatments tended to have a better success rate against leaf-feeding thrips, which did not feed in crevices, but inhabited plants that had a more open form of growth.

The drawback to using chlorinated hydrocarbons was their tendency to kill natural enemies. Even when they did succeed in killing the pest, populations could easily rebound when no natural enemies were present to reduce new populations immigrating into the crop.

In time, chlorinated hydrocarbons were replaced by semi-persistent systemic insecticides, primarily carbamates and organophosphorus derivatives, for control of thrips in crops. Some of the most effective treatments included parathion, ethion and carbophenothion dusts (Lewis 1997b).

Eventually formulations of systemic insecticides such as aldicarb and phorate were developed as seed dressings and granules. Applications of these insecticides protected seedlings during the first few weeks after emergence, which is when they are most susceptible to thrips attack and subsequent damage.

Currently directly-seeded crops such as peanut and cotton are commonly treated with prophylactic applications of systemic insecticides to control *F. fusca* instead of applying foliar insecticides as needed. This is because significant injury occurs rapidly when thrips populations suddenly invade crops and therefore there is a very limited amount of time to apply foliar insecticides. Both pre-plant applications of systemic insecticides and foliar applications may be required during years of extremely high thrips populations. Foliar insecticides may be the only required treatment during years of moderate thrips populations, and insecticide applications are not necessary when thrips are absent. Growers often assume that thrips populations will be high, and therefore apply the maximum amount of insecticide permitted because it is very difficult to predict the threat that thrips pose from year to year.

In cotton and peanuts grown in the southeastern United States, in-furrow application of aldicarb (Temik 15G, Bayer CropScience) or phorate (Thimet 20G, AMVAC Chemical Corporation) and/or early season foliar applications of acephate (Orthene 97, AMVAC Chemical Corporation) are used for traditional thrips management. Although it is a very effective control method, and has been shown to reduce both direct thrips injury to seedlings as well as incidence of spotted wilt in peanut (Herbert et al. 2007), concerns about toxicity continue to mount. Such concerns have been expressed by the Pest Management Strategic Plans for Cotton in the Midsouth (December 2003) (safe handling of insecticides) (Anonymous 2003), and tomatoes grown in Delaware, North Carolina and Virginia (June 2006) (evaluation of IPM-compatible pesticides) (Anonymous 2005). Investigating the role of cultural practices in reducing TSWV have been placed on a “To Do” list for peanuts grown in North Carolina and Virginia by the Pest Management Strategic Plan (April 2002) (Anonymous 2002).

In addition to concerns about toxic effects, there is also concern for the development of resistance (Robb et al. 1995, Dagli and Tunc 2008). Throughout history, many once-effective insecticides have gradually lost their ability to control thrips populations as these insects have developed resistance. Development of resistance can become evident within as little as a few years, as in the cases of DDT and dieldrin, which lost efficacy within a mere four years (Lewis 1997b). But even if insecticides do remain effective for a longer period of time, eventual failure is almost certain, and heavier reliance upon a single chemical only serves to reduce the amount of time that treatment is effective.

It is likely that the ability of certain species of thrips to develop resistance to various insecticides is due to their ability to metabolize and excrete toxins from their bodies very effectively. It has been shown that strains of *F. occidentalis,* which are more resistant to organophosphates such as diazinon, are able to metabolize and excrete the pesticide more rapidly than susceptible strains. Also, rapid oxidative metabolism combined with insensitive acetylcholinesterase gives them the ability to resist treatments (Liu et al. 1994, Zhao et al. 1994). In addition, butyrylcholinesterase may act as a scavenging enzyme, which helps provide thrips with some protection to anticholinesterase insecticides.

Another factor that may predispose highly polyphagous species of thrips such as *F. occidentalis* to rapidly developing resistance to xenobiotic compounds such as insecticides, is the fact that they are able to detoxify a myriad plant compounds (Lewis 1997b). High fecundity and short life cycle are other traits that greatly increase the ability of thrips to develop resistance. Understanding resistance mechanisms in field populations will likely help in choosing the most effective combinations of pesticides and synergists to slow development of resistance to various treatments. In addition, rotating treatments or mixing unrelated compounds also helps to slow resistance development.

Due to such heavy reliance on acephate (Orthene 97) for managing thrips in peanut and cotton, selection for resistance is high. Although very successful in controlling *F. fusca*, acephate is less effective against other thrips species, particularly *F. occidentalis,* which is a more recently-arrived pest in the south. Acephate (Orthene 97) applied at a rate of 0.88 L/ha only provided about 50% control of *F. occidentalis* in a cotton field test carried out in 2007 in Suffolk, VA (Herbert 2007, unpublished data).

Heavy reliance upon a very limited range of insecticides leads to a more rapid development of resistance to treatments; therefore, it is far more prudent to rely more upon other methods such as cultural control, and to only use insecticides when other methods fail. However, this often requires more work in the form of monitoring to determine the optimum time for treatments and properly maintaining crops. Therefore, monitoring is usually less desirable, despite the fact that the excessive use of pesticides is far less sustainable (Lewis 1997b). Treatment of refugia as well as other nontarget plants should also be avoided so that strains of thrips that are susceptible to treatment can dilute the populations on crops that are under very heavy selective pressure due to repeated contact with treatments.

Some insecticides, particularly pyrethroids, can actually be responsible for the buildup of *F. occidentalis* populations by several mechanisms. The suppression of key predators such as *Orius insidiosus* (Say) is one of the leading factors (Funderburk et al. 2000), although other mechanisms such as competition for resources (Paini et al. 2007) and even hormoligosis have also been attributed to an increase in pest populations (Frantz and Mellinger 2009).

Integrated pest management (IPM)

IPM is a form of pest management which tends to be more environmentally sound than many conventional practices. This strategy uses information on the life cycles of pests and their interactions with the environment in combination with various pest control methods to manage pest damage as economically as possible, while minimizing any negative impacts upon other organisms and the environment (U. S. Environmental Protection Agency 2009). IPM is used in both agricultural and non-agricultural settings and consists of four different steps in its approach: setting action thresholds, monitoring and identifying pests, prevention, and finally control.

The economic threshold is the point when pest populations or environmental conditions indicate that action must be taken to control a pest and prevent populations from reaching the economic injury level (Parrella 1995). This threshold tends to vary greatly depending on crop growth stage, pest, season, climate, tolerance for damage, as well as the market. In certain crops such as ornamentals, no damage is tolerated, which often results in the heavy use of pesticides.

Simply sighting a single pest is not necessarily an indication that the pest is going to cause economic damage to a crop. It is also very important to properly identify organisms in the field because some are innocuous or beneficial, such as natural predators. Properly identifying and monitoring pests reduces the likelihood of applying pesticides when they are not necessary, or applying the wrong treatment.

Before a pest becomes problematic, a first line of defense against an outbreak is to properly manage the crop. There are a variety of cultural methods that can be used to achieve this goal including crop rotation, time of planting and harvesting, selection of pest-resistant varieties, planting healthy rootstock free of pests, promoting rapid plant growth and keeping crops clear of weeds (Parrella 1995, CAST 2003, U. S. Environmental Protection Agency 2009). In addition, intercropping, planting trap crops, refugia, cover crops and also antagonistic plants can help enhance soil and foliage-inhabiting beneficial organisms (Parrella 1995, CAST 2003). Physical control strategies can also be utilized (Jacobson 1995). This form of non-chemical control uses barriers, screens, traps, etc. to help control pests.

Pesticides may be used when other techniques for controlling populations fail (U. S. Environmental Protection Agency 2009). Less risky, but effective pest controls are selected first. These include mating disruption from chemicals as well as mechanical control, which can be in the form of weeding or trapping. Natural enemies or biological control agents can also be used against pests. Such control agents include parasitoids, predators, pathogens, entomophyllic nematodes as well as gene products, which are derived from living organisms that kill, disable, or alter the behavior of pests or biological control products (CAST 2003). If controls are not effective in controlling the pest, then techniques such as targeted pesticides may be sprayed. Non-specific pesticides are only used when all other control measures fail.

It is also important to maintain areas around crops because thrips can easily move from nearby plants into the crops, thereby beginning a new infestation. Many species of weeds are known to harbor thrips and can also serve as a reservoir for tospoviruses; therefore, proper crop hygiene and weed control in and nearby crops is essential (Parrella and Lewis 1997). It is also important to understand that thrips can develop in adjacent crops or fields and then migrate into susceptible crops.

Natural enemies can be very important in controlling pests and can either be encouraged to inhabit a crop, or they may be introduced using inoculative/ inundative procedures (Parrella 1995). Biological control is obviously not compatible with intense use of pesticides for thrips control. This is another reason why it is important to limit pesticide applications, and to rely upon sprays that target pests when chemical control is required.

IPM is used on several crops in greenhouses to control thrips. Sticky card placement and examination and visual inspection of the plants are used to monitor thrips populations. For many years foliar applications of spinosad (Conserve SC, Dow AgroSciences LLC, Indianapolis, IN) have been applied, while more recently chlorfenapyr (Pylon, OHP, Inc., Mainland, PA) and the reduced risk insecticide pyridalyl (Overture, Valent U.S.A. Corporation, Walnut Creek, TX) are being used. Spinetoram (Radiant SC, Dow AgroSciences LLC, Indianapolis, IN) was the most effective product in controlling *F. occidentalis* in a field test conducted in cotton, as well as tomatoes in 2007 (Herbert 2007, unpublished data).

As pesticides are removed from the market due to the development of resistance by pests, and for commercial purposes, new control methods must be sought (Jacobson 1995). The development of new pesticides is very expensive and a large amount of time is required to develop and adequately test new products to insure safety. Some insecticides can also be phytotoxic to plants, which reduces their attractiveness to farmers. Consumer demand is also rising for crops raised with reduced amounts of pesticides as people become more aware of the potentially negative effects of pesticide usage.

As part of an IPM tactic, biologically derived insecticides are great alternatives to the traditional insecticides, which thrips rapidly become resistant to due to overuse. However, as with any form of chemical control, these also must be used in a strategic manner and a single insecticide should not be relied upon for pest control.

Plant-produced secondary metabolic compounds

Some plants produce secondary metabolite volatile compounds to attract specific insects to pollinate or to rid the plant of certain animals that may feed upon the plant, while others produce bitter tasting compounds that deter feeding (Breitmaier 2007). These can render the plant distasteful, or they may even be toxic to the animal feeding upon the plant. Terpenes and terpenoids are two classes of chemicals produced by plants, which are repellent and even toxic to certain insects, including thrips. These chemicals disrupt the insect's neurotransmitters by interfering with the neuromodulator octopamine (Enan 2001, Kostyukovsky et al. 2002). In addition, some oils interfere with GABA-gated chloride channels (Priestley et al. 2003). Plants that produce such compounds include rosemary, mint, and many other strongly scented plants. In general these compounds exhibit very little toxic effect on mammals, birds and fish because they do not share the same target sites for these compounds (Isman 2004, 2006). Unfortunately toxicity can also be experienced by beneficial insects such as pollinators and natural enemies.

In testing a variety of plant-derived essential oil products, Cloyd et al. (2009) found that many of these products consisting of oil combinations such as cottonseed, cinnamon, and rosemary oil, were effective against several pests. Ecotec AG (EcosSMART Technologies, Inc.) is a blend of wintergreen, peppermint and rosemary oil and is a USDA organic-approved insecticide that could be considered another tool for use against thrips because it is marketed for the control of a wide variety of insects including thrips.

Pyrethrins

Pyrethrins are a group of organic compounds produced by chrysanthemum flowers, *Chrysanthemum* spp., which exhibit insecticidal activity (Casida 1980). Three species of *Chrysanthemum* are recognized by the USDA as suitable for use in manufacturing insecticide powder; they are *Chysanthemum cinerariaefolium, C.* (synonym, *Pyrethrum carneum*), and *C. roseum,* with *C. cinerariaefolium* being the most commercially important (Gnadinger 1933, Head 1973). Although the entire plant contains pyrethrins, the daisy-like flowers possess the greatest concentration of these compounds.

Pyrethrins have been used for many years as a form of insect control. Pyrethrum flowers were used by Caucasian tribes in Persia in the early 1800's to control body lice (Casida 1980). Over the years, commercial production has shifted from Armenia, where it first began in 1828, to Dalmatia (now Croatia) around 1840, to Japan, and finally to East Africa in the 1940s

(McLaughlin 1973). In 1860, pyrethrin powder to be used against insects was imported into the United States. Numerous improvements in techniques for shipping, extraction and production and the development of an international aerosol industry, led to an increase in demand for pyrethrum. However, the development of various synthetic insecticides significantly reduced the need for pyrethrum in agriculture because the newer compounds were cheap and effective. Twenty years later, pyrethrum popularity began to escalate again with the development of resistance to many popular insecticides, as well as recognition of their deleterious effects. There were additional improvements in application methods for pyrethrum, which led to a decrease in cost. As new formulations that are more resistant to degradation and application methods improve, pyrethrum may regain its popularity.

The pyrethrins are located in the achenes where they are isolated from feeding insects as well as photodecomposition (Casida 1980). Once the flowers are mature (when four or five rows of disc florets are open), they are hand-picked, dried and then ground and extracted with hexane or another suitable solvent. The concentration is then diluted or purified to remove components, which can cause dermatitis in humans sensitive to pollen from ragweed.

The pyrethrins possessing the insecticidal activity in pyrethrum extract consist of three alcohols: pyrethrolone, cinerolone, and jasmolina and two acids: chysanthemic acid and pyrethric acid (Head 1973). They cause rapid knockdown and eventual death of pests susceptible to these esters (Katsuda 1999). Pyrethrins are contact poisons that affect the insect nervous system by delaying closure of ion channels, thereby causing multiple action potentials in nerve cells (National Pesticide Information Center 1998). In addition, insecticidal activity can be enhanced by the addition of synergists, such as piperonyl butoxide, which reduce detoxification in the insect (Elliott and Janes 1973). Other synergists can be used to slow photodegradation, and pyrethrins can also be combined with many other insecticides (Lange and Akesson 1973).

As with any compound used as a poison against organisms, there is concern for toxicity in nontargets, particularly animals and humans. Pyrethrins have been found to be one of the least poisonous insecticides to mammals, although they can cause skin irritation as well as coughing, wheezing, runny or stuffy nose, chest pain, or difficulty breathing when inhaled (Barthel 1973, National Pesticide Information Center 1998). Skin irritation rapidly disappears upon removal from exposure. Pyrethrins are not very toxic to animals and humans because they are rapidly broken down and excreted once inside the body. There is no evidence to indicate that pyrethrins cause chronic problems of toxicity, nor do they appear to pose a hazard of acute toxicity to humans. However, pyrethrum is toxic to fish and many organisms eaten by fish such as crustaceans and aquatic insects (Pillmore 1973). Bees and many natural enemies do not appear to be adversely affected by pyrethrins (Lange and Akesson 1973). Pyrethrins are not very stable in the environment, and are subject to rapid photodegredation.

Neem- Azadirachtin

Azadirachtin is a triterpinoid, which can be found in three species of neem tree *Azadirachta indica* (Rutales: Meliaceae), *A. excelsa,* and *A. siamensis* (Morgan 2009). The neem tree is closely related to mahogany and is grown in arid zones in Africa, many Asian countries, in tropical regions of the new world, and in some Mediterranean and Caribbean countries (Koul 2004). This fairly hardy tree can grow well even in nutrient poor soil and tolerates high temperatures, drought and salinity (Saxena 2004). It produces masses of pleasantly scented flowers and is very fast growing, capable of reaching a height of 25 m in some areas in Africa.

Various bioactive compounds can be harvested from different parts of the tree including the bark, leaves and seeds which have potential for use in areas ranging from agriculture to regulating human fertility. Of particular interest to agriculture are the various impacts azadirachtin can have on insect pests. This part of the tree is considered to be the most important because most of the biologically active materials are found in the seeds, with concentrations of 0.1 to 0.9 percent azadirachtins being contained within the seed core (Koul 2004).

Although neem has had quite a history as an insecticide primarily used against household and storage pests and against some crop pests in the Indian sub-continent, it was not until the 1960s that the modern study on the effects of crude neem seed extracts on crop pests really began (Morgan 2004). Neem is a fairly new pesticide, and the potential of neem as a form of pest control has only been realized in the past few decades (Koul 2004). In 1982 azadirachtin was identified by Kubo and Klocke (1982) as an antifeedant and also capable of preventing the completion of larval molting of four agricultural pest insects.

Neem's subtle effects are considered much more favorable than a quick knockdown in an IPM program because they reduce the risk of adversely affecting natural enemies (Koul 2004). Neem as a biopesticide has many advantages: neem is very effective against a variety of pests, due to its different mode-of-action, there is less risk of pests developing resistance because neem contains several different compounds, it is much safer for nontarget organisms, it is biodegradable and also can be obtained easily from a renewable resource.

Neem derivatives affect more than 400 species of insects belonging to several groups of insects including Blattodea, Caelifera, Coleoptera, Dermaptera, Diptera, Ensifera, Heteroptera, Homoptera, Hymenoptera, Isoptera, Lepidoptera, Phasmida, Phthiraptera, Siphonoptera, Thysanoptera, some ostracods and several different mites. Neem can also act as a nematicide, and against some fungal pathogens. One reason neem can affect insects such as thrips and other insects which feed upon plants by sucking is due to the fact that it can be taken up by plants from the soil and translocated to the leaves and growing tips; therefore neem has systemic action. Current ready-to-use neem biopesticides are emulsions of oil and water with azadirachtin as the active ingredient, vegetable oils, detergents and stabilizers. Neem also can be mixed with some synthetic insecticides, thereby enhancing their action.

Neem has a wide array of effects upon insects susceptible to the compounds including repellency, primary antifeedancy (detection of deterrent compounds by insect taste receptors) and secondary antifeedancy (toxic effects resulting from ingestion), growth reduction, increased mortality and abnormal molts (Seymour et al. 1995, Puri 1999, Abudulai et al. 2003, Durmusoglu et al. 2003, Luntz 2004). These compounds can also disrupt mating and sexual communication, deter oviposition, and cause sterility in adults. In insects, ecdysone production is inhibited in several species susceptible to these compounds. Such activity prevents the production of chitin and also disrupts the molting process (Ladd et al. 1978).

Insects in various orders differ in their response to azadirachtin at the feeding deterrence level (Luntz 2004). Some insects, particularly many lepidopterans and some orthopterans, are unable to ingest the compounds once they have been detected. This reaction is referred to as "primary" or gustatory antifeedancy. Feeding inhibition is caused by the stimulation of specific 'deterrent' cells in chemoreceptors and blocks 'sugar' receptor cells from firing, which normally stimulate feeding. Starvation and eventually death is the result in susceptible species because insects are unable to feed.

Some insects, such as hemipterans, are less susceptible to the primary antifeedant effects of azadirachtins and as a result will consume enough of the compound to experience physiological effects including secondary antifeedant effects. These effects include a reduction in food consumption as a result of ingestion resulting from the disruption of physiological and/or hormonal systems. For example, aphids have been shown to experience suppressed feeding activity in subsequent feedings as a result of feeding upon tobacco seedlings treated systemically with azadirachtin (Nisbet et al. 1993).

Azadirachtin can have a variety of effects on growth and molting in insects including abnormal and delayed molts, reduced growth and increased mortalities (Luntz 2004). Such effects result from a disruption between the molting hormone (20-OH ecdysone) and juvenile hormone (JH) during the molt. The disruption occurs when morphogenetic peptides responsible for the release of hormones from their endocrine glands are blocked from being released from the brain. Haemolymph ecdysteriod levels are thereby modified, which then causes the various insect growth regulatory effects previously described. In addition, altered JH titres cause slight changes in cuticullar structure.

Azadirachtin has been shown to affect reproduction in both male and female insects. Due to disruption in JH titers and ecdysteroid production, vitellogenin synthesis and uptake by the eggs is disrupted, which results in reduced fecundity and sterility. Oviposition deterrence can also occur in females ready to lay eggs (Puri 1999). Testes development and sperm production in adult male desert locusts injected with azadirachtin have been shown to be inhibited.

Insect cells grown in culture also have been shown to be affected by azadirachtin. Cells are prevented from proliferating when subjected to azadirachtin and also experience toxic effects. Such effects have been demonstrated in a variety of organisms. Following treatment with azadirachtin, Nasiruddin and Luntz (1993) showed that locust midgut epithelial cells experienced cell necrosis and a reduction in the number of regenerative cells.

Protein synthesis is inhibited by azadirachtin in various tissues such as midgut cells where enzymes are being produced. Annaduraie and Rembold (1993) showed that azadirachtin has differential effects on protein synthesis in various regions of the brain of *Schistocerca gregaria* (Forskal). Some protein bands remained the same while others disappeared following treatment with azadirachtin. Possibly azadirachtin is affecting protein synthetic machinery on the transcriptional level when it is switched on to perform a specific function.

In addition to containing many different active compounds, azadirachtin affects insects through more than one mode of action, which further reduces the likelihood of developing resistance. Azadirachtin disrupts the process of cell division, inhibits formation of spermatozoa, and blocks the release and transport of neurosecretory peptides by altering or preventing the assemblage of new organelles or cytoskeleton. It also inhibits metabolically active cells such as midgut cells meant to produce large amounts of protein from synthesizing protein.

Although neem is a renewable resource and the seeds can be harvested easily, processing and extraction of the active ingredients is fairly expensive, which is a drawback to azadirachtin (Koul 2004). Fortunately, advancements in technology are seeking to develop less expensive methods for extraction.

Azadirachtin is less toxic to natural enemies and mammals, and tends to be selective towards phytophagous insects (Jackai et al. 1992, Abudulai et al. 2004, Koul 2004, Weathersbee and McKenzie 2005, Isman 2006), which are often beneficial qualities in an insecticide used in IPM. However, pesticides derived from neem are poisons, and tend to be very effective against a variety of pests; they should therefore be treated with respect (Raguraman et al. 2004). Also, some organisms are very sensitive to neem, and this should be considered when their use is employed as it may affect nontargets and beneficials, although many adult insects including earwigs, crickets, true bugs, beetles, lacewings and wasps appear to show no or relatively low sensitivity to this compound.

Bacterial-produced toxins

Spinosyns, a group of insecticidal macrocyclic lactones, are the fermentation product of *Saccharapolyspora spinosa*, which is a soil actinomycete (Horowitz and Ishaaya 2004). Activity against a wide variety of pests, particularly lepidopterans, thysanopterans and dipterans has been shown, and they tend to have a low impact upon the environment and low toxicity towards many nontarget species. Spinosyns act upon the insect by exciting neurons in the central nervous system, which causes tremors and spontaneous muscle contractions, paralysis and a loss of body fluids, resulting in flaccid paralysis (Thompson et al. 2000). Such effects are consistent with the activation of nicotinic acetylcholine receptors, as well as GABA receptors, which may further contribute to insecticidal activity (Horowitz and Ishaaya 2004).

Spinosad is a mixture of two different spinosyns, A and D (Horowitz and Ishaaya 2004). This insecticide is quite effective against a variety of pests and was first introduced to the market by DowElanco. Spinosad is particularly active against insects in the orders Lepidoptera, Diptera, Hymenoptera, a few Coleoptera, and Thysanoptera (Leader and Dutton 2002). It is also able to control *F. occidentalis* (Jones et al. 2005, Broughton and Herron 2009), which is particularly resistant to many of the traditionally used conventional treatments. Although it does exhibit some contact activity, spinosad acts primarily through ingestion. Insect pests hidden among foliage are also affected due to the translaminar activity of spinosad. In an experiment conducted by Cloyd et al. (2009), a commercially available product called Monterey Garden Insect Spray

(Monterey Lawn and Garden Products, Inc., Fresno, CA), which contained a 0.5% spinosad solution, resulted in 100% mortality of *F. occidentalis*.

A test conducted on highbush blueberries in 2006 showed that spinosad (SpinTor, Dow AgroSciences LLC) significantly reduced the number of *F. tritici* compared to the untreated check and provided a control level of 72% (Rodriguez-Saona and Holdcraft 2006). Spinosad (SpinTor) has also been shown to control the more-difficult-to-manage *F. occidentalis* and *F. fusca* (Kuhar and Doughty 2008). In a trial conducted in snap beans in 2008, spinosad provided effective control of *F. occidentalis*, *F. tritici* and *F. fusca* compared with the untreated check. In addition to being an effective control agent against thrips, spinosad is less harmful to numerous natural enemies than many conventional insecticides (Jones et al. 2002), which allows their populations to build and therefore reduces the need for as many treatments as would be required in the absence of natural enemies (Reitz et al. 2003, Jones et al. 2005).

Spinetoram is derived from naturally occurring spinosyns J and L, which have been modified to produce a semi-synthetic insecticide (Sparks et al. 2008, Huang et al. 2009). This insecticide has a longer control period and also is more active against many key pests including codling moth *Cydia pomonella* (L.), and tobacco budworm *Heliothis virescens* (Fabricius). In addition, spinetoram has been shown to control thrips populations. In a trial conducted on staked tomatoes in 2007, spinetoram (Radiant SC, Dow AgroSciences LLC) significantly reduced *F. occidentalis* populations below the control two days following treatment (Walgenbach 2007). The mammalian toxicity, ecotoxicity and environmental fate characteristics of spinetoram are very similar to spinosad because it is derived from the same bacterial fermentation product.

Natural Enemies

As part of an IPM program, it is essential to protect and encourage natural enemies. It is important to carefully select pesticides when they must be used that have little to no adverse effects upon predators of the pests being targeted. One group of important biological control agents against many species of thrips are the voracious predators in the genus *Orius* (Hemiptera: Anthocoridae).

Several types of predators have been shown to be effective biological control agents against thrips. *Orius* spp., have been shown to be particularly successful as natural enemies in many crops. These voracious predators feed primarily upon soft-bodied insects including mites, thrips, aphids, leafhoppers and eggs and young larvae of various Lepidopterans. They also utilize pollen as an alternative food source. A few characteristics which make this insect such an efficient predator include its preference towards habitat inhabited by thrips, its ability to survive upon alternative food sources when its primary prey are absent, and it is also fairly easy to mass produce (Silveira et al. 2004). These insects are very beneficial as part of an IPM program, and it is essential to be aware of the deleterious effects which can occur as a result of many generalist pesticides applied to crops. Such pesticides tend to knock out natural enemies including *Orius* spp.

Overwintering weeds of thrips

In the southeastern United States some thrips overwinter in numerous weed species as well as volunteer crop plants and migrate into newly planted crops during spring and early summer (Chamberlin et al. 1992). Thrips harboring viruses such as TSWV will infect the new plants in the spring as they migrate into the crops (Takacs et al. 2008). Summer annual weeds may also assist with the cycling of TSWV in crops (Groves et al. 2003). In North Carolina, thrips surveys performed in tobacco, tomato, and pepper documented populations of *F. fusca, F. occidentalis,* and *T. tabaci* peaked between mid-May through early June (Eckel et al. 1996). It is important to understand such seasonal oscillations to better understand the epidemiology of TSWV because populations of viruliferous thrips likely develop on nearby weed hosts and then disperse into crops in the late spring (Johnson et al. 1995, Groves et al. 2001, 2002). In Maryland, chickweed appears to harbor the greatest proportion (70%) of thrips with wild mustard and henbit being the next most attractive (Brust 2008). While weeds can provide refuge for overwintering thrips, they may also serve as reservoirs of diseases, which spread with the movement of the vectors into crops, as well as reproductive hosts for vectors (Groves et al. 2001, Takacs et al. 2008). In understanding which weeds are likely to harbor thrips, growers will be able to more effectively forecast thrips infestation and anticipate potential damage.

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Chapter 1: Determining the relative abundance of *Frankliniella occidentalis* in thrips populations in weeds and selected agroecosystems in eastern Virginia

Abstract

In the eastern United States there are a myriad of species of thrips known to cause damage to crops both directly through feeding and oviposition, as well as indirectly by transmitting tospoviruses. Both *Frankliniella tritici* (Fitch) and *F. fusca* (Hinds) have been identified as the key pests in many crops in the eastern United States. In 2007, *Frankliniella occidentalis* (Pergande) was found for the first time in significant densities Virginia infesting several crops. While all three thrips species can cause devastating damage to crops when populations are high, both *F. fusca* and *F. occidentalis* can also transmit some very important tospoviruses. *F. occidentalis* is particularly difficult to manage due to its resistance to several commonly-used insecticides, further complicating control and increasing the urgency to determine whether this species is now established in Virginia. In 2008 and 2009, early spring weeds were sampled for the presence of *F. occidentalis* to determine if this newly introduced species is able to overwinter in southeastern Virginia. Early flowering weeds, primarily mustard, henbit and wild radish were collected from several sites on the Eastern Shore of Virginia, and examined for thrips. A variety of agroecosystems were sampled for the relative abundance of *F. occidentalis* in 2008, 2009 and 2010. In 2009, *F. occidentalis* was found in early spring weed samples, indicating that it is able to overwinter in this region. The species complex in 2008 on tomato, potato and two grass fields near nurseries consisted mainly of *F. tritici* and *F. fusca*, with *F. occidentalis* composing only 1 to 7% of the total number of thrips. In 2009, *F. occidentalis*

remained present, but in low numbers at all of the sites, and was not found in tomatoes in Painter, VA. *F. occidentalis* populations were further diminished in 2010 and were not present in the soybeans in Painter, VA and the field outside the high tunnel in Virginia Beach, VA.

Introduction

Although there are thousands of species of thrips distributed worldwide, only a few are known to be detrimental to crops (Mound 2005). Western flower thrips, *Frankliniella occidentalis* (Pergande) is a cosmopolitan pest that is responsible for millions of dollars in damage to a variety of crops (Lewis 1997). In addition to being highly polyphagous, and thus able to feed upon a variety of economically important crops (Capinera 2001), this species is an efficient vector of some important deadly plant tospoviruses including tomato spotted wilt virus (TSWV) and impatiens necrotic spot virus (INSV) (Ullman et al. 2002, Sakurai et al. 2004). Furthermore, this species has developed resistance to several commonly-used insecticides, particularly pyrethroids (Dagli and Tunc 2008, Frantz and Mellinger 2009). In the United States, originally this species was restricted mostly to the western states and greenhouses, then it became established in Georgia (Capinera 2001). Until recently, *F. occidentalis* was not present in significant numbers in Virginia (Nault et al. 2003); then, in summer 2007, outbreaks of *F. occidentalis* occurred in crops such as tomato, peanuts, and cotton in the eastern portion of the state (Kuhar, unpublished data). This outbreak, combined with the fact that *F. occidentalis* is very difficult to manage, has led to increased interest in determining the relative occurrence of this pest in Virginia agroecosystems, and in discovering whether or not it is an established overwintering species in the area.

The purpose of this study was to assess various plant habitats for the relative incidence of *F. occidentalis* in several agroecosystems in southeastern Virginia.

Materials and Methods

Survey of early spring weeds

One indicator that could help determine whether or not *F. occidentalis* has become established in Virginia would be its presence in weeds early during the spring when insects are beginning to become active because this insect uses several species of weeds as overwintering habitat. Although a myriad of weed species can serve as hosts, some of the best include black nightshade, *Solanum nigrum*; cheese weed, *Malva palviflora*; daisy fleabane, *Erigeron annuus*; dandelion, *Taraxacum officinale*; false dandelion, *Pyrhopappus carolinianus*; jimson weed, *Datura stramonium*; galinsoga, *Galinsoga parciflora*; lambsquarters, *Amaranthus* spp.; prickly lettuce, *Lactuca serriola*; sorrel, *Oxalis* spp. sowthistle, *Sonchus oleraceus*; and wild radish, *Raphanus raphanistrum* (Capinera 2001).

From mid-March to May 2008 and 2009, the most abundant early flowering weeds, including mustard, henbit, chickweed, and wild radish were sampled at several sites on the Eastern Shore of Virginia for the presence of overwintering thrips. Sampling was performed once per week. At each site a resealable 1-liter plastic bag was filled with one species of weed. In the lab, a 70% alcohol solution was added to the bag, sealed, and then shaken to dislodge thrips. After removing all plant matter, the subsequent liquid was processed using a Büchner funnel and thrips were counted and placed into the following categories: western flower thrips, *F. occidentalis,* flower thrips, *Frankliniella tritici* (Fitch)*,* tobacco thrips, *Frankliniella fusca*

(Hinds), and larvae (not included in tables). Thrips were identified using keys in (Capinera 2001) and many samples were sent to Gerald Brust, University of Maryland, for verification.

Thrips sampling of agroecosystems

In the spring and summer of 2008, several sites were sampled to establish the relative incidence of *F. occidentalis* in various agroecosystems. The sites included: a potato and tomato field in Painter, VA, a field near a commercial greenhouse in Chesapeake, VA (Teeuwen Greenhouses, Ltd.), and a field near a high tunnel at a nursery in Virginia Beach, VA (Bennett's Creek Nursery). A 4-liter yellow pan trap was placed in the center of each field site and filled approximately halfway with a mild water detergent solution. The pan trap was set upon a wooden post approximately one meter above ground and secured with two rubber straps strung through two holes drilled through the top end of the post and secured at each end to the rim of the pan trap with wire hooks.

Pan trap samples were processed approximately twice per week from 1 to 30 June. The contents were poured into plastic containers with tightly fitting lids, and the traps were refilled with fresh detergent solution. In the lab, trap contents were processed using a Büchner funnel and adult thrips were identified to species and counted using a dissecting microscope. For samples containing a large number of thrips, the total number of thrips was counted and a subsample of 50 thrips was identified. All adult thrips were identified in samples of 50 or fewer thrips. The identified insects were removed and placed in a glass vial with a 70% alcohol solution.

In spring 2009 and 2010, thrips were again sampled using the pan trap method. The same tomato and potato fields were sampled in Painter, VA. In Chesapeake, VA and Virginia Beach, VA the fields outside of the structures were sampled and an additional pan trap was placed inside the greenhouse and the high tunnel. The pan trap at the greenhouse in Chesapeake was placed onto a wooden table approximately one meter above the ground among the plants being grown there. This structure was mostly enclosed with airflow occurring primarily through screened vents located at intervals along the wall and also through the door. At the high tunnel in Virginia Beach, the potted plants were on the ground, and the pan trap was placed on the ground amongst the plants. The high tunnel was open at the base up to approximately one meter, and above that height was covered with plastic to form a roof. The plot was composed of two tunnels because the structures were smaller at this location compared with the greenhouse in Chesapeake, and the pan trap was located near the inner edge of one of the high tunnels. Pan trap samples were collected approximately twice per week from mid-May to mid-June. In 2010, identical sampling procedures were followed in the tomato and potato fields in Painter, VA, the greenhouse in Chesapeake, the high tunnel in Virginia Beach, and fields near both of these structures.

Also in 2009 in snap beans, collards and soybeans in Painter, VA, and in 2010 in soybeans in Painter, VA and peanuts and cotton near Suffolk, VA, thrips were sampled by collecting leaves and flowers and washing them in a 70% alcohol solution. Sample processing, identification, and preservation methods were performed as previously described.

Results

Survey of early spring weeds

Thrips numbers were low in early spring weed samples during both years. *Frankliniella occidentalis* was not found in any of the weed samples collected during 2008 (Table 1.1). The dominant thrips species were *F. fusca*, and *F. tritici*. In 2009, *F. fusca* and *F. tritici* were found on flowering weeds on the Eastern Shore of Virginia by late March, and by mid-April *F.*

occidentalis had appeared on the same weeds (Table 1.2). *F. occidentalis* was found at Willis Wharf, VA, on 7 April; Nassawadox, VA, on 23 April; and at Chesapeake, VA, on 1, 8 and 15, April 2009.

Thrips sampling of agroecosystems

In 2008, the thrips species complex on tomato, potato, and grass fields outside the high tunnel and greenhouse were composed primarily of *F. tritici* and *F. fusca.* However, *F. occidentalis* occurred at all locations comprising from 1 to 7% of the total number of thrips collected (Table 1.3). In 2009, *F. occidentalis* remained present in low numbers at all sites except for tomatoes in Painter, VA, where they were not found on any of the sample dates (Table 1.4). In 2009, the highest proportion of *F. occidentalis* was seen in the snap beans in Painter, VA, which contained 20% *F. occidentalis.* In 2010, *F. occidentalis* was present in even lower numbers in all fields (Table 1.5). The tomato field in Painter, VA, had the highest percentage of *F. occidentalis* (6%), while the soybeans in Painter, VA, and the field outside the greenhouse in Virginia Beach did not have this species of thrips.

Discussion

Frankliniella occidentalis was found in every year and at most locations sampled in spring 2008 to 2010. It was detected in early spring weed samples in 2009, indicating that it is likely an established and overwintering species in Virginia. Previous studies have shown that *F. occidentalis* can tolerate low temperatures (McDonald et al. 1997) and is capable of overwintering in other parts of the eastern United States (Chamberlin et al. 1992, Cho et al. 1995, Brust 2008).

Weeds or plants growing near crops can act as reservoirs for thrips populations, and when crops are planted, the insects can then move into the crops and feed and reproduce upon these new hosts which can result in damage and reduced yield in the affected crops. There are a wide variety of non-crop plants that have been shown to serve as thrips hosts when crops are absent or during the colder months of the year (Chamberlin et al. 1992, Durant et al. 1994, Capinera 2001, Brust 2008). In a weed sampling study in Maryland, Brust (2008) found that chickweed, wild mustard and henbit contained the greatest concentration of several species of thrips in Maryland, including *F. occidentalis*. It also appeared that thrips were overwintering to a greater extent in 2008 compared with previous years.

Samples from the different agroecosystems during all years yielded species complexes composed primarily of *F. fusca* and *F. tritici,* with *F. occidentalis* occurring in relatively low numbers. In soybeans grown in 2009, soybean thrips, *Neohydatothrips variabilis* (Beach), dominated the species complex, but this tended to be the exception. In general, it appears that *F. tritici* is the dominant flower-inhabiting thrips and *F. fusca* is the dominant leaf-feeding thrips on most crops in eastern Virginia. Sampling carried out by Nault et al. (2003) found *F. tritici* to be the dominant thrips species in tomatoes on the Eastern Shore of Virginia, indicating that it is likely responsible for much of the cosmetic injury to tomato fruit. *F. fusca* was also found, but in lower numbers, and is likely the primary vector of TSWV in the region. According to Eckel et al. (1996), *F. tritici* was the most commonly encountered thrips species among five regions of North Carolina, with several other species, including *F. fusca* present at many of the sites. Several studies have reported that *F. fusca* is a dominant pest species in peanuts, cotton and other crops in the eastern United States (Ananthakrishnan 1984, Eckel et al. 1996, Cook et al. 2003, Herbert et al. 2007). Feeding on early seedlings tends to result in the greatest damage, and high thrips populations can cause reduction in yields in both crops. As mentioned before, it is also likely the primary vector of TSWV in the eastern United States.

Although *F. occidentalis* was not dominant at any of the Virginia sites studied, treatments using pyrethroids could induce larger populations of that species. In insecticide efficacy trials conducted in Painter, VA in 2007, plots sprayed with pyrethroid insecticides had significantly more thrips than unsprayed plots (Fig. 1.1). This is one indicator that *F. occidentalis* was initially present at this location. Therefore, at sites where *F. occidentalis* is known to occur, growers need to exercise caution when treating their crops.

Insecticides such as pyrethroids can inflate *F. occidentalis* populations through a variety of mechanisms including destruction of important natural enemies such as *Orius* spp. *Orius* has been shown to control thrips populations remarkably well. According to Gillett et al. (2006), biological control with these predators can be obtained when there is a ratio of 1 predator (*Orius*) to 180 prey (thrips). Funderburk et al. (2000) found that a 1:40 ratio of *Orius insidiosus* (Say) to *F. occidentalis* resulted in near extinction of the pest within days, while thrips remained abundant in areas where predators were suppressed. Population spikes of *F. occidentalis* following treatments with pyrethroids have also been speculated to be a result of hormoligosis, although eradication of natural enemies is likely the primary reason for increased pest populations (Frantz and Mellinger 2009).

In conclusion, although *F. occidentalis* appears to be established in Virginia, it is likely not the primary cause of injury to crops grown in the region. However, growers need to exercise caution because this pest has a cryptic lifestyle and is relatively difficult to detect and control. In addition, improper treatment with insecticides that are known to inflate populations, such as pyrethroids, should be avoided. It is therefore important to recognize that *F. occidentalis* is now established in this area, and sampling and accurate identification of pestiferous thrips is very important in thrips management.

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Table 1.1. Total numbers of *Frankliniella tritici, F. occidentalis* and *F. fusca* found on flowering weeds at various locations sampled on the Eastern Shore of Virginia during the early spring of 2008. Other species of thrips were present on some dates, but counts were not included in table. Weed samples consisted primarily of mustard and henbit.

Location	**Dates Sampled**	**Thrips Species**		
		F. tritici	***F. occidentalis***	***F. fusca***
Painter, VA	3/12, 3/18, 3/19, 3/25	0	0	0
Daugherty, VA	3/25, 4/4, 4/14	3	0	5
Eastville, VA	3/12, 3/25, 4/9, 4/15	2	0	2
Machipongo, VA	4/2, 4/9, 4/15	3	0	1
Belle Haven, VA	4/4, 4/14	0	0	0
Birdsnest, VA	3/25, 3/26, 4/2, 4/9, 4/15	1	0	0
Cheriton, VA	3/12, 4/2	1	0	1
Chincoteague, VA	3/14	0	0	0

Table 1.2. Total numbers of *Frankliniella tritici, F. occidentalis* and *F. fusca* found on flowering weeds at various locations sampled in eastern Virginia during the early spring of 2009. Other species of thrips were present on some dates, but counts were not included in table. Weed samples consisted primarily of mustard, henbit, and some wild grasses.

		Thrips Species		
Location	**Dates Sampled**	***F. tritici***	***F. occidentalis***	***F. fusca***
Painter, VA	3/30, 4/7	0	0	0
Daugherty, VA	4/17	0	0	0
Eastville, VA	4/7	0	0	0
Machipongo, VA	4/17, 4/23, 4/29	1	0	2
Onley, VA	4/29	3	0	0
Willis Warf, VA	4/7, 4/17, 4/23, 4/29	7	1	5
Nassawadox, VA	4/7, 4/23, 4/29	1	1	9
Melfa, VA	4/17, 4/23	0	0	0
Capeville, VA	3/24	1	0	0
Cape Charles, VA	4/17	0	0	1
Pungoteague, VA	4/17, 4/23, 4/29	0	0	0
Locustville, VA	4/23	0	0	0
Wachapreague, VA	4/29	3	0	0
Virginia Beach, VA	3/30, 4/1, 4/8, 4/15, 4/22, 4/29	4	0	7
Chesapeake, VA	4/1, 4/8, 4/15, 4/22, 4/29	36	11	12

Table 1.3. Thrips species complex in several agroecosystems in eastern Virginia during 2008. All samples were collected from pan traps. Percent species composition was calculated by dividing the total number of thrips of each species by the total number of thrips collected on all sample dates for each site.

		Percent Species Composition			
Location	**Dates Sampled**	***F. tritici***	***F. occidentalis***	***F. fusca***	***Other***
Painter, VA- potato	Sampled multiple times June 1- 30	54%	1%	32%	13%
Painter, VA- tomato	Sampled multiple times June 1- 30	27%	6%	41%	26%
Virginia Beach, VA- outside greenhouse	Sampled multiple times June 1- 30	59%	2%	12%	27%
Chesapeake, VA- inside greenhouse	Sampled multiple times June 1- 30	65%	7%	14%	14%

Table 1.4. Thrips species complex in several agroecosystems in eastern Virginia during 2009. Thrips collected from potatoes, tomatoes, areas outside of greenhouses, and areas inside greenhouses were from pan trap samples. Thrips on snap beans, collards and soybeans were collected from flower and leaf samples that were washed and vacuum filtered using a Büchner funnel. Percent species composition was calculated by dividing the total number of thrips of each species by the total number of thrips collected on all sample dates for each site.

		Percent Species Composition			
Location	**Dates Sampled**	***F. tritici***	***F. occidentalis***	***F. fusca***	***Other***
Painter, VA- potatoes	5/19, 6/1, 6/9, 6/24, 6/26	81%	3%	1%	16%
Painter, VA- tomatoes	6/1, 6/9, 6/11, 6/24, 6/26	87%	0%	2%	12%
Painter, VA- snap beans	7/2	64%	20%	10%	6%
Painter, VA- collards	7/3	2%	18%	54%	26%
Painter, VA- soybeans	8/7	0%	4%	0%	96%*
Chesapeake, VA- outside greenhouse	5/18, 5/26, 6/1, 6/11, 6/15	88%	3%	7%	2%
Chesapeake, VA- inside greenhouse	5/26, 6/1, 6/4, 6/8, 6/15	75%	2%	7%	16%
Virginia Beach, VA- outside greenhouse	5/18, 5/26, 6/1, 6/4, 6/15	83%	5%	5%	6%
Virginia Beach, VA- inside greenhouse	5/28, 6/1, 6/11, 6/22, 6/29	75%	7%	10%	8%

* Dominant thrips species in soybeans during this year was *Neohydatothrips variabilis* (Beach).

Table 1.5. Thrips species complex in several agroecosystems in eastern Virginia during 2009. Thrips collected from potatoes, tomatoes, areas outside of greenhouses, and areas inside greenhouses were from pan trap samples. Thrips on soybeans, peanuts and cotton were collected from leaf samples that were washed and vacuum filtered using a Büchner funnel. Percent species composition was calculated by dividing the total number of thrips of each species by the total number of thrips collected on all sample dates for each site.

		Percent Species Composition			
Location	**Dates Sampled**	***F. tritici***	***F. occidentalis***	***F. fusca***	***Other***
Painter, VA- potato field	5/21, 5/27, 5/30, 6/4, 6/15	88%	2%	4%	6%
Painter, VA- tomato field	5/18, 5/26, 5/30, 6/4, 6/15	73%	6%	6%	15%
Painter, VA- soybean field	6/20, 6/23, 6/28	6%	0%	86%	13%
Suffolk, VA- peanut field	5/25, 6/1, 6/8	4%	1%	86%	9%
Suffolk, VA- cotton field	5/25, 6/1, 6/8	7%	1%	77%	15%
Virginia Beach, VA- outside greenhouse	5/24, 5/27, 6/1, 6/10, 6/14	86%	0%	8%	6%
Virginia Beach, VA- inside greenhouse	5/20, 5/24, 6/1, 6/3, 6/10	48%	2%	28%	22%
Chesapeake, VA- outside greenhouse	5/20, 5/24, 5/27, 6/3, 6/7	97%	1%	2%	0%
Chesapeake, VA- inside greenhouse	5/24, 5/27, 6/1, 6/7, 6/14	84%	1%	5%	10%

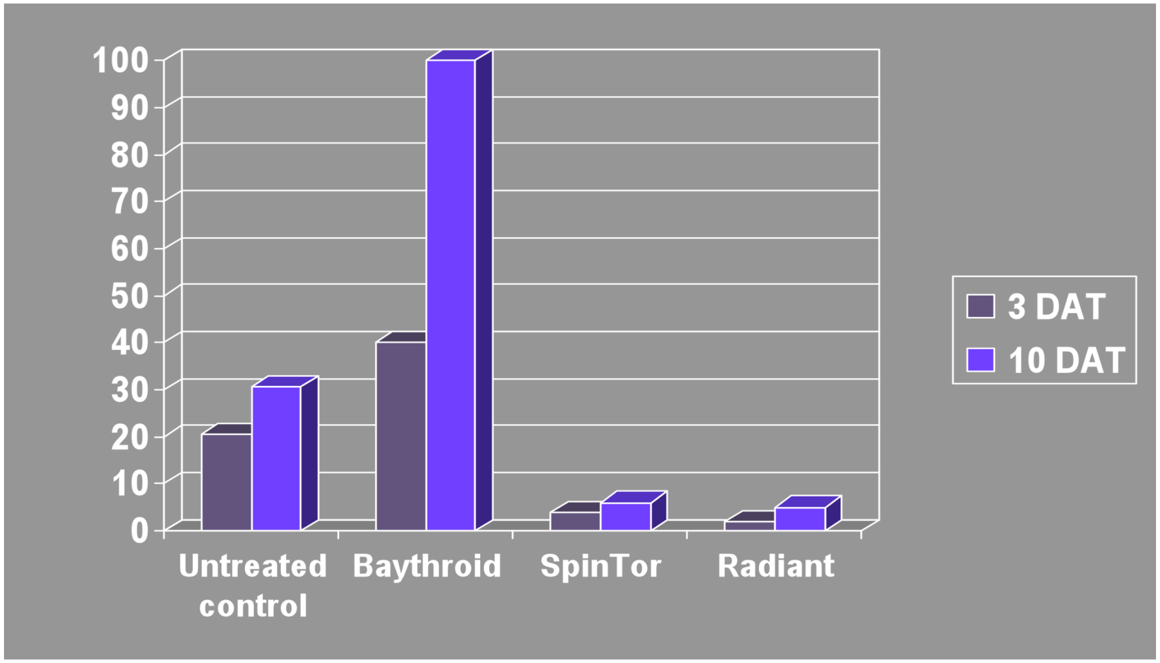

Figure 1.1. Numbers of thrips per 10 leaves (mostly *F. occidentalis*) on tomato plots at 3 and 10 days after being sprayed with three commonly-used insecticides at recommended rates on tomatoes; Painter, VA 2007.

Chapter 2: Evaluation of a synthetic thrips aggregation pheromone lure and a kairomone lure in their ability to attract thrips to sticky traps

Abstract

Effective sampling is crucial for the early identification of pests, particularly those that are difficult to detect and manage, such as thrips. One method to increase the attractiveness of sampling devices such as sticky cards is with chemical lures. Several species of thrips have been shown to exhibit olfactory responses to a variety of compounds including floral attractants and aggregation pheromones. Two types of lures were tested in their ability to attract *Frankliniella* spp. thrips in a variety of agroecosystems in eastern Virginia. The lures included Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO), composed of a proprietary floral compound mixture, and Thripline$_{AMS}$ (Syngenta Bioline Ltd., Oxnard, CA), containing the aggregation pheromone of *Frankliniella occidentalis* (Pergande). From May to June 2009 and 2010, lure experiments were conducted in eight agroecosystems including: a tomato and potato field in Painter, VA, a cotton and peanut field in Suffolk, VA, a field containing predominately grass bordering a greenhouse and the area within the greenhouse at a commercial plant nursery in Chesapeake, VA, and within a high tunnel and a nearby grass field in Virginia Beach, VA. Baited and non-baited sticky cards were arranged in a completely randomized design, with a pan trap located in the center of each plot. Traps were collected approximately twice weekly for several weeks. *Frankliniella fusca* (Hinds) sticky card catch densities were low and were not significantly affected by either lure. Sticky cards baited with the kairomone caught more flower

thrips (both *Frankliniella tritici* (Fitch) and *F. occidentalis*) than traps baited with pheromone, or the non-baited traps, especially when thrips numbers were high.

Introduction

Thrips (Thysanoptera) can be important pests of a number of agricultural crops (Lewis 1997, Mound 2005). In Virginia, the primary pest species include tobacco thrips, *Frankliniella fusca* (Hinds), and flower thrips, *Frankliniella tritici* (Fitch), although western flower thrips, *Frankliniella occidentalis* (Pergande) also occurs in low densities (Chapter 1). High densities of thrips adults and larvae and subsequent feeding injury on seedlings, flowers, pods, and fruit can cause significant yield losses in row crops such as cotton, peanuts, and vegetables, particularly tomatoes (Pohronezny et al. 1986, Childers 1997, Mound 1997, Nault et al. 2003, Herbert et al. 2007). In addition, *F. fusca* and *F. occidentalis* can transmit devastating plant pathogenic tospoviruses, such as tomato spotted wilt virus and impatiens necrotic spot virus (Johnson et al. 1995, Eckel et al. 1996, Groves et al. 2001, 2002). Because of their small size and cryptic lifestyles, thrips injury is often noticed before the actual insects have been detected.

There are several reasons to monitor for thrips in crops including detection of their initial presence, locating areas in crops considered "hot spots", predicting outbreaks of disease, to determine the timing of control measures, and to assess the effectiveness of the implemented control measures (Shipp 1995). Sticky cards and pan traps are the most commonly used tools for monitoring thrips for research and pest management purposes (McPherson and Riley 2006), but efficacy and accuracy of these devices can be limited.

Thrips have been shown to be attracted to various semiochemicals including plant volatiles and pheromones. Male *F. occidentalis* have been found to produce an aggregation pheromone, attracting both male and female thrips (Hamilton et al. 2005). The two main components of this male-specific pheromone were identified as (R)-lavandulyl acetate and neryl (*S*)-2-methylbutanoate, with the latter showing activity in field trials. In a previous study by Kirk and Hamilton (2004), this pheromone was identified as a sex pheromone, but it has now been shown to attract thrips of both sexes. Based upon the olfactory response of many thrips species, several companies are manufacturing lures to improve sampling. Thripline$_{AMS}$ (Syngenta Bioline Ltd., Oxnard, CA) is a synthetic lure septum containing the *F. occidentalis* aggregation pheromone.

Plant-produced volatiles, particularly odors given off by flowers that are within the chemical class of benzenoids and monoterpenes have been shown to be attractive to thrips (Koschier et al. 2000). For instance, Hollister et al. (1995) found that the compound *p*-anisaldehyde increased capture of *F. occidentalis* more than 100-fold over non-baited black pan traps. Other attractive compounds have been synthesized into products including Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO), which is composed of a variety of floral compounds.

Coupling attractive lures with attractively-colored traps such as yellow or blue sticky cards and pan traps can enhance sampling and allow for better detection of thrips. Yellow and blue traps are more attractive to many species of flower-dwelling thrips, probably because these colors are often associated with the plant hosts.

The purpose of this study was to evaluate the efficacy of the commercially-available Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO) and the Thripline$_{AMS}$ (Syngenta Bioline Ltd., Oxnard, CA) pheromone lure in their ability to increase catch of *Frankliniella* thrips in various agroecosystems.

Materials and Methods

Completely randomized experiments were conducted from May to June 2009 and 2010 in several different agroecosystems in eastern Virginia. In 2009, the experiment was conducted in: a tomato and potato field at the Eastern Shore Agricultural Research and Extension Center (ESAREC) in Painter, VA, a cotton and peanut field at the Tidewater Agricultural Research and Extension Center (TAREC) in Suffolk, VA, a grass field bordering a greenhouse and within the greenhouse at a nursery in Chesapeake, VA (Teeuwen Greenhouses, Ltd.) and a high tunnel nursery and nearby grass field located in Virginia Beach, VA (Bennett's Creek Nursery). At each location, yellow 10.16 x 15.24 cm sticky cards (Olson Products, Medina, OH) with one side exposed were placed upon wooden stakes or wire sticky card holders approximately 30 cm above the ground using various metal clamps or wire to secure the traps. Lures were attached to the stakes or wire traps so that they were either contacting the edge of the cards, or were at most 3 cm away from the cards.

Each plot contained a total of 15 sticky cards, which consisted of five non-baited controls; five baited with Thripline$_{AMS}$ 8061-02 (Syngenta Bioline Ltd., Oxnard, CA), a synthetically-produced *F. occidentalis* aggregation pheromone on rubber septum, hereafter referred to as "pheromone"; and five traps baited with Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO), a mixture of floral volatiles contained in lure sachets, hereafter referred to as "kairomone". The sticky cards were placed approximately 10 m apart in a completely randomized design at each field site. A 4-liter yellow pan trap was placed in the center of each field site upon a wooden post approximately one meter above ground and secured with two rubber straps strung through two holes drilled through the top end of the post and secured at each end to the rim of the pan trap with wire hooks at quarter intervals around the rim of the pan trap.

At the greenhouse and high tunnel sites, a set of traps was placed within each structure, and another set was placed outside in nearby grass fields. Sticky cards within the greenhouse and high tunnel were closer together compared with the cards in the fields, about 3 m, due to the limited amount of space within the enclosures. A pan trap was located in the approximate center of each plot. The high tunnel located in Virginia Beach was open from the ground up to approximately one meter, whereupon it was covered with clear plastic. Plant species changed from one week to the next and therefore were not a constant factor. Plants were located on the ground in rows, as well as in hanging pots suspended from the ceiling. The pan trap was placed on the ground in the middle of the inner edge of one of the high tunnels. The greenhouse at the Chesapeake site was a more securely enclosed building, with airflow occurring primarily through screened vents with fans, and a single sliding door. The plants grown in this facility were placed on wooden tables about 1.5 m high and consisted primarily of geraniums. Sticky cards were placed in pots filled with gravel so that the bases of the traps were approximately 12 cm above the soil.

In 2010, traps were set up at the same sites used the previous year, except the cotton and peanut fields located at TAREC. A different, but similarly constructed, greenhouse was used at the Chesapeake site because the greenhouse used the previous year did not contain any plants. This new greenhouse contained a greater variety of plants than the greenhouse used the previous year, and the plant species were changed often. Identical protocols were followed in the establishment and subsequent sampling of traps at all sites.

Data collection

Sticky cards were collected every three to four days (approximately two times per week), placed in plastic wrap, and replaced with clean cards. Cards were stored in a freezer to extend preservation. Thrips caught on sticky cards were recorded into three categories: 1) tobacco thrips, *F. fusca* 2) flower thrips, which included both *F. tritici* and *F. occidentalis,* and 3) other, which included a variety of thrips species, but were not analyzed in the statistical model.

The contents of the pan traps was sampled at the time of sticky card collection. The contents within the pan was poured into a plastic container with a sealable lid and then taken back to the lab. Pan trap contents were passed through a Büchner funnel and adult thrips were identified to species using a dissecting microscope. Thrips were categorized as western flower thrips, *F. occidentalis,* flower thrips *F. tritici,* tobacco thrips *F. fusca*, onion thrips *Thrips tabaci,* soybean thrips *Neohydatothrips variabilis* (Beach), and *Limothrips cerealium* (Haliday). All thrips were identified when the pan trap sample contained at least 50 thrips and were then removed using a fine metal point and placed into a vial labeled with date and location containing a 70% alcohol solution. When more than 50 thrips were present, a subsample of 50 thrips was identified and stored in a labeled vial with 70% alcohol while additional thrips were counted and then discarded. Identifying thrips collected from pan traps allows for improved accuracy in identifying thrips beyond that attainable from sticky cards because all parts of the body can be viewed whereas only a limited portion of the body can be viewed when a thrips is on a sticky card.

Data Analysis

Data from both years were analyzed separately, but in an identical manner.

Determination of Significant Treatment Effect- A negative binomial generalized linear model (SAS® 9.2 Software) was used to analyze both years for a significant treatment effect on catches of flower thrips (*F. tritici* and *F. occidentalis*) and *F. fusca*. The test was adjusted for multiple comparisons with the control using the Dunnett-Hsu procedure.

At sites where a significant treatment effect was detected, total flower thrips (both *F. tritici* and *F. occidentalis*) and *F. fusca* were averaged per treatment for each sample date. Standard deviations were generated, and bar graphs were produced to provide a visual representation of mean thrips catches for each sample date. Standard error bars were fitted to the graphs according to data.

Results

Thrips densities varied significantly depending upon sample date, although it can be readily observed from the mean and standard error bars in figures 2.1-2.8 that, on some dates, thrips catches were significantly affected by the treatments.

Tobacco thrips. In 2009, *F. fusca* catches were not significantly affected by either lure (Tables 2.1 and 2.2). In 2010 *F. fusca* catches were increased by the kairomone at the greenhouse in Chesapeake by a factor of 1.82, but catches appeared to decrease in the potatoes by a factor of 0.55 when traps were baited with this lure (Table 2.3 and Fig. 2.8). *F. fusca* catches were also reduced by the pheromone by a factor of 0.55 in the field near the greenhouse at Chesapeake (Table 2.4).

Flower thrips. Flower thrips (*F. tritici* and *F. occidentalis*) catches were increased in 2009 by the kairomone at all sites except the high tunnel in Virginia Beach (Table 2.1 and Fig. 2.1, 2.2, 2.3, 2.4a). The greatest catch increase in traps baited with the kairomone seen during this year occurred at the greenhouse located in Chesapeake, where flower thrips catches were increased by a factor of 5.04. The pheromone increased trap catches of flower thrips at the greenhouse in Chesapeake, the grass field in Virginia Beach, and the tomatoes (Table 2.2). Trap catches baited with this lure in the greenhouse in Chesapeake were increased by a factor of 7.03, which was the largest trap catch increase seen at any of the sites throughout the course of this study.

In 2010 flower thrips catches were increased by the kairomone at all of the sites (Table 2.3 and Fig. 2.4b, 2.5, 2.6, 2.7). The greatest increase in trap catch during this year occurred at the greenhouse in Chesapeake where trap catches were increased by a factor of 5.63 by the kairomone. Traps baited with the pheromone caught more flower thrips than the non-baited control at the greenhouse in Chesapeake and in the tomatoes (Table 2.4). At the grass field in Chesapeake, pheromone-baited trap catches were reduced, catching approximately half as many flower thrips compared with the non-baited control

Discussion

Monitoring pest populations is essential for successful pest control. Early detection and accurate identification of thrips could help alert an invasion early enough to prevent serious damage to crops. Yellow sticky traps have been shown to increase catches of thrips (Smits et al. 2000), but increasing trap attractiveness using a second attractive cue such as a chemical lure could help to increase sampling efficacy.

Results of this study showed that the kairomone and pheromone both increased catch of flower thrips (*F. tritici* and *F. occidentalis*) in certain agroecosystems. In general, sticky cards baited with the kairomone caught more flower thrips than non-baited cards or cards baited with the pheromone, particularly when thrips numbers were highest.

Frankliniella fusca, densities were lower overall than the flower thrips, and the treatment effects were less obvious. Overall, it appears that neither commercial lure had a significant effect on *F. fusca* catch. These results are similar to those of Kirk (1985) who found that water traps baited with several floral scents significantly increased trap catches of several species of floral-dwelling thrips, but did not have a significant effect upon several species of thrips that prefer other plant parts such as leaves. While *F. occidentalis* and *F. tritici* are often found among flowers, *F. fusca* tends to prefer to feed and reside upon plant foliage.

Frey et al. (1994) found that traps tested in greenhouses experienced a decrease of efficacy compared with studies conducted in the laboratory using the same attractants. Several factors can have an impact on lure attractiveness in greenhouses and the field. Release rates of the lures can be significantly altered by changes in temperature and/or humidity (Frey et al. 1994). When concentrations of odors such as salicylaldehyde and *p*-anisaldehyde are above a certain threshold, the scents will act as repellents for certain insects (Koschier et al. 2000). In contrast, a low odor concentration may not elicit a response. Air movement may also affect the build-up of an odor gradient being emitted by a baited trap. This could have an effect on the insect's ability to locate the trap. Such factors could explain the moderate increase in most trap catches observed during this study.

In some cases, flowering plants may compete with attractant lures, thereby decreasing the attractiveness of the lures. This could have been the case in some of the habitats in this study such as the greenhouses where flowering plants were often present, and also in the fields near the greenhouses where a myriad of flowering weeds and shrubs were growing nearby. However, some of the greatest increases in flower thrips caught on sticky cards were observed in the greenhouse in Chesapeake, and flower thrips capture was also increased in the fields near the greenhouse in Chesapeake and the high tunnel in Virginia Beach. Perhaps the species of flowers being grown in the greenhouse and high tunnel did not compete with the kairomone, and instead served as synergists by initially attracting the flower thrips, which were more attracted to the lures once they were within the vicinity of the attractive cues. The fields near the greenhouse and high tunnel may have also contained attractive features such as flowering weeds, which helped to attract thrips that were then more attracted to the lures once they were close enough to detect the kairomone volatiles.

Species composition is another important factor to consider when reviewing the results from this study. In general, a large portion of thrips found in and near the greenhouse and high tunnel were flower thrips. Many species of flower dwelling thrips are attracted to floral compounds (Kirk 1985, Koschier et al. 2000). Therefore, there was a higher number of thrips that are attracted to floral compounds within these locations compared with other habitats in this study such as the cotton and peanut fields, which contained higher populations of *F. fusca* and other species of thrips.

Frankliniella occidentalis was present at most of the sites in low numbers. *F. occidentalis* and *F. tritici* look very similar, with morphological differentiation between these two species based upon only a couple of features, particularly the length of post ocular setae. It is therefore important to be able to view the dorsal portion of both species. Sticky cards can make this a difficult task if the thrips are attached to the cards by their backs. When thrips were counted on the sticky cards, *F. occidentalis* and *F. tritici* were both put into the same category due to the difficulty and reduced ability to differentiate between these two species on sticky cards. Thrips were identified to species level in the pan trap, and therefore species composition at each site for each sample date was based upon this sampling method. It is possible that the results for the pheromone do not accurately reflect the efficacy of this lure on *F. occidentalis* because it was present in such low numbers, with the population of flower thrips being primarily composed of *F. tritici.*

For thrips sampling, trap color alone has been shown to markedly increase thrips capture in certain cases (Cho et al. 1995). Although there is still debate as to which color is the most effective (Shipp 1995), yellow, white or blue appear to have the greatest success. Results obtained from this study indicate only a moderate increase in thrips capture when traps are baited with an attractive kairomone in most habitats. Other reports such as Hollister et al. (1995) have shown a greater increase in trap capture, and this could be due to differing habitats or perhaps different compounds used as attractants.

The lures tested in this study would likely be beneficial in a variety of cropping systems where early detection of pestiferous flower-dwelling species of thrips could help alert growers to begin monitoring more intensely for injury to crops, or to treat their crops if thrips numbers become high enough to warrant action. Additional experiments should also be carried out with the pheromone in locations where *F. occidentalis* is present in higher numbers to more accurately assess the attractiveness of this lure because *F. occidentalis* was present in very low numbers during the course of this experiment.

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Table 2.1. Catches of thrips in 2009 on sticky cards baited with Chemtica P-178 floral kairomone lure (Kairomone) and a non-baited control (Non-Baited Control). For thrips categories the mean number of thrips for each treatment is given averaged over all other factors in the model including date, site, and card number. Thrips are divided into two categories: tobacco thrips (*F. fusca*) and flower thrips (*F. tritici* and *F. occidentalis*). The ratio of baited trap catches to control catches (Ratio of Baited Sticky Card Catches to Control), their standard errors of difference (SED), and the significance level adjusted using Dunnett-Hsu (Adj P) are also given.

Location*	**Category of Thrips**	**Non-Baited Control**	**Kairomone**	**Ratio of Baited Sticky Card Catches to Control**	**SED**	**Adj P**
COT	tobacco thrips	1.97	1.57	0.80	0.19	0.37
COT	flower thrips	8.78	13.74	1.57	0.12	<0.001
GH-C	tobacco thrips	3.95	4.09	1.04	0.30	0.99
GH-C	flower thrips	6.58	33.11	5.04	0.21	<0.001
HT-VB	tobacco thrips	0.93	0.84	0.90	0.21	0.84
HT-VB	flower thrips	5.33	4.53	0.85	0.16	0.51
GRA-C	tobacco thrips	2.19	1.82	0.83	0.21	0.58
GRA-C	flower thrips	24.39	53.50	2.19	0.15	<0.001
GRA-VB	tobacco thrips	4.06	3.38	0.83	0.15	0.34
GRA-VB	flower thrips	19.81	47.28	2.39	0.16	<0.001
PNT	tobacco thrips	1.65	1.30	0.79	0.22	0.43
PNT	flower thrips	8.32	12.40	1.49	0.14	0.01
POT	tobacco thrips	7.10	5.38	0.76	0.18	0.21
POT	flower thrips	7.49	13.64	1.82	0.16	<0.001
TOM	tobacco thrips	19.27	14.46	0.75	0.13	0.07
TOM	flower thrips	5.64	11.15	1.98	0.16	<0.001

* Locations in table include: COT= cotton field, GH-C= inside greenhouse in Chesapeake, HT-VB= high tunnel in Virginia Beach, GRA-C= grass field near greenhouse in Chesapeake, GRA-VB= grass field near high tunnel in Virginia Beach, PNT= peanut field, POT= potato field, TOM= tomato field

Table 2.2. Catches of thrips in 2009 on sticky cards baited with Thripline$_{AMS}$ aggregation pheromone lure (Pheromone) and a non-baited control (Non-Baited Control). For each category of thrips the mean number of thrips for each treatment is given averaged over all other factors in the model including date, site, and card number. Thrips are divided into two categories: tobacco thrips (*F. fusca*) and flower thrips (*F. tritici* and *F. occidentalis*). The ratio of baited trap catches to control catches (Ratio of Baited Sticky Card Catches to Control), their standard errors of difference (SED), and the significance level adjusted using Dunnett-Hsu are also given (Adj P).

Location*	Category of Thrips	Non-Baited Control	Pheromone	Ratio of Baited Sticky Card Catches to Control	SED	Adj P
COT	tobacco thrips	1.97	1.46	0.74	0.19	0.21
COT	flower thrips	8.78	9.51	1.08	0.12	0.73
GH-C	tobacco thrips	3.95	5.07	1.28	0.29	0.60
GH-C	flower thrips	6.58	19.65	7.03	0.21	<0.001
HT-VB	tobacco thrips	0.93	1.14	1.23	0.20	0.47
HT-VB	flower thrips	5.33	5.22	0.98	0.17	0.99
GRA-C	tobacco thrips	2.19	2.26	1.03	0.21	0.98
GRA-C	flower thrips	24.39	28.12	1.15	0.15	0.55
GRA-VB	tobacco thrips	4.06	3.78	0.93	0.15	0.84
GRA-VB	flower thrips	19.81	45.13	2.28	0.17	<0.001
PNT	tobacco thrips	1.65	1.29	0.78	0.22	0.41
PNT	flower thrips	8.32	6.95	0.84	0.14	0.34
POT	tobacco thrips	7.10	6.54	0.92	0.18	0.86
POT	flower thrips	7.49	10.31	1.38	0.16	0.08
TOM	tobacco thrips	19.27	22.66	1.18	0.13	0.36
TOM	flower thrips	5.64	8.86	1.57	0.16	0.01

* Locations in table include: COT= cotton field, GH-C= inside greenhouse in Chesapeake, HT-VB= high tunnel in Virginia Beach, GRA-C= grass field near greenhouse in Chesapeake, GRA-VB= grass field near high tunnel in Virginia Beach, PNT= peanut field, POT= potato field, TOM= tomato field

Table 2.3. Catches of thrips in 2010 on sticky cards baited with Chemtica P-178 floral kairomone lure (Kairomone) and a non-baited control (Non-Baited Control). For each category of thrips the mean number of thrips for each treatment is given averaged over all other factors in the model including date, site, and card number. Thrips are divided into two categories: tobacco thrips (*F. fusca*) and flower thrips (*F. tritici* and *F. occidentalis*). The ratio of baited trap catches to control catches (Ratio of Baited Sticky Card Catches to Control), their standard errors of difference (SED), and the significance level adjusted using Dunnett-Hsu (Adj P) are also given.

Location*	Category of Thrips	Non-Baited Control	Kairomone	Ratio of Baited Sticky Card Catches to Control	SED	Adj P
GH-C	tobacco thrips	1.88	3.43	1.82	0.25	0.04
GH-C	flower thrips	14.59	82.18	5.63	0.25	<0.001
HT-VB	tobacco thrips	1.58	1.57	0.99	0.22	1.00
HT-VB	flower thrips	3.05	5.12	1.68	0.19	0.01
GRA-C	tobacco thrips	2.53	3.54	1.40	0.20	0.16
GRA-C	flower thrips	64.70	107.53	1.66	0.17	0.01
GRA-VB	tobacco thrips	6.13	9.33	1.52	0.21	0.09
GRA-VB	flower thrips	26.05	61.83	2.37	0.16	<0.001
POT	tobacco thrips	5.33	2.92	0.55	0.14	<0.001
POT	flower thrips	14.44	22.90	1.59	0.14	<0.001
TOM	tobacco thrips	5.85	5.31	0.91	0.13	0.65
TOM	flower thrips	6.73	8.92	1.33	0.10	0.01

* Locations in table include: GH-C= inside greenhouse in Chesapeake, HT-VB= high tunnel in Virginia Beach, GRA-C= grass field near greenhouse in Chesapeake, GRA-VB= grass field near high tunnel in Virginia Beach, POT= potato field, TOM= tomato field

Table 2.4. Catches of thrips in 2010 on sticky cards baited with Thripline$_{AMS}$ aggregation pheromone lure (Pheromone) and a non-baited control (Non-Baited Control). For each category of thrips the mean number of thrips for each treatment is given averaged over all other factors in the model including date, site, and card number. Thrips are divided into two categories: tobacco thrips (*F. fusca*) and flower thrips (*F. tritici* and *F. occidentalis*). The ratio of baited trap catches to control catches (Ratio of Baited Sticky Card Catches to Control), their standard errors of difference (SED), and the significance level adjusted using Dunnett-Hsu (Adj P) are also given.

Location*	**Category of Thrips**	**Non-Baited Control**	**Pheromone**	**Ratio of Baited Sticky Card Catches to Control**	**SED**	**Adj P**
GH-C	tobacco thrips	1.88	2.78	1.47	0.26	0.23
GH-C	flower thrips	14.59	27.82	1.91	0.25	0.02
HT-VB	tobacco thrips	1.58	1.68	1.06	0.22	0.94
HT-VB	flower thrips	3.05	4.01	1.32	0.20	0.27
GRA-C	tobacco thrips	2.53	1.40	0.55	0.22	0.02
GRA-C	flower thrips	64.70	35.22	0.54	0.17	<0.001
GRA-VB	tobacco thrips	6.13	8.24	1.35	0.21	0.27
GRA-VB	flower thrips	26.05	30.26	1.16	0.17	0.57
POT	tobacco thrips	5.33	5.56	1.04	0.13	0.92
POT	flower thrips	14.44	18.11	1.25	0.14	0.18
TOM	tobacco thrips	5.85	7.05	1.21	0.12	0.22
TOM	flower thrips	6.73	8.91	1.33	0.10	0.01

* Locations in table include: GH-C= inside greenhouse in Chesapeake, HT-VB= high tunnel in Virginia Beach, GRA-C= grass field near greenhouse in Chesapeake, GRA-VB= grass field near high tunnel in Virginia Beach, POT= potato field, TOM= tomato field

Figure 2.1. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in cotton in Suffolk, VA (A) and inside a greenhouse in Chesapeake, VA (B) in 2009.

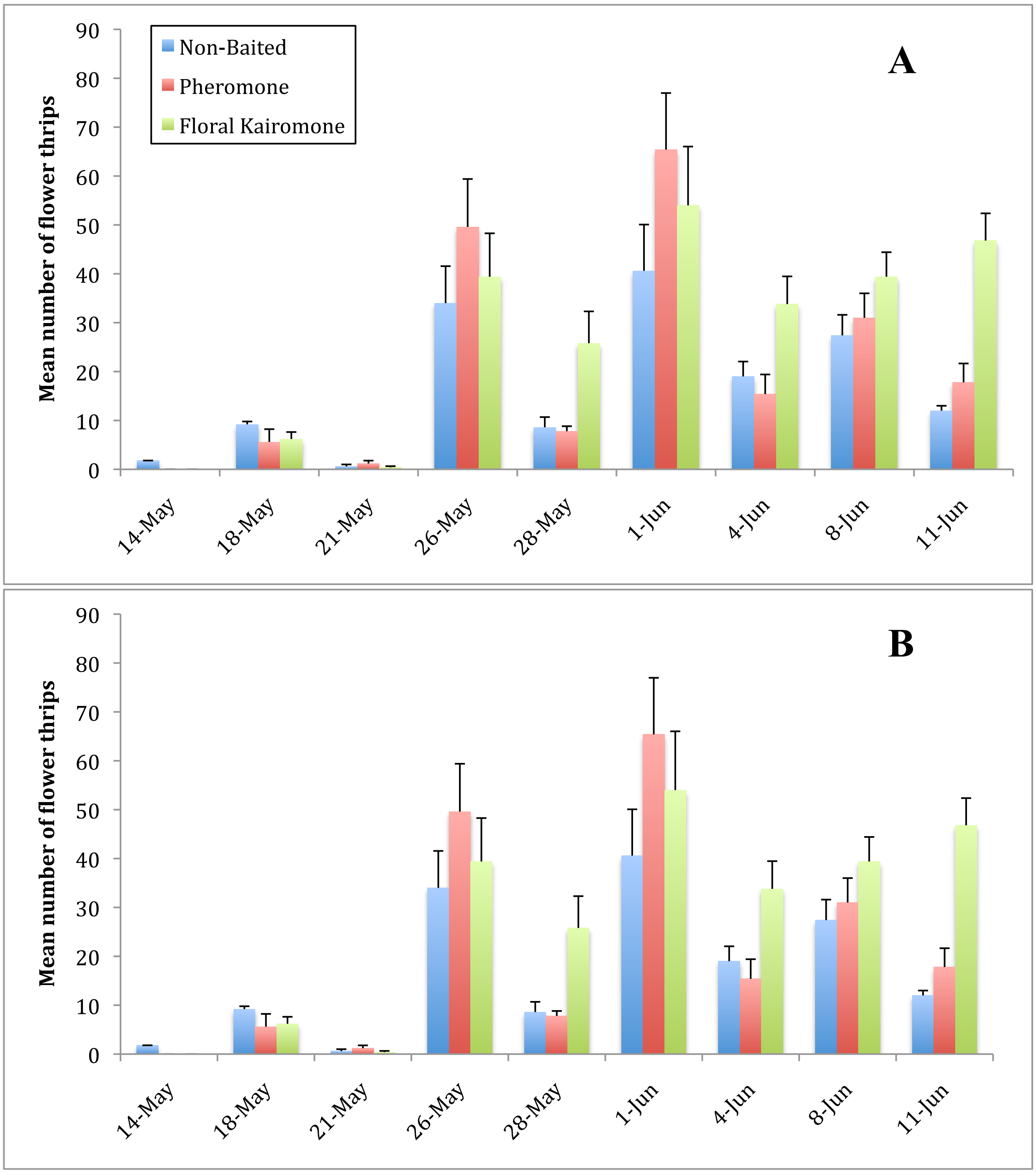
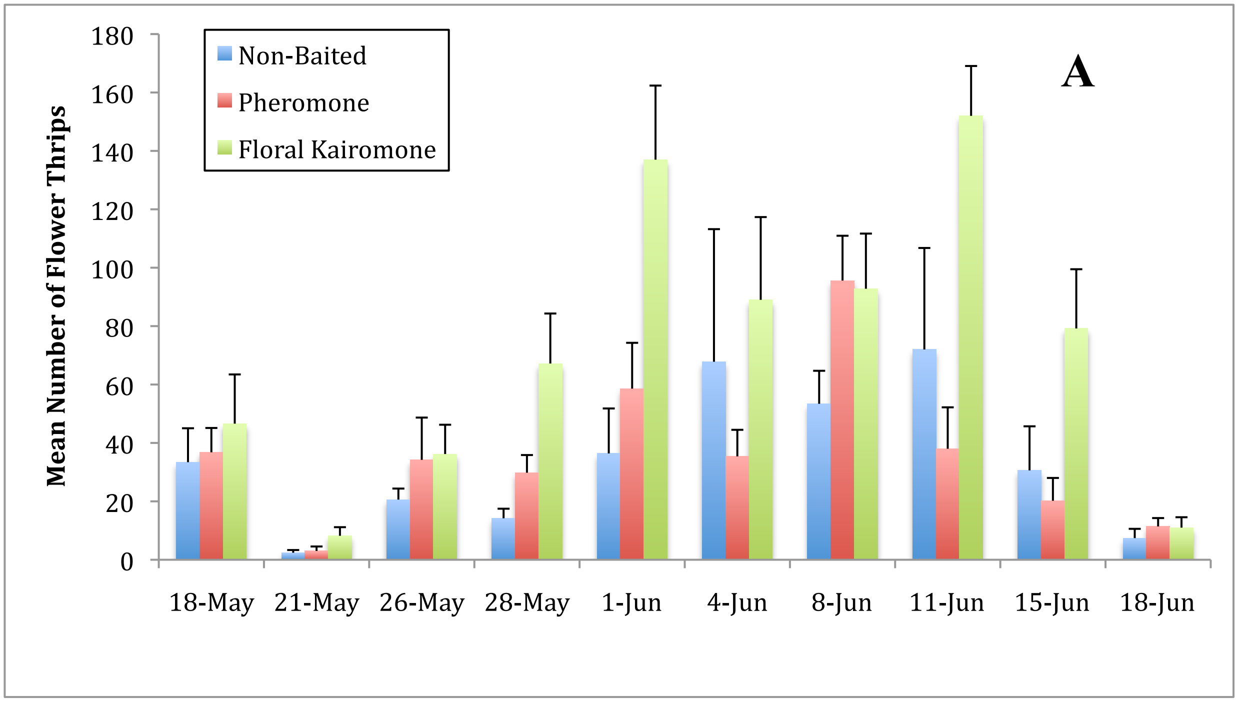


Figure 2.2. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a grass field near a greenhouse in Chesapeake, VA (A) and in a grass field near a wind tunnel in Virginia Beach, VA (B) in 2009.



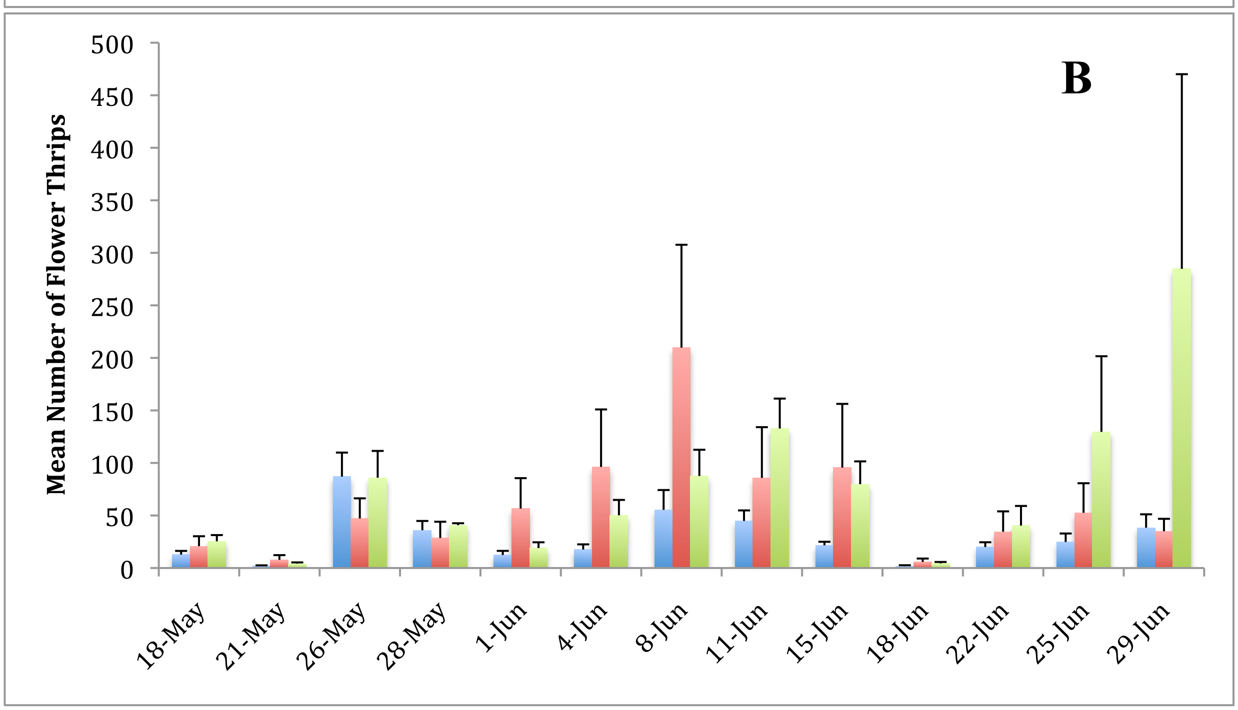


Figure 2.3. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a peanut field in Suffolk, VA (A) and in a potato field in Painter, VA (B) in 2009.

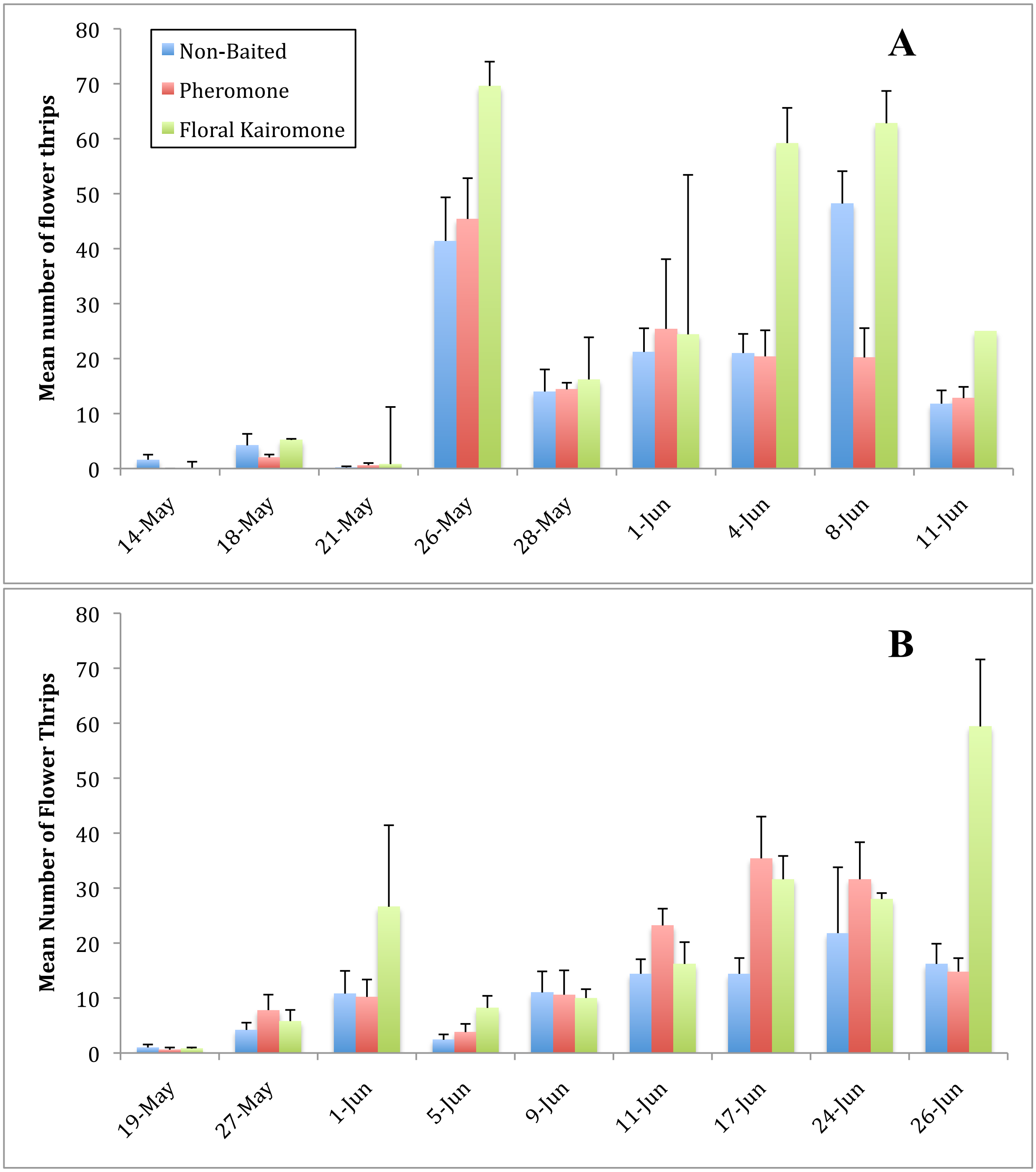
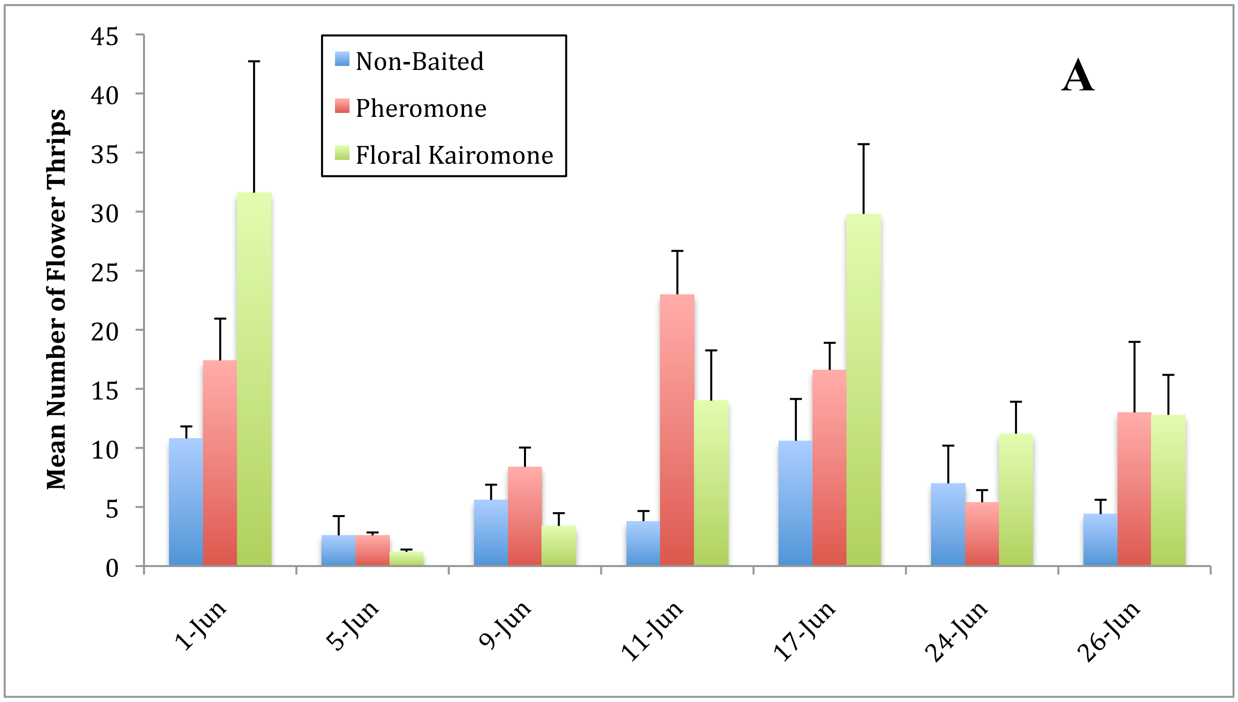


Figure 2.4. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a tomato field in Painter, VA (A) in 2009 and inside a greenhouse in Chesapeake, VA (B) in 2010.



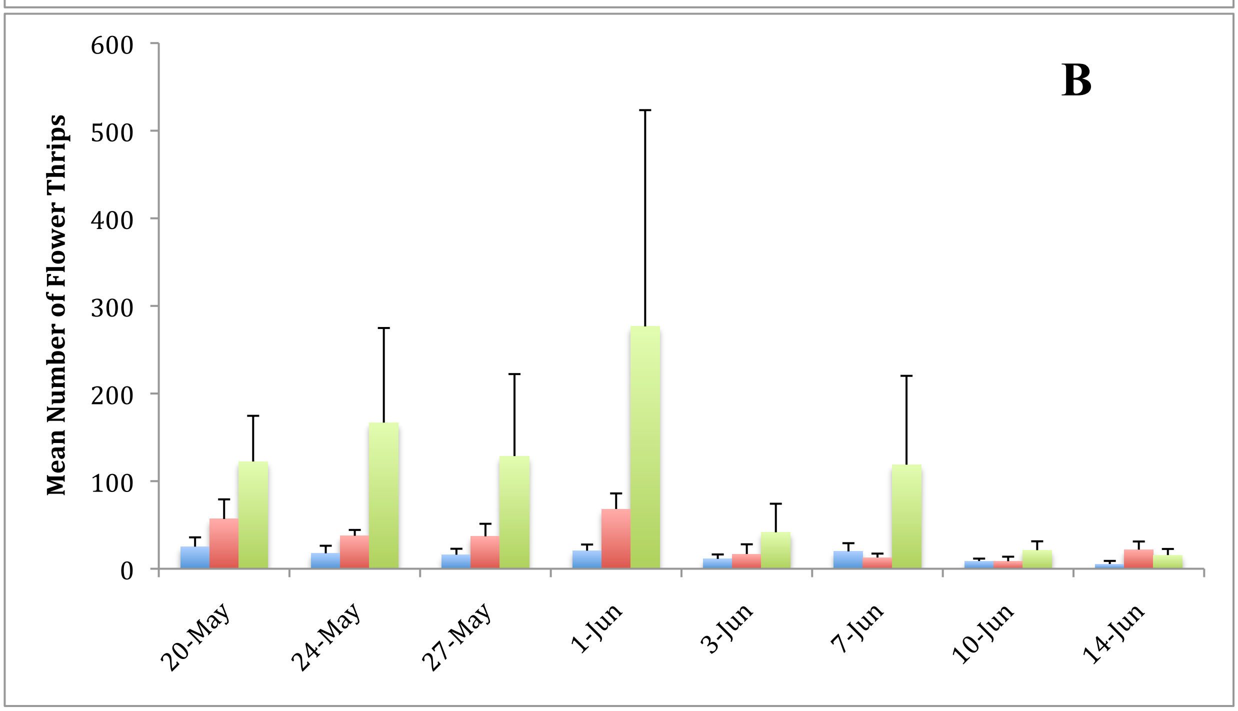


Figure 2.5. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) inside a high tunnel in Virginia Beach, VA (A) and in a grass field near a greenhouse in Chesapeake, VA (B) in 2010.

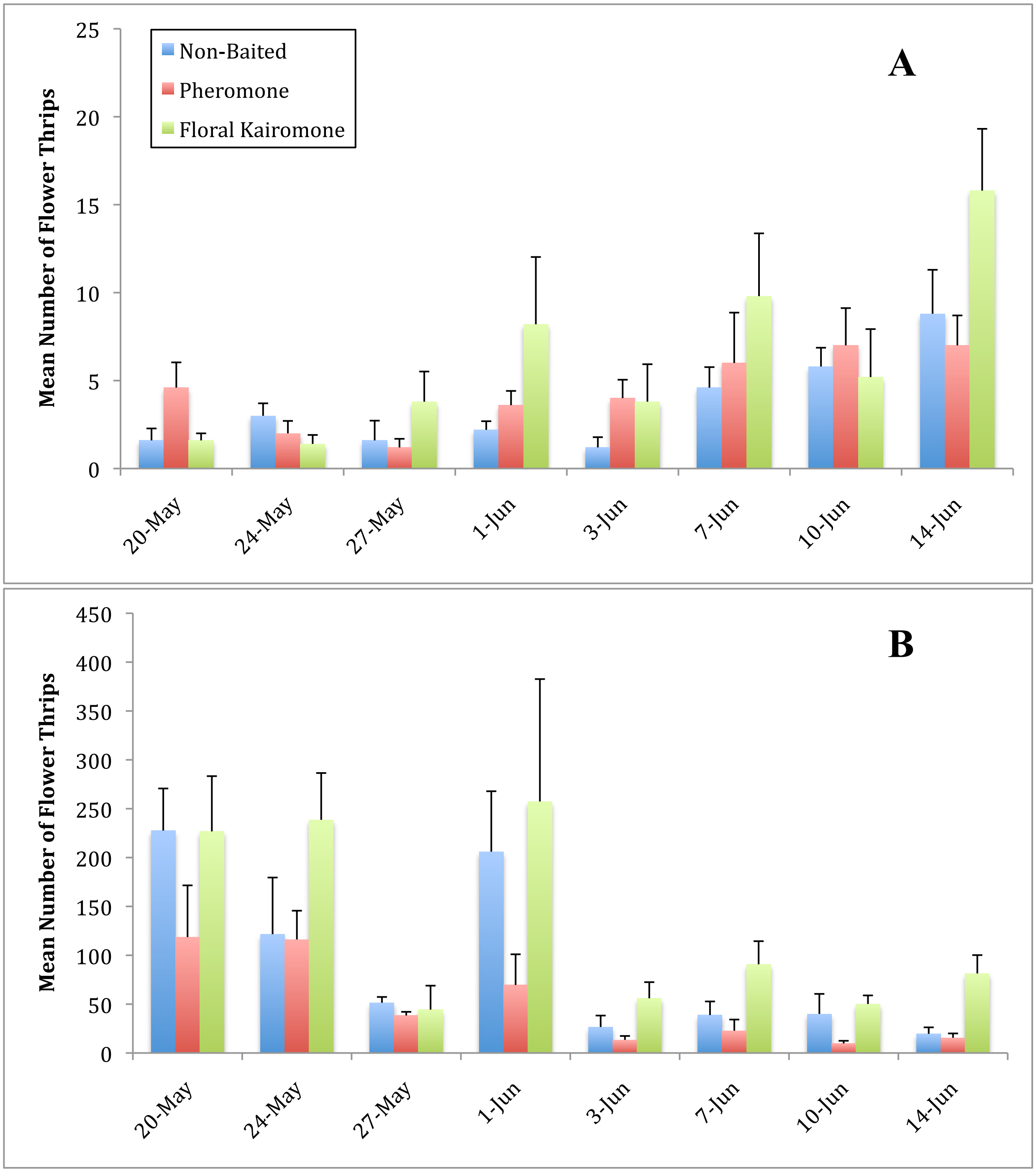


Figure 2.6. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a grass field near a high tunnel in Virginia Beach, VA (A) and in a potato field in Painter, VA (B) in 2010.

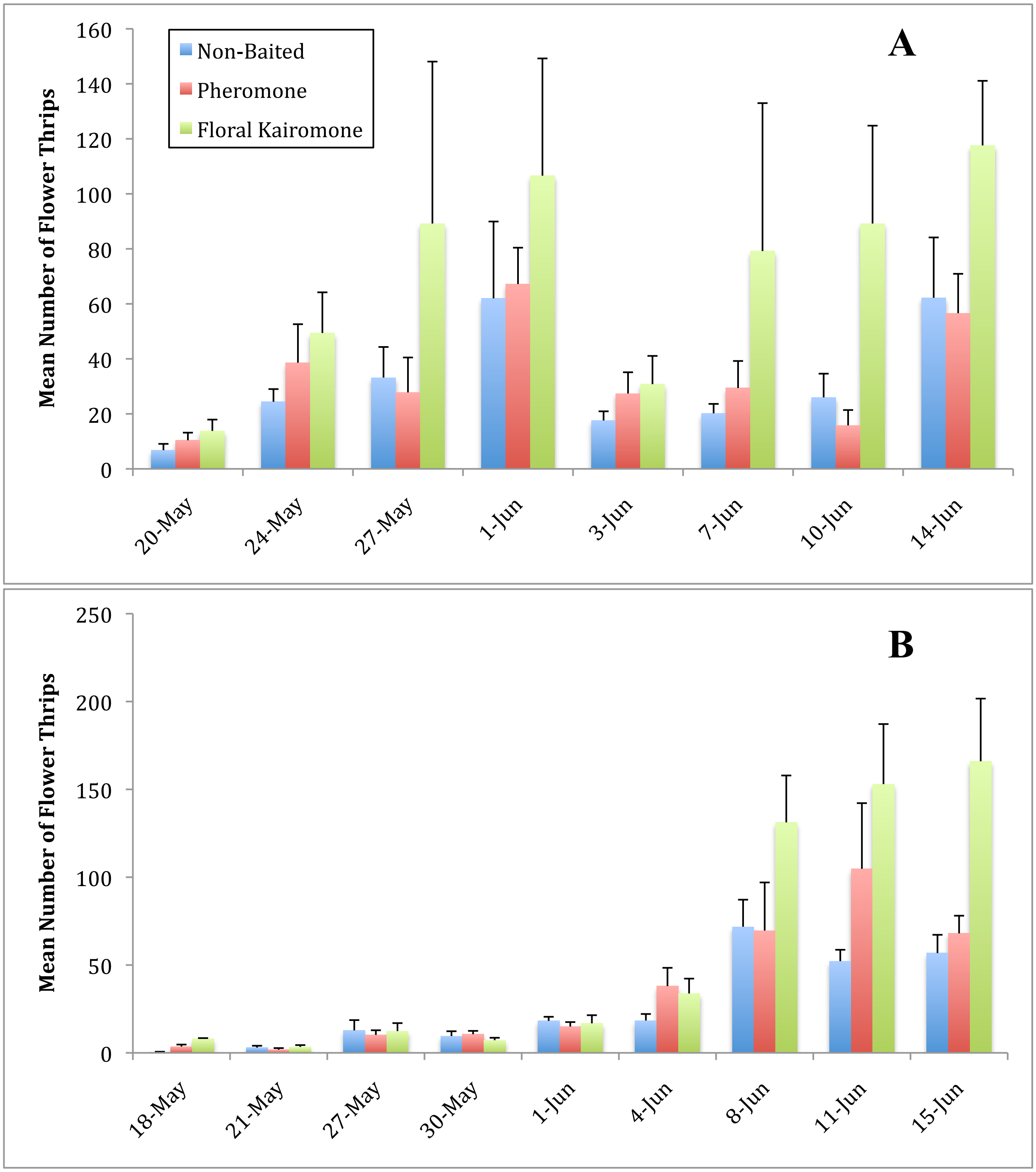


Figure 2.7. Mean ± SE catch of flower thrips (*F. occidentalis* and predominately *F. tritici*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a tomato field in Painter, VA in 2010.

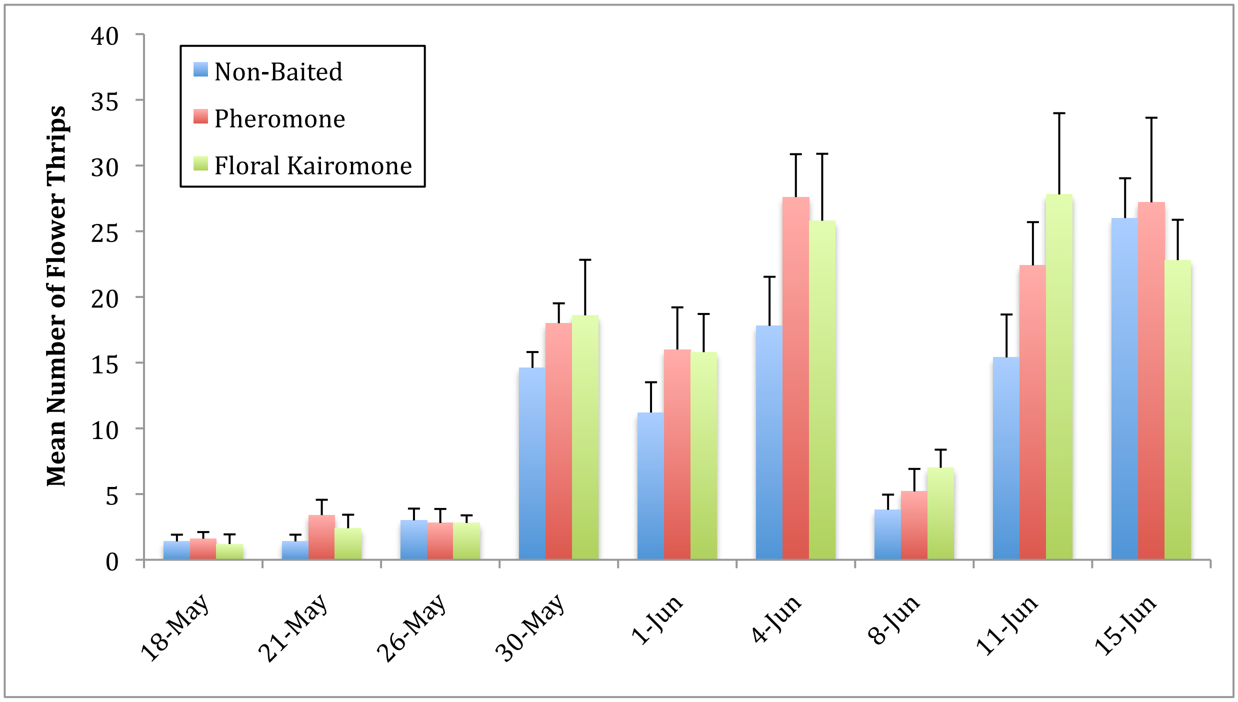
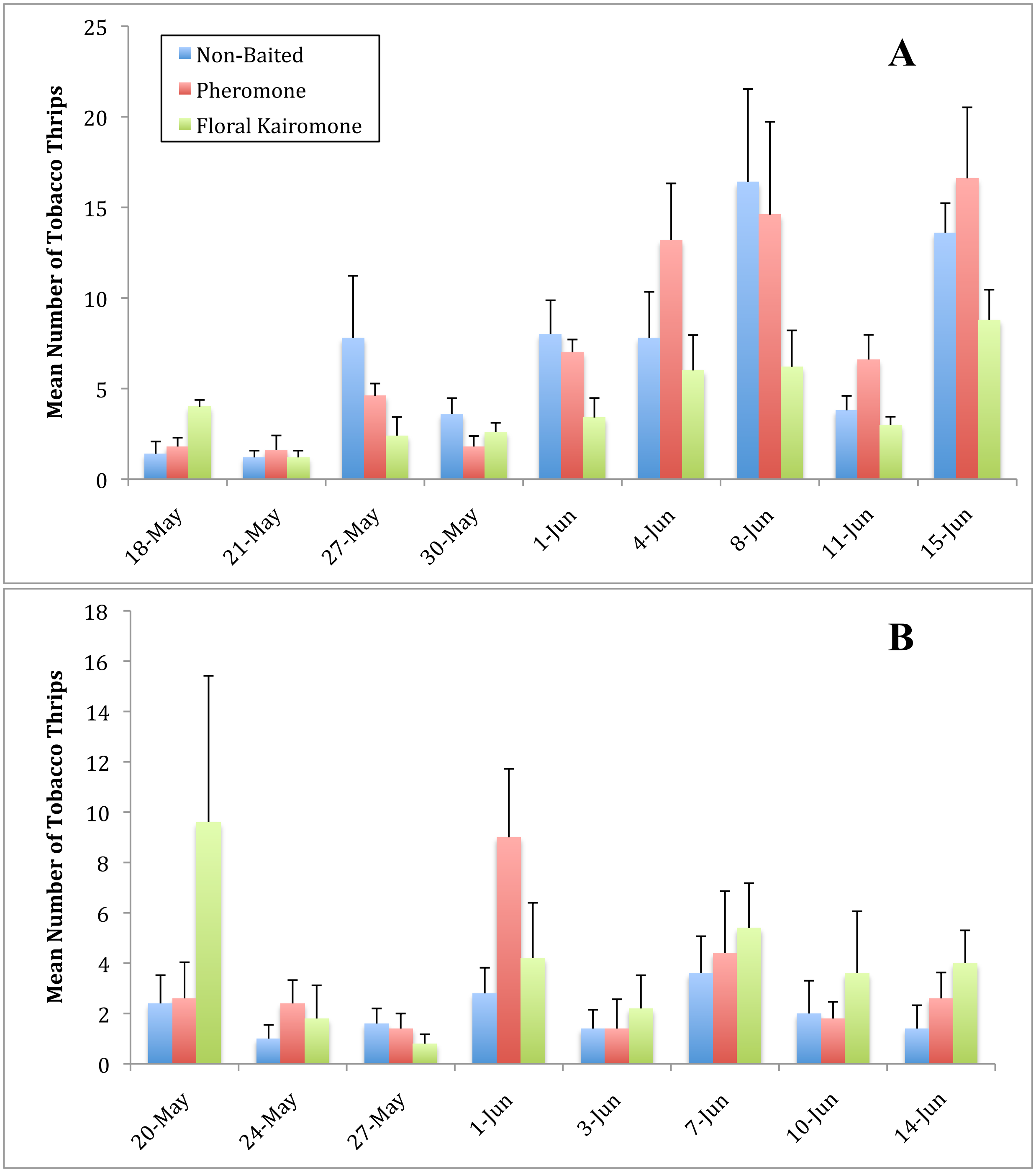


Figure 2.8. Mean SE catch of tobacco thrips (*F. fusca*) per sample date on non-baited sticky traps (Non-Baited in key) and traps baited with *F. occidentalis* pheromone (Thripline$_{AMS}$) (Pheromone in the key) and floral kairomone (Chemtica P-178) (Floral Kairomone in key) in a potato field in Painter, VA (A) and inside a greenhouse in Chesapeake, VA (B) in 2010.



Chapter 3: Efficacy of biologically derived insecticides in controlling thrips in tomato, snap beans, collards, soybeans, cotton and peanuts.

Abstract

Many naturally-derived compounds have been used as insecticides by humans for thousands of years, and recently research has focused more upon insecticides that are not only naturally-derived, but are also more specific against certain pests, thus reducing adverse effects on other organisms and allowing for the retention of natural enemies. Several biologically derived insecticides including: essential oils, spinetoram, spinosad, pyrethrins, and azadirachtin were tested in their ability to combat thrips in several different crops. Small plot field experiments were carried out in: tomatoes, snap beans, collards, soybeans, cotton and peanuts grown in southeastern Virginia in 2009 and 2010. Both spinetoram and spinosad reduced thrips numbers the most effectively compared with the untreated control. Peanut and cotton treated with spinosad, spinetoram, and spinetoram combined with essential oils had less thrips injury compared with the untreated control and yields in cotton plots treated with spinetoram were significantly higher than the untreated control.

Introduction

Thrips (Thysanoptera) are serious pests of a number of agricultural crops (Lewis 1997). In Virginia the primary pest species include tobacco thrips, *Frankliniella fusca* (Hinds), and flower thrips, *Frankliniella tritici* (Fitch), although western flower thrips, *Frankliniella occidentalis* (Pergande) occurs in low numbers. High densities of thrips and subsequent feeding injury on seedlings, flowers, pods, and fruit can cause significant yield losses in row crops such as cotton, peanuts, and vegetables, particularly tomatoes (Pohronezny et al. 1986, Childers 1997, Mound 1997, Nault et al. 2003, Herbert et al. 2007). In addition, *F. fusca* and *F. occidentalis* can transmit devastating plant pathogenic tospoviruses, such as tomato spotted wilt virus (Johnson et al. 1995, Eckel et al. 1996, Groves et al. 2001, 2002).

Despite numerous strategies that have been employed to control thrips, these insects continue to be difficult to manage. Short lifespan and production of many progeny help thrips rapidly become resistant to specific insecticides. *F. occidentalis* is more difficult to control than many other pestiferous thrips. Some insecticides, particularly pyrethroids, can actually be responsible for the buildup of *F. occidentalis* population on crops by several mechanisms. The suppression of key predators such as *Orius insidiosus* (Say) is one of the leading factors (Funderburk et al. 2000), although other mechanisms such as competition for resources (Paini et al. 2007) and even hormoligosis have been attributed to an increase in pest populations (Frantz and Mellinger 2009). Consequently, there is a need to continuously evaluate alternative strategies and pesticide chemistries to control thrips. Today, there are a wide variety of pesticides with less risk to nontarget organisms and the environment. Some of these compounds are naturally-derived from plants, fungi, or bacteria, and often, can be considered for use in integrated pest management (IPM) and organic production systems.

There are a myriad of advantages to using naturally-derived compounds to control insects, the main reason being that many such compounds have a reduced negative effect upon other organisms. Several plants produce toxins to deter animals from feeding upon them, and some are harvested and used as insecticides. Many essential oils (i.e., rosemary, peppermint, cinnamon, and wintergreen) have multiple modes of action on organisms (Chiasson et al. 2004). Certain chemicals can have a neurotoxic effect on susceptible insects; whereas, several nonneurotoxic essential oils cause antifeedent effects, disrupt molting and respiration, and reduce growth and fecundity. Some essential oils may also affect the cuticle of soft-bodied insects (Chiasson et al. 2004).

Compounds, such as azadirachtin, derived from the neem tree, have a low knockdown effect, and are compatible with many natural enemies when applied strategically to target the pest (Thoeming et al. 2003). Azadirachtin has a wide array of effects upon susceptible insects including repellency, primary and secondary antifeedancy, growth reduction, increased mortality and abnormal molts (Seymour et al. 1995, Puri 1999, Abudulai et al. 2003, Durmusoglu et al. 2003, Luntz 2004). Other plant-derived compounds include terpenes and terpenoids, which are often repellent and even toxic to certain insects including thrips. These chemicals disrupt the insect's neurotransmitters by interfering with the neuromodulator octopamine (Enan 2001, Kostyukovsky et al. 2002). Plants that produce such compounds include rosemary, mint, and many other strongly-scented plants.

Pyrethrins are a group of organic compounds produced by chrysanthemum flowers, *Chrysanthemum* spp., which exhibit insecticidal activity (Casida 1980). Pyrethrins are contact poisons that affect the insect's nervous system by delaying closure of ion channels, thereby causing multiple action potentials in nerve cells (National Pesticide Information Center 1998). Insecticidal activity can be enhanced by the addition of synergists, which reduce detoxification in the insect (Elliott and Janes 1973). These compounds are not very toxic to most terrestrial animals, however, pyrethrum is toxic to fish and many organisms eaten by fish such as crustaceans and aquatic insects (Pillmore 1973).

There are numerous pathogens and their derivatives that also can be employed as insecticides. Spinosyns are a group of insecticidal macrocyclic lactones from the fermentation product of *Saccharapolyspora spinosa*, which is a soil actinomycete (Horowitz and Ishaaya 2004). Activity against a wide variety of pests, particularly lepidopterans, thysanopterans and dipterans has been shown, and spinosyns tend to have a low impact upon the environment and low toxicity towards many nontargets (Jones et al. 2002, Cloyd et al. 2009). Spinosyns act upon the insect by exciting neurons in the central nervous system, which causes tremors and spontaneous muscle contractions, paralysis and loss of body fluids (Thompson et al. 2000).

Naturally derived compounds can also be modified to enhance insecticidal activity, and to increase persistence in the environment. Spinetoram is derived from spinosyns J and L, which have been modified to produce a semi-synthetic insecticide (Sparks et al. 2008, Huang et al. 2009). This insecticide has a longer control period and also is more active against many key pests including codling moth *Cydia pomonella* (L.), and tobacco budworm *Heliothis virescens* (F.). In addition, spinetoram has been shown to control thrips (Srivastava et al. 2008, Funderburk 2009).

With an ongoing need to develop new insecticides as thrips become resistant to current treatments, and an increase in public demand for safer more environmentally friendly compounds, it seems prudent to consider biologically derived insecticides as alternatives to many of the conventional insecticides. Such compounds are often safer for mammals including humans and many are specific enough to target primarily pest insects while allowing natural predators to persist. The purpose of this research was to evaluate the efficacy of various biologically derived insecticides in controlling thrips in tomato, snap beans, collards, soybeans, cotton and peanuts.

Materials and Methods

Field experiments were conducted in tomatoes, snap beans, collards and soybeans grown at the Eastern Shore Agricultural Research and Extension Center (ESAREC) in Painter, VA in 2009. In 2010, similar studies were conducted in soybeans at the ESAREC, and on peanuts and cotton grown at the Tidewater Agricultural Research and Extension Center (TAREC) in Suffolk, VA.

All crops planted in 2009 for this study were maintained according to standard commercial practices. Each crop was arranged in a randomized complete block design with four replications. Foliar treatments were applied with a three-nozzle boom equipped with D3 tips and 45 cores powered by a CO_2 backpack sprayer at 28,123 kg/m^2 delivering 355.5 L/ha. Insecticides used for all experiments in 2009 and in 2010 were identical and are listed in Table 3.1. On selected dates, a sample of approximately 50 thrips was collected in the field and preserved in a 70% alcohol solution to determine species composition in each crop. Thrips were identified to species using taxonomic keys found in Capinera (2001).

Tomatoes

Seedling tomatoes variety 'BHN602' were transplanted 26 May 2009 into plots that were 6.10 m long with 1.83 m long row centers. Tomato plots were treated 11 and 25 June and 2, 8, 15, 22 and 29 July, 2009.

Ten compound leaves were randomly selected from each plot on 17 June 2009 (6 days after the first treatment [6 DAT1]), placed in 3-liter-size resealable plastic bags, and washed with a soapy solution in the lab. Leaves were shaken to remove excess water, and then discarded. The remaining solution was filtered using a Büchner funnel and presence of thrips was examined under the dissecting microscope. The number of adult and larval thrips was recorded. On 3 July

2009 (1 DAT3) and 13 July 2009 (5 DAT4), 20 blossoms were randomly selected from each plot and processed in the same manner as the leaves. The number of adult and larval thrips was then recorded after viewing under a dissecting microscope. Thirty fruit were harvested from each plot on 17 August 2009 (19 DAT7) and examined for thrips injury such as gold-flecking and dimpling.

Snap Beans

Snap beans variety 'Hystyle' were planted 23 May 2009 in two 6.10 m rows (0.91 m row centers) with no guard rows. Foliar treatments were applied on 16, 24 June, 2 and 8 July 2009.

On 18 June 2009 (2 DAT1) ten trifoliate leaves were collected from each plot, placed into 3-liter-size resealable plastic bags with a 70% ethyl alcohol solution, and shaken to dislodge thrips. Leaves were removed from the bag and contents were filtered using a Büchner funnel. The number of adult and larval thrips present was counted for each plot using a dissecting microscope. Twenty blossoms were collected from each plot on 2 July 2009 (8 DAT2) and were processed in the same manner. Adult and larval thrips were counted under a dissecting microscope and recorded. On 15 July 2009 (7 DAT4) all marketable pods were harvested and weighed. A sub-sample of 50 pods was examined for thrips injury in the form of scarring and deformed pods.

Collards

Collards variety 'Vates' were planted 23 May 2009 in 6.10 m rows (0.91 m row centers) with planted guard rows. Plots were treated on 29 June and 10 July 2009.

Twenty leaves collected from each plot on 3 July 2009 (4 DAT1) and 14 July 2009 (4 DAT2) were placed in 3-liter-size resealable plastic bags containing a 70% alcohol solution. Bags were shaken to dislodge thrips and leaves were removed. The solution was then filtered using a Büchner funnel and thrips were counted under a dissecting microscope. The number of adult and larval thrips was recorded for each plot.

Soybeans

In 2009, soybeans variety 'NK S48-C9' were planted on 3 June 2009 and arranged in 6.10 m rows (0.91 m row centers) with guard rows. Plots were sprayed on 3 and 10 August 2009.

On 7 August 2009 (4 DAT1) and 13 August 2009 (3 DAT2) 20 trifoliates were collected from each plot, placed in 3-liter-size resealable plastic bags containing a 70% alcohol solution and shaken. Leaves were removed from the bags and contents were filtered using a Büchner funnel. The number of adult and larval thrips was counted using a dissecting microscope.

In 2010, soybeans variety 'NK S48-C9' were planted on 5 June 2010 and were arranged in a similar pattern as the year before. Plots were treated with foliar sprays on 21 June 2010 using the same spray equipment and settings as the previous year.

On 23 June 2010 (2 DAT1), 28 June 2010 (7 DAT1), and 2 July 2010 (11 DAT1), ten trifoliates were collected from each plot and placed into liter- size resealable plastic bags. In the lab samples were processed in an identical manner to the previous year.

Cotton

Cotton variety 'DP 0920B2RF' was planted 5 May 2010 at the TAREC in Suffolk, VA. Plots were arranged in a randomized complete block design with four replications and rows were spaced one meter apart. All plots were maintained according to standard commercial practices and foliar treatments were applied at 133.8 L/ha using a three-nozzle-boom equipped with 8002VS nozzles and powered by a CO_2 backpack sprayer at 12,655 kg/m^2. Treatments were applied 21 and 27 May 2010.

Five seedling cotton plants were randomly collected from each plot on 25 May 2010 (4 DAT1), 1 June 2010 (5 DAT2) and 8 June 2010 (12 DAT2). Stems were cut near the ground and plants were placed in liter-size glass jars with metal screw top lids approximately half-filled with a mild soapy water solution. In the lab, the jars were shaken to dislodge thrips, and plants were then removed using tweezers. The remaining solution was filtered using a Büchner funnel and adult and larval thrips were counted under a dissecting microscope. On 1 and 8 June, plants were assessed for thrips injury. Injury ratings for each plot were conducted using the following criteria: 0= no injury; 1= 10% injured leaves, no bud injury; 2= 25% injured leaves, no bud injury; 3= 75% injured leaves, 0-25% buds injured; 4= 90% injured leaves, >25% buds injured; 5= dead plants. On 12 October (138 DAT2) cotton was harvested using a two-row commercial picker modified for research plots. Four samples were ginned to determine a mean of 45.05% lint and 54.96% seed and trash.

Peanut

Peanuts variety 'CHAMPS' were planted 29 April 2010 at the TAREC in Suffolk, VA. Plots were arranged in a randomized complete block design with four replications and rows spaced one meter apart. All plots were maintained according to standard commercial practices and foliar treatments were applied at 133.8 L/ha using a three-nozzle-boom equipped with 8002VS nozzles and powered by a CO_2 backpack sprayer at 12,655 kg/m^2. Treatments were applied 20 and 27 May 2010.

Ten newly-opened leaflets were randomly selected from each plot on 25 May 2010 (5 DAT1), 1 June 2010 (5 DAT2) and 8 June 2010 (12 DAT2). Leaves were placed into small plastic bottles with screw top lids, and were half-filled with soapy water. In the lab, the bottles were shaken to dislodge thrips, and leaves were removed using forceps. The remaining solution was filtered using a Büchner funnel. Adult and larval thrips were counted using a dissecting microscope. On 1 and 8 June, plants were assessed for thrips injury. Rating values were given to each plot using the following guidelines: 0= no injury; 1= 10% leaves injured; 2= 20% leaves injured; 3= 30% leaves injured; 4= 40% leaves injured; 5= $\geq$ 50% leaves injured + $\leq$ 5% terminal buds injured; 6= $\geq$ 50% leaves injured + 25% terminal buds injured; 7= $\geq$ 50% leaves injured + 50% terminal buds injured; 8= $\geq$ 50% leaves injured + 75% terminal buds injured; 9= $\geq$ 50% leaves injured + 90% terminal buds injured; 10= dead plants. Peanut plants were dug on 23 September and allowed to remain exposed to environmental conditions until 6 October (132 DAT2), when peanuts were then harvested.

Data Analysis-

Data for all experiments were analyzed using analysis of variance procedures and means were separated using Tukey's HSD at the 0.05 level of significance (JMP version 8.0.1, SAS Institute Inc. 2009).

Results

Tomatoes

Leaf samples collected 17 June, yielded a thrips species complex composed of 71% *F. fusca*, 18% *F. tritici* and 11% *F. occidentalis*. *F. tritici* comprised approximately 80% of the species complex found in blossom samples collected on 2 and 13 July 2009. Adult thrips populations were significantly reduced by all treatments except azadirachtin on 17 June 2009 (Table 3.2). There was no significant treatment effect on thrips numbers on any other sample date. At harvest, gold-flecking and dimpling from thrips injury was variable and generally low (8.3% in the untreated control) compared with previous years at the ESAREC (Table 3.2). Plots treated with azadirachtin had very little thrips injury on the fruit. There were no signs of phytotoxicity, which can result from using certain plant-derived compounds.

Snap Beans

The species complex present in snap beans on 2 July 2009 was comprised of 64% *F. tritici*, 20% *F. occidentalis*, 10% *F. fusca*, 4% *Neohydatothrips variabilis* (Beach) and 2% *Thrips tabaci* Lindeman. On 18 June 2009, there was a statistically significant treatment effect on leaf counts of both larvae and adults (Table 3.3). All insecticide treatments significantly reduced thrips larvae compared with the control. Adult thrips counts were lower in plots treated with pyrethrins. On 2 July 2009, none of the treatments significantly reduced thrips populations compared with the untreated control. Although bean yield based on pod weight was highly variable, there does not appear to be a correlation between treatment and yield variation. Yield was likely impacted by weeds and a poor stand establishment instead. Injury was probably caused by stink bugs, which had been found in the plots as the pods were forming, instead of thrips.

Collards

A species complex consisting of 54% *F. fusca*, 26% *T. tabaci*, 18% *F. occidentalis* and 2% *F. tritici* was present on 3 July 2009 according to a subsample. None of the treatments significantly reduced thrips populations compared with the control on either of the sample dates (Table 3.4).

Soybeans

Samples collected 7 August 2009 revealed a species complex consisting of 94% *N. variabilis*, 4% *F. occidentalis,* and 2% *T. tabaci*. On 7 and 14 August 2010, none of the treatments provided a statistically significant reduction in thrips numbers compared with the untreated control (Table 3.5). A leaf sample collected on 28 June 2010 revealed a complex comprised of 68% *F. fusca*, 18% *T. tabaci* 12% *N. variabilis,* and 2% *F. tritici*. There was a significant treatment effect on numbers of thrips on 28 June 2010 (Table 3.6). Immature thrips populations were reduced by treatments of essential oils combined with spinetoram, spinetoram alone, and spinosad, compared with the untreated control.

Cotton

Samples collected 5 May 2010 indicated a thrips species complex composed of 88% *F. fusca*, 6% *F. tritici*, 2% *F. occidentalis*, 2% *T. tabaci* and 2% *N. variabilis*. None of the treatments significantly reduced thrips numbers compared with the untreated control on any of the sample dates (Table 3.7). However, some treatments reduced thrips seedling injury. On both 1 and 8 June 2010 plots treated with essential oils combined with spinetoram, spinetoram alone, and spinosad had significantly less thrips injury compared with the control. Plots treated with spinetoram also had a significantly higher yield compared with the untreated control plots (Table 3.7)

Peanut

A sample collected 5 May 2010 indicated that there was a thrips species complex composed of 86% *F. fusca*, 10% *T. tabaci,* 2% *F. occidentalis*, and 2% *N. variabilis*. On 5 May 2010, larval thrips numbers were extremely low. None of the treatments significantly reduced thrips populations on any of the sample dates, although plots treated with essential oils combined with spinetoram and spinetoram alone had less injury compared with the control on 1 and 8 June 2010 (Table 3.8). Plants treated with spinosad also had less injury on 1 June 2010. Yield was not affected by any of the treatments compared with the untreated control (Table 3.8).

Discussion

During this study, both spinetoram and spinosad reduced thrips numbers the most effectively compared with the untreated control. Population surges did occur in certain cases, perhaps as a result of immigrating adult thrips, or large numbers of eggs hatching after treatments were applied. A few treatments also had an effect on thrips injury in crops that were assessed. Cotton and peanut plots treated with spinosad, spinetoram, and spinetoram combined with essential oils, suffered less thrips injury compared with the control, and cotton plots treated with spinetoram had a higher yield than the untreated control. It is also interesting to note that when the tomatoes were assessed for thrips injury, fruit collected from plots treated with azadirachtin had very little thrips injury. This could be due to the fact that azadirachtin does not immediately kill thrips, but rather induces antifeedancy and molting inhibition, and therefore, although thrips were present, they were perhaps unable to feed as a result of the treatment.

Both spinosad and spinetoram tend to be fairly specific against pests and have been shown to control several common species, including *F. occidentalis*, with minimal negative effects on nontarget organisms and key natural enemies, such as *O. insidiosus* (Jones et al. 2005, Srivastava et al. 2008, Cloyd et al. 2009). Spinetoram shares many qualities with spinosad because its structure is based upon the bacterial fermentation product of *S. spinosa*, and has improved insecticidal activity, longer duration of control, and targets a wider range of pests (Dripps et al. 2008, Huang et al. 2009). The number of insecticide treatments needed to lower pest populations can potentially be reduced by applying a specific insecticide, such as spinosad or spinetoram, because natural enemy populations are allowed to build and help combat the pest (Jones et al. 2002, Reitz et al. 2003). This is particularly important in IPM where one of the main goals is to reduce insecticide usage.

Because of their cryptic lifestyles, thrips are capable of evading spray applications by hiding among foliage and blossoms, and macropterous adults can further escape treatments by flying (Thoeming et al. 2003). This is one of the main challenges in effective thrips treatment with topical insecticides because the insects must come into contact with an adequate amount of insecticide in order for treatment to be effective. Many naturally-derived plant extracts have very short residual activity, and degrade rapidly under environmental conditions (Barry et al. 2005, Cloyd and Chiasson 2007). It is therefore important for the insecticide to either contact the insect during application, or immediately following treatment. Efficacy of some insecticides can be improved via different application methods. For example, systemic insecticides can be particularly effective against sucking insects such as thrips because the insecticides do not need to come into contact with the insect. Thoeming et al. (2003) found that neem was effective against *F. occidentalis* when used as a systemic insecticide on green beans. Contact mortality and repellent effects on soil-inhabiting stages were also observed following soil applications of neem.

Thrips adults tend to be highly vagile, and can therefore quickly re-establish populations in treated crops. Those that have been treated with short-residual insecticides could be re-colonized outside of the effective time period by incoming thrips, resulting in a population build-up that may not reflect the efficacy of the insecticide. This likely occurred in some treated plots, particularly when sampling was performed several days after the initial treatment.

Spinosad and spinetoram reduced thrips populations on many sample dates, although the results were often not statistically significant compared with the control. Injury ratings in the peanut and cotton plots appeared to reflect the efficacy of these two insecticides with a significant reduction in thrips injury. Cotton plots treated with spinetoram also had a significantly higher yield compared with the control. In addition, some insecticides such as azadirachtin may prove to be more effective when applied using methods other than foliar sprays, such as soil or systemic treatments.

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Table 3.1. List of insecticides, active ingredients, trade names, manufacturer, and rates used on crops in thrips efficacy experiments conducted in Virginia in 2009 and 2010.

Insecticides	**Active Ingredients**	**Trade Name**	**Manufacturer**	**Product Rate/Hectare**
Essential oils	10% rosemary oil, 2% peppermint oil, 88% wintergreen oil, lactic acid, n-butyl ester, vanillin and lecithin	Ecotec®	Brandt Consolidated, Inc., Springfield, IL	1.17 L/ha
Spinetoram	11.7% spinosyn-J and spinosyn-L	Radiant™ Insecticide	Dow AgroSciences LLC, Indianapolis, IN	0.44 L/ha
Spinosad	80% spinosyn A and spinosyn D	Entrust® Naturalyte Insect Control	Dow AgroSciences LLC, Indianapolis, IN	0.21 kg/ha
Pyrethrins	1.40% pyrethrins	PyGanic® Crop Protection EC 1.4 II	McLaughlin Gormley King Company, Minneapolis, MN	2.34 L/ha
Azadirachtin	1.2% azadirachtin	Aza-Direct®	Gowan Company, Yuma, AZ	4.09 L/ha

Table 3.2. Efficacy of insecticides tested against thrips in tomatoes grown at the ESAREC in 2009. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05). Percent thrips fruit injury at harvest was based on a sample of 30 fruit per plot.

Tomatoes (3 sprays)

Treatment	Number of thrips per 10 compound leaves 6/17 (7 DAT1)		Number of thrips per 20 flowers 7/3 (1 DAT3)		Number of thrips per 20 flowers 7/13 (5 DAT4)		% thrips fruit injury at harvest
	Imm.	Adults + Imm.	Imm.	Adults + Imm.	Imm.	Adults + Imm.	
Untreated control	6.00 ± 1.87 ab	10.25 ± 1.89 ab	0.25 ± 0.25	5.50 ± 2.22	0.75 ± 0.48	12.00 ± 3.58 ab	8.30 ± 0.02 abcd
Essential oils	3.00 ± 1.22 ab	3.75 ± 1.49 ab	0.00 ± 0.00	7.25 ± 2.71	1.25 ± 0.48	15.50 ± 1.44 ab	15.83 ± 0.04 a
Spinetoram	0.75 ± 0.48 b	2.00 ± 0.71 b	0.00 ± 0.00	6.50 ± 1.32	0.25 ± 0.25	7.00 ± 2.74 ab	3.33 ± 0.02 cd
Essential oils + spinetoram	0.00 ± 0.00 b	1.00 ± 0.41 b	0.50 ± 0.50	7.25 ± 2.25	0.00 ± 0.00	8.75 ± 0.48 ab	12.50 ± 0.03 abc
Spinosad	0.75 ± 0.25 b	1.00 ± 0.41 b	0.00 ± 0.00	5.00 ± 1.08	0.25 ± 0.25	5.75 ± 2.14 b	4.17 ± 0.02 bcd
Pyrethrins	16.00 ± 6.38 a	16.75 ± 6.70 a	0.50 ± 0.50	8.00 ± 0.71	0.75 ± 0.48	12.75 ± 1.25 ab	15.00 ± 0.03 ab
Azadirachtin	10.50 ± 3.01 ab	13.50 ± 3.48 ab	0.00 ± 0.00	4.50 ± 0.96	0.00 ± 0.00	13.50 ± 5.63 ab	0.83 ± 0.01 d
p-value	0.0055	0.0049	NS	NS	NS	NS	0.0012

Table 3.3. Efficacy of insecticides tested against thrips in snap beans grown at the ESAREC in 2009. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

Snap Beans (2 sprays)					
Treatment	**Number of thrips per 10 trifoliate leaves 6/18 (2 DAT1)**		**Number of thrips per 20 flowers 7/2 (8 DAT2)**		**Yield (weight in g.)**
	Imm.	**Adults + Imm.**	**Imm.**	**Adults + Imm.**	
Untreated control	8.75 ± 1.25 a	16.25 ± 1.31 a	1.25 ± 0.25 ab	4.00 ± 0.41 a	452.50 ± 138.10
Essential oils	3.50 ± 0.65 b	6.00 ± 1.55 b	2.00 ± 0.00 a	5.25 ± 1.60 a	265.00 ± 63.97
Spinetoram	0.75 ± 0.48 b	2.75 ± 1.00 b	0.25 ± 0.25 b	1.75 ± 0.75 a	515.00 ± 150.91
Essential oils + spinetoram	0.50 ± 0.29 b	2.00 ± 0.00 b	0.75 ± 0.48 ab	2.50 ± 1.50 a	592.50 ± 157.61
Spinosad	0.75 ± 0.25 b	2.25 ± 0.87 b	0.25 ± 0.25 b	1.75 ± 0.63 a	545.50 ± 51.88
Pyrethrins	1.00 ± 1.00 b	2.25 ± 0.63 b	1.00 ± 0.41 ab	3.00 ± 0.41 a	542.50 ± 131.24
Azadirachtin	1.25 ± 1.25 b	3.50 ± 0.85 b	0.75 ± 0.25 ab	3.75 ± 1.31 a	572.50 ± 124.32
p-value	<0.0001	< 0.0001	0.0046	0.0549	NS

Table 3.4. Efficacy of insecticides tested against thrips in collards grown at the ESAREC in 2009. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

Collards (2 sprays)				
Treatment	**Number of thrips per 20 leaves 7/3 (4 DAT1)**		**Number of thrips per 20 leaves 7/14 (4 DAT2)**	
	Imm.	**Adults + Imm.**	**Imm.**	**Adults + Imm.**
Untreated control	5.00 ± 2.04	11.75 ± 2.63 ab	5.25 ± 2.06	12.25 ± 3.89
Essential oils	3.75 ± 2.50	13.00 ± 3.54 a	5.25 ± 2.18	12.25 ± 4.67
Spinetoram	0.25 ± 0.25	0.75 ± 0.48 b	0.00 ± 4.99	0.50 ± 6.44
Essential oils + spinetoram	0.00 ± 0.00	0.25 ± 0.25 b	0.00 ± 0.48	6.50 ± 1.44
Spinosad	1.00 ± 1.00	3.75 ± 3.75 ab	0.00 ± 1.80	0.75 ± 4.29
Pyrethrins	1.75 ± 0.75	13.00 ± 0.41 a	7.25 ± 1.75	9.50 ± 3.28
Azadirachtin	3.50 ± 1.32	7.00 ± 1.44 ab	0.00 ± 0.85	2.00 ± 1.25
p-value	NS	0.0039	NS	NS

Table 3.5. Efficacy of insecticides tested against thrips in soybeans grown at the ESAREC in 2009. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

Soybeans (2 sprays)				
Treatment	**Number of thrips per 20 trifoliates 8/7 (4 DAT1)**		**Number of thrips per 20 trifoliates 8/13 (3 DAT2)**	
	Imm.	**Adults + Imm.**	**Imm.**	**Adults + Imm.**
Untreated control	10.5 ± 4.37	17.75 ± 4.15	8.50 ± 4.37	10.00 ± 5.55
Essential oils	10.50 ± 5.12	14.50 ± 5.72	2.50 ± 1.19	5.00 ± 1.96
Spinetoram	1.25 ± 0.25	5.25 ± 1.49	1.50 ± 1.50	3.50 ± 1.19
Essential oils + spinetoram	1.25 ± 0.25	6.25 ± 1.49	5.00 ± 4.34	7.50 ± 5.52
Spinosad	5.25 ± 1.60	11.00 ± 1.00	1.50 ± 0.96	3.00 ± 0.82
Pyrethrins	9.25 ± 3.71	17.25 ± 6.37	19.50 ± 9.61	20.50 ± 10.08
Azadirachtin	11.75 ± 4.33	20.25 ± 6.63	12.75 ± 6.73	14.75 ± 7.03
p-value	NS	NS	NS	NS

Table 3.6. Efficacy of insecticides tested against thrips in soybeans grown at the ESAREC in 2010. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

Soybeans (1 spray)						
Treatment	Number of thrips per 20 trifoliates 6/23 (2 DAT1)		Number of thrips per 20 trifoliates 6/28 (7 DAT1)		Number of thrips per 20 trifoliates 7/2 (11 DAT1)	
	Imm.	Adults + Imm.	Imm.	Adults + Imm.	Imm.	Adults + Imm.
Untreated control	25.75 ± 6.45 ab	34.50 ± 5.98 ab	67.75 ± 7.86 a	78.75 ± 8.84 a	11.50 ± 4.94 ab	44.00 ± 10.79 a
Essential oils	24.25 ± 6.64 ab	30.50 ± 7.31 ab	73.50 ± 19.30 a	90.00 ± 21.46 a	24.25 ± 7.89 a	43.50 ± 8.85 a
Spinetoram	5.75 ± 2.95 b	10.75 ± 3.25 b	6.50 ± 3.40 b	12.50 ± 6.02 b	2.50 ± 2.18 b	15.50 ± 6.74 a
Essential oils + spinetoram	12.00 ± 5.87 ab	16.00 ± 7.19 b	3.25 ± 0.25 b	9.75 ± 1.03 b	6.50 ± 4.84 ab	16.25 ± 6.17 a
Spinosad	6.00 ± 1.47 b	8.50 ± 0.65 b	5.75 ± 1.55 b	13.75 ± 3.35 b	1.00 ± 0.71 b	9.00 ± 1.58 a
Pyrethrins	22.50 ± 9.26 ab	29.75 ± 8.99 ab	44.50 ± 15.95 ab	55.50 ± 16.59 ab	9.25 ± 3.45 ab	33.50 ± 13.19 a
Azadirachtin	37.50 ± 6.38 a	47.50 ± 6.79 a	66.00 ± 16.03 a	84.00 ± 18.33 a	10.00 ± 4.02 ab	29.50 ± 6.29 a
p-value	0.0142	0.0041	0.0005	0.0005	0.0461	0.0398

Table 3.7. Efficacy of insecticides tested against thrips in cotton grown at the TAREC in 2010. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

	Cotton (2 sprays)								
Treatment	**Number of thrips per 5 seedlings 5/25 (4 DAT1)**		**Number of thrips per 5 seedlings 6/1 (5 DAT2)**			**Number of thrips per 5 seedlings 6/8 (12 DAT2)**			**Yield (Unginned) 10/12 (harvest)**
	Imm.	**Adults + Imm.**	**Imm.**	**Adults + Imm.**	**Dmg. Rating**	**Imm.**	**Adults + Imm.**	**Dmg. Rating**	**(kg per 21.34 m row)**
Untreated control	15.25 ± 5.63	23.25 ± 4.91 a	43.50 ± 16.84 ab	46.25 ± 16.80 ab	4.25 ± 0.18 a	29.75 ± 8.29 ab	47.75 ± 10.36 ab	4.31 ± 0.19 a	4.47 ± 0.37 bc
Essential oils	13.25 ± 2.12	20.25 ± 6.38 a	65.00 ± 14.66 ab	67.50 ± 14.92 ab	4.38 ± 0.30 a	25.25 ± 9.38 ab	39.50 ± 11.86 ab	4.06 ± 0.26 a	4.48 ± 0.43 abc
Spinetoram	6.00 ± 0.65	10.50 ± 2.63 a	10.50 ± 2.33 ab	12.25 ± 1.93 ab	1.56 ± 21 b	6.50 ± 2.72 b	18.25 ± 4.55 ab	2.13 ± 0.13 b	7.06 ± 0.94 a
Essential oils + spinetoram	3.25 ± 0.48	5.75 ± 0.63 a	5.75 ± 1.80 b	9.50 ± 3.12 b	1.44 ± 0.16 b	6.75 ± 2.56 ab	14.75 ± 1.70 b	1.06 ± 0.26 b	6.61 ± 0.37 ab
Spinosad	7.75 ± 1.22	10.75 ± 3.90 a	38.50 ± 27.22 ab	41.50 ± 27.88 ab	1.38 ± 0.30 b	10.50 ± 1.19 ab	19.50 ± 2.33 ab	1.69 ± 0.31 b	6.68 ± 0.56 ab
Pyrethrins	11.25 ± 2.29	22.00 ± 4.78 a	56.25 ± 12.40 ab	60.00 ± 12.42 ab	3.75 ± 0.10 a	37.50 ± 8.72 a	55.75 ± 11.67 a	4.06 ± 0.21 a	4.61 ± 0.22 abc
Azadirachtin	7.50 ± 2.84	19.75 ± 5.34 a	79.25 ± 19.82 a	83.00 ± 19.99 a	4.19 ± 0.12 a	37.25 ± 7.85 ab	55.25 ± 10.62 a	3.44 ± 0.43 a	3.82 ± 0.53 c
p-value	NS	0.0367	0.0250	0.0271	<0.0001	0.0065	0.0065	<0.0001	0.0014

Table 3.8. Efficacy of insecticides tested against thrips in peanuts grown at the TAREC in 2010. All data analyzed using analysis of variance procedures. Means were separated using Tukey's HSD at the 0.05 level of significance. Means followed by the same letter within a column are not significantly different (P<0.05).

Peanut (2 sprays)

Treatment	Number of thrips in 10 leaflets 5/25 (5 DAT1)		Number of thrips in 10 leaflets 6/1 (5 DAT2)			Number of thrips in 10 leaflets 6/8 (12 DAT2)			Yield 10/6 (harvest)
	Imm.	Adults + Imm.	Imm.	Adults + Imm.	Dmg. Rating	Imm.	Adults + Imm.	Dmg. Rating	(kg per 24.38 m row)
Untreated control	0.75 ± 0.75 b	13.00 ± 0.91	57.00 ± 26.25 ab	57.00 ± 26.25 ab	6.50 ± 0.00 a	3.75 ± 1.44	5.00 ± 1.78	7.00 ± 0.00 a	9.87 ± 0.51 abc
Essentail oils	4.25 ± 1.11 ab	18.00 ± 4.92	78.75 ± 19.07 ab	80.00 ± 18.73 ab	6.50 ± 0.00 a	1.50 ± 0.65	3.00 ± 1.08	6.88 ± 0.13 a	10.65 ± 0.76 abc
Spinetoram	11.25 ± 2.02 a	32.25 ± 6.05	13.00 ± 5.12 ab	14.00 ± 5.12 ab	4.13 ± 0.22 b	2.00 ± 1.08	3.75 ± 0.18	4.38 ± 1.09 b	12.17 ± 0.30 a
Essentail oils + spinetoram	3.00 ± 1.35 b	14.00 ± 6.92	6.50 ± 5.74 b	8.00 ± 5.74 b	2.44 ± 0.39 c	3.25 ± 0.63	5.75 ± 1.11	2.13 ± 0.13 c	11.63 ± 0.53 ab
Spinosad	3.50 ± 1.94 b	12.75 ± 3.25	8.75 ± 1.75 b	9.25 ± 2.25 b	4.69 ± 0.37 b	1.25 ± 0.48	2.00 ± 0.58	6.50 ± 0.35 a	11.02 ± 0.43 abc
Pyrethrins	4.00 ± 2.27 ab	16.75 ± 3.95	53.50 ± 11.81 ab	54.00 ± 11.75 ab	6.50 ± 0.00 a	4.25 ± 2.17	5.00 ± 2.31	7.25 ± 0.14 a	9.71 ± 0.45 bc
Azadirachtin	1.50 ± 0.29 b	12.50 ± 1.19	81.50 ± 16.82 a	82.75 ± 16.94 a	6.50 ± 0.00 a	4.25 ± 1.80	6.00 ± 2.16	7.13 ± 0.13 a	9.11 ± 1.14 c
p-value	0.0053	NS	0.0059	0.0059	<0.0001	NS	NS	<0.0001	0.0037

Conclusion

The overall objective of this study was to monitor several pestiferous thrips species, particularly the newly introduced *Frankliniella occidentalis* (Pergande), in eastern Virginia in a wide range of agroecosystems, and to evaluate the efficacy of a variety of biologically derived insecticides in their ability to control thrips in myriad crops grown in the region.

In 2008 and 2009, early spring weeds were sampled for the presence of *F. occidentalis* to determine if this recently introduced species is able to overwinter in southeastern Virginia. Early flowering weeds, consisting primarily of mustard, henbit and wild radish were collected from several sites on the Eastern Shore of Virginia and examined for thrips. Moreover, in 2008, 2009 and 2010, several agroecosystems were sampled for the relative incidence of *F. occidentalis*. In 2009, *F. occidentalis* was detected in early spring weed samples, indicating that it is able to overwinter in this region. Thrips populations in 2008 on tomato, potato and grassy fields consisted mainly of *Frankliniella tritici* (Fitch) and *Frankliniella fusca* (Hinds), with *F. occidentalis* composing 1 to 7% of the total number of thrips. In 2009, *F. occidentalis* remained present but in low numbers at most sites except in the tomatoes in Painter, VA, where they were not found during this year. In 2010 *F. occidentalis* populations dropped further and this species was not present in the soybeans in Painter, VA and the grass field in Virginia Beach, VA.

In May and June 2009 and 2010, two lures were evaluated in their attractiveness to *Frankliniella* spp. thrips. The lures were Chemtica P-178 floral kairomone (AgBio Inc., Westminster, CO), a floral kairomone lure composed of a proprietary floral compound mixture, and $Thripline_{AMS}$ (Syngenta Bioline Ltd., Oxnard, CA) pheromone lure, containing the aggregation pheromone of *F. occidentalis*. Lure experiments were conducted in a tomato and potato field in Painter, VA, a cotton and peanut field in Suffolk, VA, grass fields near a greenhouse in Chesapeake, VA and high tunnel in Virginia Beach, VA, as well as within the structures. Baited and non-baited sticky cards were used to monitor thrips catches, and a pan trap was used for collecting thrips for species identification. Traps were changed approximately twice per week and sampling lasted for several weeks each year from mid-May to mid-June. Overall *F. fusca* densities were low and neither lure had a significant effect on catch. The kairomone lure increased the sticky card catches of flower thrips (both *F. occidentalis* and *F. tritici*), while traps baited with $Thripline_{AMS}$ increased flower thrips catches in some habitats. Trap catch differences were most noticeable when thrips numbers were high.

Efficacy of selected biologically derived insecticides (essential oils, spinetoram, spinosad, pyrethrins, and azadirachtin) were evaluated in several different crops grown in southeastern Virginia during 2009 and 2010. Randomized complete block design experiments were conducted in: tomatoes, snap beans, collards, soybeans, cotton and peanuts. Compared with the untreated control, spinetoram and spinosad reduced thrips populations the most effectively. Peanut and cotton treated with spinosad, spinetoram and spinetoram combined with essential oils showed less thrips injury compared with the untreated control, and yield in cotton plots treated with spinetoram was also significantly higher. Population surges did occur on some dates, probably as a result of migrating thrips populations or mass egg hatches. According to these results spinosad and spinetoram appear to be effective biologically derived insecticides, even against the very difficult to control thrips species *F. occidentalis*.