FRANKLINIELLA SCHULTZEI (TRYBOM), AN INVASIVE FLOWER THRIPS ATTACKING VEGETABLE CROPS IN SOUTHEASTERN FLORIDA: IDENTIFICATION, DISTRIBUTION AND BIOLOGICAL CONTROL

By

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To my father and my husband
ACKNOWLEDGMENTS

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

FRANKLINIELLA SCHULTZEI TRYBOM, AN INVASIVE FLOWER THRIPS ATTACKING VEGETABLE CROPS IN SOUTHEASTERN FLORIDA: IDENTIFICATION, DISTRIBUTION AND BIOLOGICAL CONTROL

By
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Major: Entomology and Nematology

Frankliniella schultzei Trybom (Thysanoptera: Thripidae), is an important pest of vegetable and ornamental crops across the globe. In Florida, it is a new pest of vegetable crops. The objective of this study was to determine the abundance of F. schultzei on five vegetable crops including cucumber, pepper, snap beans, squash and tomatoes. Among the five vegetable crops, cucumber was the most preferred host of F. schultzei. The number of larvae exceeded the adults count on cucumber plants indicating that cucumber is the true host of this pest.

In addition, the distribution pattern of F. schultzei within the field and within the host plant was investigated. The distribution study was conducted in field cucumbers and F. schultzei was found feeding on flowers of cucumber plants. Adult counts on leaf samples were significantly lower than flowers in all the experimental plots sampled during three seasons of the study. Both larvae and adults were aggregated in the field during peak populations of the pest. The distribution of F. schultzei varied in response to
the pest density. At high pest density, a clumped pattern was observed whereas regular or random distribution was observed at low pest density.

Results from seasonal abundance study suggested that the population of *F. schultzei* build up in a week after the onset of flowering in a crop. Thrips density was higher during fall 2009 than fall 2008. High temperatures during the fall 2009 may have increased the population growth rate and thus high thrips density during the season. Results from this study helped determine the peak population period during the season, which could be useful to develop sampling protocols for *F. schultzei*. Field trial to determine the efficacy of two predatory mites, *Amblyseius swirskii* (Athias-Henriot) and *A. cucumeris* (Oudemans) suggested that none of the two mite’s species were effective in regulating *F. schultzei* population on cucumbers.
CHAPTER 1
LITERATURE REVIEW

Introduction

Florida harbors a large number of native as well as invasive species of thrips. High temperatures and the humid climate are important factors supporting huge populations of thrips (Aliakbarpour et al. 2010, Kannan et al. 2001) in the state. In the past few years, more than 130 species of thrips from Africa, Europe and the Mediterranean region were intercepted at various ports of entry in the United States (Nickle 2004). The most frequently encountered species were *Frankliniella occidentalis* Pergande, *F. schultzei* (Trybom), *F. intonsa* Trybom and *F. tenuicornis* (Uzel).

The genus *Frankliniella*, including flower thrips, is one of the highly evolved groups of thrips (Waring 2005) inhabiting tropical and temperate areas of the world (Mound 1997). In Florida, *Frankliniella* consists of a huge complex of species (Salguero-Navas et al. 1991, Chellemi et al. 1994, Puche et al. 1995, Eckel et al. 1996), many of which are polyphagous, feeding mainly on the contents of plant cells including fruits, leaves, inflorescence tissues and pollen (Waring 2005) of various vegetables, fruits and ornamental crops (Kendall and Capinera 1987).

In the genus *Frankliniella, F. schultzei*, is a new vegetable pest in south Florida (Frantz and Fasulo 1997). It is a key pest in tomato and cucumber fields in South America (Jones 2005 and Monterio et al. 2001). *Frankliniella schultzei* has a wide distribution range and it is mainly found in tropical and subtropical areas throughout the world (Vierbergen and Mantel 1991). It has been reported from Belgium, mainland Spain, Netherlands, United Kingdom in Europe; Bangladesh, India, Indonesia, Iran, Iraq, Israel, Malaysia, Pakistan and Sri Lanka in Asia; Angola, Botswana, Cape Verde,
Chad, Congo, Egypt, Ethiopia, Gambia, Ghana, Kenya, Libya, Madagascar, Mauritius, Morocco, Namibia, Niger, Somalia, South Africa, Sudan, Uganda, Zimbabwe in Africa; Central and southern Florida (Funderburk et al. 2007) and Hawaii in USA; Barbados, British Virgin Islands, Cuba, Dominican Republic, Haiti, Jamaica and Puerto Rico in the Caribbean; Argentina (Rio de Janeiro), Brazil (Minas Gerais, Parana, Rio Grande do Norte, Santa Catarina, Sao Paulo), Colombia, Chile, Guyana, Paraguay, Peru, Uruguay, Venezuela in South America; New South Wales, Northern Territory, Queensland, South Australia, Victoria, Western Australia, French Polynesia and Papua New Guinea in Australia and the South Pacific (CABI, 1999).

**Polyphagy**

*Frankliniella schultzei* has a wide host range and it is known to feed on various ornamental and vegetable hosts in different parts of the world (Palmer 1990, Vierbergen and Mantel 1991, Milne et al. 1996). *Frankliniella schultzei* along with *F. bispinosa* (Morgan), *F. occidentalis*, and *F. tritici* (Fitch) (Cho et al. 2000, Hansen 2000) are anthophilous species, inhabiting flowers of numerous field crops (Johansen 2002), and are mainly attracted to the color of the host flowers (Menzel & Shmida 1993, Lunau 2000). The majority of flower thrips feeding on floral parts derive nutrition from pollen. A pollen diet rich in protein and other nutrients increases the fecundity of adult thrips and shortens the development period of larval stages (Tsai et al. 1996). Milne (1996) studied the fecundity and development of *F. schultzei* and did not find any significant difference between petal and pollen diets.

The major recorded hosts of *F. schultzei* are cotton (*Gossypium sp*.), groundnut (*Apios Americana*), beans (*Phaseolus vulgaris*) and pigeon pea (*Cajanus cajan (L.*) (Gahukar 2004). However, due to its polyphagous feeding behavior, it also attacks
tomato (*Lycopersicon esculentum*), sweet potato (*Ipomoea batatas*), coffee (*Coffea* sp.), sorghum (*Sorghum* sp.), chillies (*Capsicum annuum*), onion (*Allium cepa*), sunflower (*Helianthus annuus*), rose (*Rosa* sp.), tobacco (*Nicotiana tabacum*), cotton (*Gossypium* sp.), grain legumes (various sp.), lettuce (*Lactuca sativa*), cucumber (*Cucumis sativus*), okra (*Abelmoschus esculentus*), Japanese daisies, irises (*Iris ensata*), spinach (*Spinacia oleracea*), carnation (*Dianthus caryophyllus*), pumpkin (*Cucurbita* sp.), *Carola aubergine* and kidney beans (*Phaseolus vulgaris* subsp. *Nunas*), in different parts of the world (Hill 1975, Monteiro et al. 2001). In Australia, the primary host of this pest is a South American shrub, *Malvaviscus arboreus*, in the absence of which *F. schultzei* infests other non-native plants. The other hosts of this pest, belonging to 12 families includes; *Hibiscus rosasiensis* L. (Malvaceae), *Bauhinia variegata* L. and *B. galpinii* N. E. Brown (Caesalpiniaceae), *Vigna caracalla* L. and *Erythrina crista-galli* L. (Fabaceae), *Ipomoea cairica* (Convolvulaceae), and *Jacaranda mimosifolia* D. Don (Bignoniaceae) (Kirk 1984, 1987; Wilson et al. 1996; Milne and Walter 2000; Coutts et al. 2004; Coutts and Jones 2005).

In addition to the vast herbivore group of thrips, many saprophytic, predatory and parasitic thrips species also exists. Surprisingly, a few pest thrips are also known to be predaceous. The list includes, *F. occidentalis*, *T. tabaci* and *F. schultzei*, all three of which are known to feed on mite eggs (Agrawal et al. 1991 and Milne et al. 1997). Milne et al. (1997) studied the comparative effect of different diets on development of *F. schultzei* and found that diets containing cotton (*Gossypium hirsutum* L.) leaf tissue supplemented with mite eggs, decreased the development period and increased fecundity when compared to simple plant diet. In a choice test between pollen of the
most preferred host and mite eggs under laboratory conditions, an Australian population of *F. schultzei* showed equal preferences to *Malaviscus arboreus* Cav. pollen and mite eggs, suggesting that mite eggs could serve as a desirable food for this pest (Milne et al. 1997).

**Host Preference**

The association between a polyphagous pest like *F. schultzei* and its subsequent host plant is difficult to understand. There are many factors influencing the host preference by thrips. These interactions can be influenced by host cultivar (Fery and Schalk 1991, de Kogel et al. 1998), food availability, host age (Ram and Mathur 1984, Stoddard 1986), plant architecture and flower color. Flower color and structure plays an important role in attracting thrips populations (Menzel & Shmida 1993, Lunau 2000). Mound (2005) suggested that widely open flowers or flowers with high nectar that attracts birds were usually not preferred by thrips. Such selective action is due to thigmotactic behavior whereby thrips like to stay in touch with the surface of their substrate and thus do not prefer widely open flowers. *Frankliniella schultzei* exhibits such behavior and is thus abundant in the rolled petals of its most preferred host *Malaviscus arboreus* in Brisbane, Australia (Milne & Walter 2000). Presence of secondary metabolites is another important character for selecting host plants. However, little research has been done in identifying the chemicals important for host plant and thrips interaction. The secondary metabolites include both kairomones and allomones produced by various plant parts, which may function as attractants or deterrents. The preference of *F. occidentalis* for unopened flower buds to flowers and leaves of chrysanthemum plants due to the presence of (E)-β- farnesene in flower buds suggests an important role of these metabolites in influencing thrips attraction.
(Manjunatha et al. 1998). Flavonoids and carotenoids constitutes another group of such metabolites responsible for attracting thrips by giving colors to plant flowers (Ananthakrishnan and Gopichandaran 1993). For populations of *F. occidentalis* (Mound 2004), *Heliothrips haemorrhoidalis* and Pachaetothripine species, nitrogen content is an important component of host selection in addition to other factors (Fennah 1965). Brodbeck et al. (2001) found increased *F. occidentalis* populations inhabiting tomatoes provided with high nitrogen fertilizer. However, in a study on cucumber and tomato, Leite et al. (2005) evaluated several factors that may influence host preference of *F. schultzei* and determined that the abundance of *F. schultzei* on these plants was not influenced by plant age, leaf chemical composition, levels of leaf nitrogen and potassium and presence of trichomes.

Besides behavioral and nutritional stimuli, many other factors interact in a complex manner to draw thrips population to the host plant (Mound 2004). Given that all potential host plants have an equal chance of being exploited by the population of a polyphagous pest, it has been speculated that thrips show local preferences. *Scirtothrips dorsalis* Hood, a major pest of mango in Puerto Rico, has never been reported as a pest elsewhere on this host. Similarly, *F. schultzei* is a major pest of tomato in Cuba, but in my study in Homestead, Florida, I found it to prefer cucumber over tomato. Considering this, there is a need to conduct an area specific study to assess feeding behavior as a step to protect economically important crops.

**Nomenclature**

*Frankliniella schultzei* has been misidentified or assigned various names by different authors in the past. Mound (1968) reported the presence of a population of adults exhibiting different taxonomy under the taxon *F. schultzei*, which was later
suggested to be a complex of different thrips species (Vierbergen and Mantel 1991). In Australia, the population of *F. schultzei* was earlier known as *F. lycopersici* Steele and the South American population was described as *F. paucispinosa* Moulton (Sakimura 1969). The list of synonyms of *F. schultzei* includes *F. interocellaris* Karny, *F. sulphurea* Schmutz, *F. delicatula* Bagnall and *F. dampfi* Priesner (Mound 1968, Sakimura 1969) and others are listed in the table below.

<table>
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<td>• Cotton bud thrips</td>
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<td><em>Frankliniella lycopersici</em> Andrewartha, 1937</td>
<td>• Tomato thrips</td>
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<td><em>Parafrankliniella nigripes</em> Firault, 1928</td>
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<td><em>Frankliniella paucispinosa</em> Moulton, 1933</td>
<td>• Common blossom thrips</td>
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<td><em>Frankliniella sulphurea</em> Schmutz, 1913</td>
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<td><em>Physopus schultzei</em> Trybom, 1910</td>
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<td><em>Euthrips gossypii</em> Shiraki, 1912</td>
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<td><em>Frankliniella delicatula</em> Bagnall, 1919</td>
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<td><em>Frankliniella trybomi</em> Karny, 1920</td>
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<td><em>Frankliniella tabacicola</em> Karny, 1925</td>
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<td><em>Frankliniella africana</em> Bagnall, 1926</td>
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<td><em>Frankliniella anglicana</em> Bagnall, 1926</td>
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<td><em>Frankliniella aeschyli</em> Girault, 1927</td>
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<td><em>Frankliniella kellyana</em> Kelly &amp; Mayne, 1934</td>
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<td><em>Frankliniella dampfi nana</em> Priesner, 1936</td>
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<td><em>Frankliniella favoniana</em> Priesner, 1938</td>
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<td><em>Frankliniella pembertoni</em> Moulton, 1940</td>
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<td><em>Frankliniella clitoriae</em> Moulton, 1940</td>
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<td><em>Frankliniella schultzei nigra</em> Moulton, 1948</td>
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<td><em>Frankliniella ipomoeae</em> Moulton, 1948</td>
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<td><em>Frankliniella insularis</em> (Franklin) Morison, 1930</td>
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**Color Morphs**

Frankeiella schultzei exists as two different color morphs, a dark and a pale form (Sakimura 1969). The two forms are morphologically similar to each other (Mound 1968) and exhibit a varied distribution across the globe (Sakimura 1969). The dark form is mainly distributed in the south of Sudan to the Cape in Africa, from the Philippines to the south shore of Australia in western Pacific region, from the Caribbean to the south of Argentina in South America, Florida and Colorado in North America, Netherlands in Europe, and throughout India in Asia. The pale form exists in Egypt, Sudan, Uganda and Kenya in Africa; Hawaii in North America, India, and New Guinea in the western Pacific region.

Mixed colonies of both color forms were reported by Mound (1968) in Egypt, India, Kenya, Puerto Rico, Sudan, Uganda, and New Guinea. The two color forms are known to interbreed freely and produce an intermediate form. Frankeiella schultzei is known to reproduce both sexually and parthenogentically by arrhenotoky where males are produced from unfertilized eggs.

**Damage Potential**

Thrips are economic group of insects with broad host range and global expanse. Their small size, high reproductive rate and polyphagous nature allow them to disperse and successfully establish in new geographical regions. Thrips can cause both direct and indirect damage to their host crops where direct damage is due to feeding and oviposition on host plants. Both adults and nymphs of F. schultzei feed on pollen and floral tissue, leading to flower abortion. Serious infestations by this pest may lead to discoloration and stunted growth of the plant (Amin & Palmer 1985) (Figure 1-1). Indirect damage by F. schultzei is due to transmission of plant diseases to various
economically important plants. *Frankliniella schultzei* was reported as the major vector of various plant viruses in north of Australia until the introduction of western flower thrips (Mound 2004). Tomato Spotted Wilt Virus (TS WV), belonging to genus *Tospovirus*, causes serious damage to wide range of plant species (Prins and Goldbach 1998). In Florida, mainly three species in the genus *Frankliniella* are responsible for the transmission of TSWV: *Frankliniella fusca* (Hinds), *F. occidentalis* and *F. schultzei* (Mound 2002, 2004). The dark form of *F. schultzei* is known to vector at least four tospoviruses that includes Tomato Spotted Wilt Virus (TS WV) (Sakimura 1969), Tomato Chlorotic Spot Virus (TCSV), Groundnut Ring Spot Virus (GRSV) and Chrysanthemum Stem Necrosis Virus (CSNV) (Nagata and de Avila 2000). However, the light form of *F. schultzei* reported to be a weak vector of TSWV and TCSV and a non-vector of GRSV (Shakimura 1969, Cho et al. 1988, Mau et al. 1991).

**Dispersion**

*Frankliniella schultzei*, known to originate from South America, is now distributed throughout the world (Mound 2002, Nagata et al. 1999). This small insect could have dispersed either artificially or naturally. The artificial mode of dispersal includes the import of various agricultural products including cut flowers, fruits and vegetables infested with this tiny thrips, air passengers, crew and their baggage, air cargo etc. The natural mode includes the dispersal via flying. Another important mode of dispersal is wind; it affects the flight of these tiny thrips, thereby causing the widespread scattering of this group. Mound (2004) reported that *F. schultzei* is highly vagile and thus has the ability to migrate great distances. Vagile behavior, good dispersal capability and the ability to feed on alternative hosts supports rapid dispersal and increase in population of this group including *F. schultzei* in a new habitat. Thus, the incidence of a large number
of exotic thrips species in Florida emphasizes the need for correct identification to
distinguish them from native thrips in the fauna.

**Biology and Management**

Thysanopterans have always been recorded as opportunistic species well adapted
to dwell in unfavorable conditions. Descendents from detriophagous ancestral group,
their life cycle pattern is much developed to survive in a habitat where optimal condition
for survival is minimal (Funderburk et al. 2001). Thrips life cycle is greatly influenced by
abiotic factors including temperature that affects the development rate until their
threshold level is reached.

Given that thrips are hemimetabolic, there are two inactive and non-feeding stages
in its life cycle: the prepupa and the pupa. The life cycle of *F. schultzei*, includes egg,
first and second instar larvae, prepupa, pupa and adult. Gravid females lay eggs inside
the plant tissue, which hatch in 2-5 days depending upon environmental conditions.
*Frankliniella schultzei* takes around 12.6 days to complete its life cycle at 24.5°C (Silvia
et al. 1998) The embryonic stage lasts for four days, and the first instar, second instar,
prepupa and pupa takes an average of 2.5, 2.5, 1.2, and 2.1 days, respectively. Adult
female and male longevity is approximately 13 days (Silvia et al. 1998).

**Sampling**

Early detection and identification is a primary step towards developing an efficient
management practice of any invasive pest. Monitoring provides information about
composition of pest species, pest and crop status, weather and soil factors affecting
crops. From an IPM point of view, monitoring is done to evaluate the presence of the
pest in the field, to determine its population density and field distribution in order to
apply the most appropriate management practice. Monitoring may involve different
sampling methods using sticky cards, \textit{in situ} counts, traps, knockdown sampling or netting. In the past, different sampling strategies have been developed by entomologists to determine the presence and status of thrips species. To study flight behavior of \textit{S. dorsalis}, Takagi (1978) constructed a sticky suction trap, which could also monitor abundance of various other pests. Use of colored sticky traps is one of the most commonly used sampling strategies for thrips. It has been found that white sticky traps was the best trap to use for monitoring \textit{F. occidentalis} in avocados (Hoddle et al. 2002), \textit{S. dorsalis} on pepper (Saxons et al. 1996), \textit{F. bispinosa} and \textit{F. tritici} in blueberries (Finn, 2003, Liburd et al. 2009, Saona et al. 2010) and citrus (Childers and Brecht, 1996). Tsuchiya et al. (1995) performed a color preference test for \textit{S. dorsalis} and found that the pest was attracted to yellowish-green, green, and or yellow boards. Similarly, \textit{F. tritici} also exhibits preference among different colored sticky traps. In a study, \textit{F. tritici} was found to be most attracted to yellow in comparison to blue or white traps when sampled in tomato crop (Cho et al. 1995). All these studies suggest a possible role of host flower color, which may influence the attraction of thrips to a particular color of sticky trap. \textit{Frankliniella schultzei}, an anthophilous thrips species, is frequently found feeding on flowers of its host plants. Like other thrips, \textit{F. schultzei} can also be sampled using colored sticky traps. Yaku et al. (2007) studied the color preference of male and female \textit{F. schultzei} separately. The two sexes were found to exhibit varied preference for the different colored sticky traps, where male thrips were captured more on yellow sticky traps and female thrips were captured more on pink sticky traps.

\textbf{Chemical and Biological Control}

The most commonly used tool to control thrips in a field is chemical insecticides. However, the use of these insecticides is limited by the “three R’s”: 1. Resistance-
selection of resistant strain in response to non-judicious use of insecticides, 2. Resurgence of the pest and 3. Replacement of a primary pest by the secondary pest in a treated area. Thus, in order to overcome these adverse effects of insecticide use, there has been much emphasis on developing cultural and biological control strategies.

Biocontrol agents known for controlling thrips include minute pirate bugs, big eyed bugs (Hemiptera); green lacewing larvae (Neuroptera); phytoseiid mites (Parasitiformes) and predatory thrips (Thysanoptera); (Miles et al. 1997). However, among various predators, the most commonly used predators for thrips control belong to genus Orius (Hemiptera: Anthocoridae) and Amblyseius (Acari: Phytoseiidae). In the genus Orius, O. insidiosus Say is known to feed on a wide range of thrips making it one of the most promising biocontrol agents for thrips control in Florida (Funderburk et al. 2000). Silveira et al. (2005) studied the association between F. schultzei and O. insidiosus by sampling various crops and weeds in field and greenhouse conditions in Brazil. Orius insidiosus was found to be associated with F. schultzei population and it was suggested as a predator of this pest.

Phytoseiid mites constitute another important group of predators. Variable reports have been documented regarding the use of predatory mites as biocontrol agents of thrips. Gillespie (1989) found A. cucumeris (Oudemans) as an effective predator of Thrips tabaci Lindeman, on greenhouse cucumber in Europe. Later, Van de Veire and Degheele (1995), and Jacobson (1997) evaluated the efficacy of A. cucumeris against flower thrips (Frankliniella sp.) in greenhouse conditions and reported these mites to be an efficient biocontrol agent. In a greenhouse study on tomato, A. cucumeris was found to give a better suppression than O. insidiosus on F. occidentalis (Shipp and Wang
2003). Despite numerous success reports, *A. cucumeris* did not perform well in some studies and such variability was attributed to its response to change in humidity and temperature (Shipp and Van Houten 1997). Another phytoseiid mite widely known for its generalist behavior is *Amblyseius swirskii* (Athias-Henriot). *Amblyseius swirskii* has shown promising results in regulating chilli thrips on pepper (Arthurs et al. 2009) and broad mites and whitefly on ‘Serrano’ pepper in field studies (Stansly and Castillo, 2009) in Florida. *Amblyseius swirskii* was compared with *A. cucumeris* in regulating broad mites and was found to be more effective than *A. cucumeris* in suppressing broad mites (Stansly and Castillo, 2009). Similar results were obtained by Arthurs et al. (2009) who reported *A. swirskii* as a better predator of chilli thrips on pepper than *A. cucumeris*. However, in the majority of the studies done in past, the use of these mites are restricted to greenhouse or controlled conditions. Thus, evaluating and comparing the effect of these mites under field conditions will be an interesting study.

**Research Goals**

Considering the damage potential of *F. schultzei* to various vegetable crops in Miami-Dade County and other adjoining regions largely under agriculture, it is important to develop a management program for this pest. Biology, identification and monitoring techniques of a pest are one of the few important areas to focus before developing control programs. Thus, one of the goals of my study is to learn how to correctly identify *F. schultzei*, which is difficult because of its small size and ambiguous common features amongst various thrips species that co-exists in the environment. In addition, I evaluated the role of phytoseiid mites in regulating *F. schultzei* population in the field and determined seasonal abundance and distribution pattern of *F. schultzei*. 
Specific Objectives

1) Investigate the abundance of *F. schultzei*, and identify associated thrips species on various vegetable crops in south Florida.

2) Examine the distribution and seasonal abundance of *F. schultzei* in south Florida cucumber fields.

3) To evaluate *A. cucumeris* and *A. swirskii* as potential biocontrol agents for *F. schultzei* in field cucumbers.

![Cucumber flower showing discoloration due to feeding by adult Frankliniella schultzei](image)
CHAPTER 2
ABUNDANCE OF *FRANKLINIELLA SCHULTZEI* AND IDENTIFICATION OF ASSOCIATED THRIPS SPECIES ON VARIOUS VEGETABLE CROPS IN SOUTH FLORIDA

Thrips are ubiquitous due to their ability to exist in a wide range of habitats. Tropical and temperate regions are the most suitable regions for thrips survival (Mound 1997), making Florida a vulnerable region for thrips invasion and subsequent establishment. Furthermore, the diverse flora of Florida offers free choice to this opportunistic group of insects. While the majority of economically important thrips species are polyphagous in nature, there exists a range of preferred hosts for this group of insects. Several thrips species including *Frankliniella occidentalis* (Pergande) have been reported to exhibit varied host preference in the same geographical region (Doederlein and Sites 1993). However, very little information is presently available on the host switching behavior of this group.

In the past, various researchers have often been confused in identifying host plants of a polyphagous thrips species leading to incorrect documentation of hosts of thrips. Plant species have been merely designated as a host based on the presence or absence of adults of the thrips species under study (Mound 2005). There is a very thin line between primary and a secondary host of thrips, thus defining proper host plants is very important. While thrips are known to forage on wide range of plant species, a primary host is known by its ability to support thrips reproduction in addition to providing food and shelter. However, a provisional or secondary host is usually sought for food and shelter, and does not provide a substrate for reproduction (Mound 2005). Once a true host is identified, it is important to determine the preferred hosts among broad list of host species.
Host preference by a species dwelling in a geographical region can be affected by several factors. Doederlein and Sites (1993) found that *F. occidentalis* did not possess any preferred host among the 11 plant species sampled during the season as the abundance of *F. occidentalis* on different plant species varied in a two-year study. They suggested that environmental factors and presence of flowers on hosts can be important in explaining such plasticity in feeding preferences. Other factors influencing host selection include drought, flooding and various other stressing factors. Lewis (1973) suggested that stressed plants are vulnerable to thrips attack, as the stress restrains plants from protein synthesis resulting in increased nitrogenous compounds.

*Frankliniella schultzei* (Trybom) is a polyphagous herbivore known to feed on wide range of plant species. However, it is not an exclusively phytophagous pest, as it has been reported to feed on eggs of twospotted mites *Tetranychus urticae* Koch on cotton (Trichilo and Leigh 1986, Wilson et al. 1996). *Frankliniella schultzei* is one of the major pests of various ornamental and vegetable crops around the globe. It has been cited as a pest of cotton, groundnut and beans in many parts of the world. In Cuba and Brazil, it is one of the key pests of tomato and hence called tomato thrips (Haji et al. 1998 and Jones 2005). However, in Florida it has been found to be associated more with flowers of ornamental plants (Funderburk et al. 2007), and cucumbers in southeastern Florida (Personal observation).

Considering the paucity of information on host plants under risk in Miami-Dade County, largely known for fresh vegetable production, this study was conducted with an objective to determine abundance of *F. schultzei* on five major vegetable crops in this new geographical area of infestation. The results from the study helped in determining
crops susceptible to attack by *F. schultzei* in this region. This information will be useful to growers and scouting personnel in developing monitoring programs and applying preventive measures in time.

**Materials and Methods**

Abundance of *F. schultzei* was studied on Cucumber (*var. Vlaspek*), Pepper (*var. King Arthur*), snap beans (*var. Opus*), squash (*var. Straightneck*) and tomato (*var. Flora-Dade*). These are commonly grown vegetable crops in south Florida. The study was conducted in a field at the University of Florida, Tropical Research and Education Center (TREC), Homestead during fall 2009. Five vegetable hosts selected for this study were planted in plots laid adjacent to each other in a same field. A 9-m wide non-planted buffer area separated two adjacent crops. The soil type of the field was Krome gravelly loam (loamy-skeletal, carbonatic hyperthermic lithic Udorthents), which consists of about 33% soil and 67% limestone pebbles (>2mm). Fields were prepared using standard commercial practices (Olson and Santos 2010).

**Crop Management**

**Cucumber** (*Cucumis sativus* L.): `Vlaspek` cucumber was directly seeded on Aug, 17 on a flat ground. Seeds were sown 15.2 cm apart within the row and 91.4 cm apart between rows. The plot measured 251 m² with 30 m long ten rows of plants. At planting, 8-16-16 (N-P-K) was applied at 908 Kg/ha in a furrow (20 cm apart from the seed row). Halosulfuron methyl (Sandea®, Gowan Company LLC., Yuma, Arizona) at 55 ml/ha was used as a pre emergence herbicide to control weeds. Copper hydroxide (Kocide® 3000, BASF Ag Products, Research Triangle Park, NC) at 0.8 l/ha and Chlorothalonil (Bravo®, Syngenta Crop Protection, Inc., Greensboro, NC) at 1.75 l/ha were used in rotation at two-week intervals to prevent fungal diseases. The crops were irrigated twice a week.
with 3 cm of water using overhead sprinklers in fields. Fertilizer 4-0-8 (N-P-K) at 236 l/ha/wk was used once a week as an in-furrow band in the field to provide 2.4 kg-$N_2$/ha/wk and its use was initiated three weeks after planting. *Bacillus thuringiensis* based insecticides, Dipel DF® (var. kurstaki) at 1.1 Kg/ha and Xentari DF® (var. kustaki) at 1.2 l/ha (Valent Biosciences Corporation, Libertyville, IL) were used to control melon and pickle worms in the experimental field.

**Pepper** (*Capsicum annuum*): `King Arthur` pepper transplants were planted 25 cm apart within rows on raised beds in the field on Aug, 17. These raised beds were 91 cm wide, 15 cm high, and 182 cm apart between centers, covered with 1.5 ml thick black polyethylene mulch. Each plot consisted of 10 raised beds 30 m long making a plot of ~500 m². Management of crop including use of fertilizers, herbicides and fungicide is same as described for cucumber. *Bacillus thuringiensis* based insecticides, Dipel DF® (var. kurstaki) at 1.1 Kg/ha and Xentari DF® (var. kustaki) at 1.2 l/ha (Valent Biosciences Corporation, Libertyville, IL) were used to control beet army worm, *Spodoptera exigua* (Hubner) in the experimental field. Thiamethoxam (Actara® 25WG, Syngenta Crop Protection, Inc., Greensboro, NC) was added at the rate of 220 ml/ha twice during the cropping to control pepper weevil, *Anthonomus eugenii* Cono and whitefly, *Bemisia argentifolii*.

**Snap beans** (*Phaseolus vulgaris*): `Opus` snap beans were directly seeded on a flat ground, placed 7.5 cm apart within the row and 91.4 cm between rows on Aug, 18. The plot measured 251 m² with ten rows 30 m long in the plot. The field was prepared using standard cultural practices as described for cucumber. Crops were irrigated twice a week with 3 cm of water using overhead sprinklers in fields. Fertilizer 4-0-8 (N-P-K) at 236 l/ha/wk was used once a week as an in-furrow band in the field to provide 2.4 kg-
N\textsubscript{2}/ha/wk and its use was initiated three weeks after planting. Imidacloprid (Admire\textsuperscript{®} Pro, Bayer CropScience, NC) was applied to the soil at the rate of 591 ml/ha once when plants were two weeks old plants to control whitefly.

**Squash** (*Cucurbita maxima*): `Straightneck` squash were directly seeded on a flat ground in a plot measuring 251 m\textsuperscript{2} on Sep, 2. Each plot consisted of 30 m long ten rows of squash plants. Seeds were sown 21 cm apart within the row and 91.4 cm apart between rows. Crop management practices were similar to cucumber. Squash was planted two weeks after the other four crops, to avoid differences in the onset of flowering. This was to help ensure that all the plant species had equal chances of being infested by *F. schultzei*.

**Tomato** (*Solanum Lycopersicon*): `Flora-Dade` tomato seedlings were transplanted 30 cm apart within rows on raised beds on Aug, 18. The raised beds were 91 cm wide, 15 cm high, and 182 cm apart between centers, covered with 1.5 ml thick black polyethylene mulch. Each plot consisted of 10 raised beds 30 m long, making a plot of 501 m\textsuperscript{2}. The field was prepared in accordance with standard cultural practices as described above. The crop was drip irrigated twice a week. Fertilizer 4-0-8 (N-P-K) was used at 236 l/ha once a week beginning three weeks after planting through the drip to provide 2.4 kg-N\textsubscript{2}/ha. *Bacillus thuringiensis* based insecticides, Dipel DF\textsuperscript{®} (var. kurstaki) at 1.1 Kg/ha and Xentari DF\textsuperscript{®} (var. kustaki) at 1.2 l/ha (Valent Biosciences Corporation, Libertyville, IL) were used to control beet armyworm in the experimental field. Imidacloprid (Admire\textsuperscript{®} Pro, Bayer CropScience, NC) was applied as a soil drench at the rate of 591 ml/ha once to three week old plants to control whitefly.
Sampling

Samples were collected and processed independently for each of the plant hosts. In each plot belonging to a host plant type, five flowers (a flower/plant) were randomly collected from every row of the plot. All flower samples belonging to each row of the various hosts were placed in separate ziplock® bags (17 X 22 cm) marked with the date of collection, row number and host type. Samples were transported to the Univ. of Florida, vegetable IPM laboratory, TREC, Homestead where samples were placed in a one-quart plastic cup with 75% ethanol for 30 minutes to dislodge various life stages of thrips. The samples were carefully taken out of the cup leaving the thrips in alcohol. The contents in alcohol were sieved using a 25-µm grating, USA Standard Testing Sieve (W. S. Tyler, Inc.) as per Seal and Baranowski (1992). The residue in the sieve was washed off with 75% alcohol in to a Petri dish and checked under a dissecting microscope at 12X to record various species of thrips. *Frankliniella schultzei* and adult thrips not identified as *F. schultzei* were separated and stored in 75% ethanol for further identification (discussed in chapter 3). Samples were taken during fifth, sixth and seventh week after planting.

Statistical Analysis

Data were analyzed independently for larvae and adults. Data on the abundance of larvae and adults on each crop were averaged for all samplings. The mean number of larvae and adults per crop was compared using one way analysis of variance (ANOVA) (PROC GLM, SAS Institute Inc. 2003). Data were transformed by \( \log_{10}(x+1) \) to comply with model assumptions before analysis. Untransformed means and standard
errors are reported in figures. Differences among means of larvae and adult on various crops were separated using Tukey’s HSD (Honestly Significant Difference) procedure ($P < 0.05$).

**Results**

**Frankliniella schultzei abundance on five host plants:** Four of the five potential hosts were found to be infested with *F. schultzei* adults. Infestation of adults on tomato flowers was not significantly different to squash and cucumber flowers (Tukey’s HSD test, $P < 0.005$) (Figure 2-1). The least number of adults were captured on beans and none was found on pepper flowers. The number of adults on tomato and squash was significantly greater than in bean flowers ($F = 6.56; \text{df} = 4, 45; P < 0.001$).

Mean number of *F. schultzei* larvae was highest on cucumber flowers (Figure 2-2). The infestation level on cucumber flowers was significantly higher than the other four hosts ($F = 32.52; \text{df} = 4, 45; P < 0.001$) (Figure 2-2). Not a single larva was found on pepper (Figure 2-2). There was no significant difference in the number of larvae sampled from squash, tomato and beans (Tukey’s HSD test, $\alpha = 0.05$).

In the course of sampling five vegetable crops, other thrips species were also encountered. The predominant species besides *F. schultzei* was *Thrips palmi* Karny, followed by *Frankliniella occidentalis* (Pergande) and *F. fusca* (Hinds) (Figure 2-3). While, *T. palmi* was the second most abundant species encountered on various vegetable crops (excluding tomato) after *F. schultzei*, the number of *T. palmi* encountered in total was low when compared with *F. schultzei* counts in flowers on various crops. The highest number of *T. palmi* was found in beans flowers with an average of six adults per five flowers, followed by squash, and cucumber. The least number of *T. palmi* was collected from pepper flowers (Figure 2-3).
*Frankliniella bispinosa* (Morgan) was found on bean flowers but the number was fewer than four specimens in the total number of thrips adults collected from various crops during the study. *Frankliniella occidentalis* was collected from squash, tomato, cucumber and bean flower, with the number of adults ranging between 0.2-1.0 per five flowers sampled from various hosts in the study. *Frankliniella fusca* was collected from tomato flowers with an average number of 0.6 adults per five flowers (Figure 2-3).

**Discussion**

Results of this study suggested that the five vegetable crops were infested with at least four different thrips species. These were *F. schultzei, F. fusca, F. occidentalis* and *T. palmi*. The number of thrips species other than *F. schultzei* encountered on flowers of sampled crops was small with insignificant damage potential. Thus, the pest of major concern was *F. schultzei*.

*Frankliniella schultzei* is reported as a pest of several ornamental and vegetable crops in the scientific literature. In my study, adult *F. schultzei* densities on various plant species sampled was consistent for the top three preferred host, which includes tomato, squash and cucumber. High density of *F. schultzei* on tomato flowers was in agreement with Jiminez et al. (2006), Monteiro et al. (2001) and Sakurai (2004), who reported tomato to be a major host of *F. schultzei* in Cuba, Brazil and Paraguay, respectively. However, when plant species in the present study were ranked according to host status based on larval density, a different pattern was observed. Only cucumber was identified as a suitable host of *F. schultzei*, with the number of larvae greater than the adults, suggesting cucumber to be a breeding site for this pest. The larval counts were lower than adult counts on other hosts and thus these crops including tomato can be regarded as lower-ranked hosts of this pest in south Florida.
Variation in host plant preferences of pests inhabiting different geographical regions has also been reported for several other insect species in the literature. Probable reasons for such plasticity in behavior are suggested to be genetic or environmental variations (Jaenike 1990), although there is little published evidence for the role of genetic variation in host preference. Environmentally induced variation is known to cause differences in host preferences for a species in different regions. Jaenike (1990) explained that the abundance of the most preferred host in a region can result in higher thresholds for low ranked host plant, which may be disregarded by the pest in that area. Absence of this preferred host in another geographic region changes the threshold and thus the preference level for low ranked host changes for a species. However, preference for cucumber over tomato by *F. schultzei* in my study, where the pest was given free choice is still ambiguous. Nevertheless, a more comprehensive picture about the true host range for *F. schultzei* in this region has emerged. Further studies on the seasonal abundance on these hosts will broaden our knowledge on the dispersal and interaction with various hosts on which *F. schultzei* prospers.
Figure 2-1. Number of *F. schultzei* adults (Mean ± SEM) on flowers of five host plants sampled during fall 2009. Means with the same letter are not significantly different (P > 0.05, Tukey’s HSD test).
Figure 2-2. Number of *F. schultzei* larvae (Mean ± SEM) on flowers of five host plants sampled during fall 2009. Means with the same letter are not significantly different (P > 0.05, Tukey’s HSD test)
Figure 2-3. Average number of adults of three thrips species sampled from five vegetable crops.
The beginning step of any management practice is correct identification of the insect pest. In the past several integrated pest management strategies have been developed against various thrips species. The success and sustainability of these approaches are dependent on several factors including correct identification of the target species because management practices vary greatly with different thrips species belonging to a single genus. In Florida, the genus *Frankliniella* consists of a huge complex of species (Salguero-Navas et al. 1991). The presence of several other thrips species makes thrips identification difficult for non-specialists including growers.

The small size and presence of several dark colored thrips including *F. fusca* (Hinds), *F. schultzei* Trybom, and *F. insularis* (Franklin) makes color based field identification difficult. Thus, identification of thrips is mainly based on characters like antennal segments, body setae, presence or absence of a comb on the VIII abdominal segments in addition to color. By using traditional taxonomic keys, thrips can be assigned to a particular genus, but due to high intraspecific morphological variation (color morphs) of many conspecifics of *Frankliniella*, expertise is required for identification to species level. The objective of this study is to present important identification features of six thrips species inhabiting five vegetable crops, cucumber, squash, pepper, snap beans and tomato in south Florida. The images of morphological features of thrips species will help scouting personnel and researchers in the identification of major thrips species encountered.
Materials and Methods

Sampling

In the present study, adult thrips were collected during the course of sampling various crops (mentioned above) to study the abundance of *F. schultzei*. Thrips were processed for identification to the species level. In addition to the selected crops, 10 flowers were collected from a weed, *Bidens alba* (L.) DC var. Radiate, growing adjacent to experimental plots at the TREC field. These flowers were infested with dark thrips and samples were collected to determine if this weed served as a reservoir for *F. schultzei* in the field.

Determination of Adult Thrips

**Slide mounting:** Twenty thrips of each distinct morph and five specimen of *F. fusca*, collected from the various host crops were used for the identification. Thrips collected in vials containing 75% alcohol and then transferred to a 10% KOH (Potassium hydroxide solution) solution prepared in 50% ethanol to lighten the dark color of cuticle on various body parts. Duration of keeping specimen in the solution was standardized to 15-25 min depending on the darkness of thrips cuticle. While still in KOH, the insect was then gently punctured in the abdomen (close to the thorax to avoid disrupting features near the ovipositor) using a fine insect pin to facilitate the removal of its abdominal contents. Specimen were then passed through a series of alcohol concentrations starting from 65%, followed by 75%, 85%, 90% and 95% ethanol to initiate gradual dehydration of the specimen. Thrips were placed in each of the above mentioned alcohol concentrations for 5-8 minutes to avoid any moisture interaction at the final stage of slide mounting. Each specimen was placed ventrally on a slide with a small drop of Hoyer’s mounting media and covered with a glass cover slip. The adult
female thrips were identified using thrips identification key (Nakahara 1994) and pictures were taken using Insight Firewire Spotimaging (4 Megapixel) camera (Vegetable IPM Lab, TREC, University of Florida).

**Insect Identification**

The identification of thrips collected from various crops and weeds surrounding (*B. alba*) the experimental plot in Homestead suggested that the plants were infested with at least six different thrips species. They were *F. schultzei*, *F. insularis*, *F. fusca*, *F. occidentalis*, *T. palmi*, *Thrips florum* Schmutz and *Microcepalothrips abdominalis* (Crawford). Following are the morphological features that helped in the identification of these thrips.

**Frankliniella schultzei** (Trybom)

The common blossom thrips, *F. schultzei* (Thysanoptera: Thripidae) exists in two different color morphs (Figure 3-1 and 3-2). The dark form of *F. schultzei* is dark brown in color (Frantz and Fasulo 1997) (Figure 3-1). There are eight antennal segments with pale bases of segments 3-5. The eighth segment is slightly longer than seventh segment (Figure 3-3). The interocellar setae or the third pair of ocellar setae arises between the anterior ends of the two hind ocelli (Figure 3-4). The postocular setae (1) are shorter than interocellar setae (2) on the head (Figure 3-5). The pronotum of species from genus *Frankliniella* have five pairs of developed setae. In *F. schultzei* the anteromarginal setae (1) are slightly shorter than anteroangular setae (2) (Figure 3-6). The metanotum lacks campaniform sensilla (Figure 3-7). Forewing bears two complete rows of veinal setae (Figure 3-8). Posteromarginal comb on the eight abdominal segment is not fully developed and is incomplete medially bearing short microsetae on either ends (Figure 3-9).
**Frankliniella insularis** (Franklin)

*Frankliniella insularis* (Thysanoptera: Thripidae) is a pest of South America origin. Its geographical range extends from Central America to Argentina and southern states of the US. In my study, I sampled this species from *B. alba* flowers. Females are dark brown in color (Figure 3-10). There are eight antennal segments with pale bases of third, fourth and fifth segments (Figure 3-11). The segment three and four bears a forked sensorium. The head bears three pairs of ocellar setae and third pair of ocellar setae arises from the sides of the ocellar triangle and a long pair of fourth postocular setae (Figure 3-12). The pronotum have five pairs of developed setae and the anteromarginal setae are shorter than anteroangular setae (Figure 3-13). Metanotum bears two pairs of long setae on anteromarginal end and two campaniform sensilla (Figure 3-15). Forewing has a distinct pale base bearing two complete rows of veinal setae (Figure 3-16). Posteromarginal comb on eight abdominal segment is fully developed, arising from triangular bases (Figure 3-17).

**Frankliniella fusca** (Hinds)

The tobacco thrips, *F. fusca* (Thysanoptera: Thripidae) has two different wing morphs. Morphs with wings are known as micropterous and wingless forms are known as brachypterous (Figure 3-18). Adults of *F. fusca* are brown in color with eight antennal segments (Figure 3-19). Head bears three pairs of ocellar setae and the third pair originates above the two hind ocelli and out of the ocellar triangle. The postocular setae on head are small (Figure 3-20). The pronotum have five pairs of developed setae and in *F. fusca*; anteromarginal setae are distinctively shorter than anteroangular setae (Figure 3-20). The metanotum has the campaniform sensilla (Figure 3-21) and forewing
has two complete rows of veinal setae (Figure 3-22). Posteromarginal comb on eight abdominal segment is absent in this species (Figure 3-23).

**Frankliniella occidentalis** (Pergande)

The western flower thrips, *F. occidentalis* (Thysanoptera: Thripidae) is a pest of US origin. It exists in dark, pale and an intermediate color forms. The adult body is yellow in color with brown bands on tergite (Figure 3-24). There are eight antennal segments, where the yellow colored third, fourth and fifth segments have brown apices (Figure 3-25). Head possesses three pairs of ocellar setae and the third pair arises from the anterior margin of the two hind ocelli (Figure 3-26). The ocellar setae and postocular setae on head are equal in length (Figure 3-26). The pronotum have five pairs of developed setae and in *F. occidentalis*; anteromarginal setae are slightly shorter than anteroangular setae. The metanotum bears campaniform sensilla (Figure 3-27). Forewing consists of two complete rows of veinal setae (Figure 3-28). Posteromarginal comb on eight abdominal segment is fully developed bearing a row of microtrichia.

**Microcephalothrips abdominalis** (Crawford)

The composite thrips, *M. abdominalis* (Thysanoptera: Thripidae) is the only species of genus *Microcephalothrips*. Childers et al. (1999), Childers and Nakahara (2006) reported the infestation of this species on citrus trees and citrus groves in southern Florida. In my study, I sampled this species from *B. alba* flowers. The adult body is brown in color (Figure 3-29) bearing seven segmented antennae (Figure 3-30). Head possesses only two pairs of ocellar setae unlike various species of *Frankliniella* as described above (Figure 3-31). The third pair arises from the anterior end of ocellar triangle. Anteromarginal setae on pronotum are absent. The pronotum bears two pairs of small posteroangular setae and five pairs of posteromarginal setae. A pair of setae is
centrally located in mesonotum (Figure 3-32) and metanotum (Figure 3-33) separating this species from other thrips collected in the study. The metanotum bears campaniform sensilla (Figure 3-33). Forewing has incomplete rows of setae, with two and three setae on the distal end of the rows, respectively (Figure 3-35). Posteromarginal comb on eight abdominal segment is fully developed bearing a complete row of microtrichia (Figure 3-34).

*Thrips palmi* Karny

The melon thrips *Thrips palmi* Karny (Thysanoptera: Thripidae), is an economically important pest of various greenhouse and field crops in south Florida. It is a pest of Southeast Asia origin from where it spread to the rest of Asia, North Africa, Australia, Central and South America, and the Caribbean. In Florida, it was first observed in 1990. The adult body is yellow in color (Figure 3-36). The antenna is seven segmented with darker terminal segments (Figure 3-37). Head bears two pairs of ocellar setae and the interocellar setae arises from a region closer to the posterior end of the apical ocelli. The interocellar setae are longer than the postocular setae (Figure 3-38). The pronotum of *T. palmi* has two pairs of posteroangular setae and it lacks both the pairs of anteroangular and anteromarginal setae. The anterior end of metanotum possesses distinct transverse lines and toward the posterior end are present a pair of campaniform sensilla (Figure 3-39). The first vein of forewing has three setae on distal end (Figure 3-40). Posteromarginal comb on eight abdominal segment is fully developed bearing a row of long microtrichia (Figure 3-41).
Figure 3-1. Dorsal view of an adult *F. schultzei*, with dimensions marked

Figure 3-2. Slide mount of an adult of light form of *F. schultzei*
Figure 3-3. Antenna of adult *F. schultzei* showing 8 antennal segments with pale bases of 3-5\textsuperscript{th} segment

Figure 3-4. Head of an adult *F. schultzei* showing interocellar setae at 40 X magnification
Figure 3-5. Head of an adult *F. schultzei* showing postocular setae (1) smaller than interocellar setae (2) at 40 X magnification.

Figure 3-6. Prothorax of an adult *F. schultzei* showing the anteromarginal setae (1) slightly shorter than anteroangular setae (2) on the anterior of the prothorax.
Figure 3-7. Metanotum of an adult *F. schultzei* lacks campaniform sensilla

Figure 3-8. Forewing of an adult *F. schultzei* showing two complete rows of veinal setae
Figure 3-9. Abdomen of an adult *F. schultzei* showing a weakly developed comb on the eighth abdominal segment

Figure 3-10. Slide mount of an adult *F. insularis* showing dorsal view
Figure 3-11. Antenna of adult *F. insularis* showing 8 antennal segments with pale 3-5th

Figure 3-12. Head of an adult *F. insularis* showing 3rd pair of interocellar setae and 4th pair of postocular setae
Figure 3-13. Pronotum of an adult *F. insularis* showing five pairs of pronotal setae, anteromarginal setae are shorter than

Figure 3-14. Mesonotum of an adult *F. insularis* bearing longitudinal lines
Figure 3-15. Metanotum of an adult *F. insularis* bearing two campaniform sensilla

Figure 3-16. Forewing of an adult *F. insularis* with a pale base, bearing two complete rows of setae
Figure 3-17. Abdomen of an adult *F. insularis* showing comb on the eighth abdominal segment

Figure 3-18. Slide mount showing an adult of *F. fusca*
Figure 3-19. Antenna of adult *F. fusca* showing 8 antennal segments with pale 3rd and 4th.

Figure 3-20. Head of an adult *F. fusca* with 3 pairs of ocellar setae and pronotum bearing anteroangular and anteromarginal setae.
Figure 3-21. Metanotum of an adult *F. fusca* bears campaniform sensilla.

Figure 3-22. Forewing of an adult *F. fusca* bears two complete rows of setae.
Figure 3-23. Abdomen of an adult *F. fusca* lacks comb on the eight abdominal segment

Figure 3-24. Slide mount of a pale form of an adult of *F. occidentalis*
Figure 3-25. Antenna of adult *F. occidentalis* showing 8 antennal segments with dark apices of 3-5\textsuperscript{th} segment

Figure 3-26. Head of an adult *F. occidentalis* showing pair of ocellar and postocular setae
Figure 3-27. Metanotum of an adult *F. occidentalis* bears two campaniform sensilla

Figure 3-28. Forewing of an adult *F. occidentalis* showing two complete rows of setae
Figure 3-29. Slide mount of an adult *M. abdominalis*

Figure 3-30. Antennae of an adult *M. abdominalis*
Figure 3-31. Head of an adult *M. abdominalis* showing third pair of ocellar setae

Figure 3-32. Mesonotum of an adult *M. abdominalis* with a pair of setae
Figure 3-33. Metanotum of an adult *M. abdominalis* with campaniform sensilla

Figure 3-34. Adult of *M. abdominalis* with a complete comb on the eight abdominal segment
Figure 3-35. Forewing of an adult *M. abdominalis* with incomplete rows of setae

Figure 3-36. Slidemount of an adult *T. palmi*
Figure 3-37. Seven segmented antennae of an adult *T. palmi*

Figure 3-38. Head of an adult *T. palmi* with two pairs of ocellar setae
Figure 3-39. Metanotum of an adult *T. palmi* bears a pair of campaniform sensilla

Figure 3-40. Forewing of an adult *T. palmi*
Figure 3-41. Abdomen of *T. palmi* showing complete comb on the eight abdominal segment
CHAPTER 4
DISTRIBUTION AND SEASONAL ABUNDANCE OF FRANKLINIELLA SCHULTZEI

Thrips are economic group of insects posing serious threats to various commercially important crops. Owing to their polyphagous behavior, they attack a wide range of hosts in variable environments and geographical locations. Thrips exhibit diverse population dynamics, which are largely affected by overlapping host ranges of different thrips species (Cho et al. 2000, Ramachandran et al. 2001 and Reitz et al. 2003). Reitz et al. (2003) reported that Frankliniella occidentalis (Pergande) is very common in Florida during winter, but later at the beginning of spring, it is displaced by competitive Frankliniella tritici (Fitch) and Frankliniella bispinosa (Morgan) populations. Similarly, Thrips palmi Karny population on `Pod squad` beans was found to be denser in fall and spring seasons, compared to summer due to their low reproductive success at high summer temperatures (Seal 1997). In Florida, T. palmi infestations begin early in the crop-growing season during October and continue until June (Seal 1997). Such information on seasonal population dynamics of various insects helps scouting personnel to locate pest infestation early in the season. Unfortunately, information on seasonal abundance of F. schultzei in Florida is lacking. Thus, we studied the population dynamics of this pest in fall-2008 and 2009, which is a growing season for cucumber in Homestead.

Thrips distribution in any field is highly affected by its ecology. Due to its ability to exploit vast range of host plants, thrips take refuge in weeds (uncultivated area) surrounding the cultivated crops until the availability of preferred host. Once crop is ready, these thrips are attracted to food offered by the cultivated crops (preferred host) and make local dispersions to the cultivated areas. Seal and Stansly (2000) found that
the distribution of *T. palmi* was aggregated in field grown beans. Similar distribution pattern was observed for *Scirtothrips dorsalis* infesting pepper plants in St. Vincent (Seal et al. 2006). Seal et al. (2001) also found that infestation by *T. palmi* began on the edges of the field, slowly progressing toward the interior.

In general, insect populations can be random, clumped or uniformly distributed. The distribution of insects is often influenced by its density in a field. Lower number of insects in a field leading to low capture rate during sampling suggests a random distribution of insects in the field (Southwood 1978). Similarly, denser the population of the pest, the more aggregated is the pest in a field. Majority of the insect group possesses such population distribution trends in the field. However, regardless of the clumped, regular or random distribution pattern of insects, traditionally, insecticides are applied uniformly to fields (Weisz et al. 1996) aggravating ecological, economical and environmental damage. Consequently, such disturbances to the natural system inaugurate the need of within field distribution study of a pest. But, the distribution patterns of insects affect the number of samples required and the reliability of data to be used for thrips population estimation in a field. Thus, it is important to validate the minimum number of samples required from an area to reduce the variability of the data to an acceptable level. Considering the lack of information in this area, I also calculated the desired number of samples required for monitoring clumped population of *F. schultzei* in cucumber field.

In addition to within field distribution, lack of information on within plant distribution of this pest is another important issue to be addressed. Thrips exhibit differential feeding preference for various parts of its host plants. Thrips like *F. fusca* feeds on flowers; thus,
blossoms have commonly been used as a sampling unit for this pest. Likewise, *F. schultzei* is also an anthophilous species, but there are reports indicating its presence on leaves as well. Jacobson (1997) reported that adults of *F. schultzei* mainly feed on young leaves and flowers of apple, whereas larvae were found feeding on leaf buds. Tavella et al. (1996) reported that adults and larvae of *F. occidentalis* were most abundant in flowers of greenhouse grown pepper (*Capsicum annum* L.), which is contradictory to the study by Higgins (1992), who found larvae feeding on leaves of greenhouse-grown pepper. Seal et al. (2006) observed variable abundance of chilli thrips, *S. dorsalis* on different parts of ‘Scotch Bonnet’ pepper *Capsicum chinense* Jacq. They found chilli thrips were most abundant on the top young leaves followed by middle leaves and lower leaves. Such reports trigger the need to select an appropriate sampling unit for an in-depth study of *F. schultzei* feeding behavior.

The goal is to develop management programs to address populations according to pest distribution patterns. Thus, the main objective of this study is to investigate within field distribution of *F. schultzei* and determine the sample size required for estimation of *F. schultzei* population in a field. Furthermore, efforts have been made to understand the within plant distribution pattern of *F. schultzei* and its abundance during the cucumber growing season in Homestead, FL.

**Materials and Methods**

**Field Preparation:** The soil type for all experimental fields in this study is Krome gravelly loam (loamy-skeletal, carbonatic hyperthermic lithic Udorthents), which consists of about 33% soil and 67% limestone pebbles (> 2mm). The fields were prepared by using standard commercial practices (Olson and Santos 2010).
Crop management: At all the sites, cucumber, *Cucumis sativus* L. var. ‘Vlaspek’, was directly seeded on a flat ground. Seeds were sown 15.2 cm apart within the row and 91.4 cm apart between rows. At planting, 8-16-16 (N-P-K) was applied at 908 Kgs/ha in furrow; and Halosulfuron methyl 55 ml/ha (Sandea®, Gowan Company LLC., Yuma, Arizona) was used as a pre emergence herbicide to control weeds. Pyraclostrobin at 0.8 l/ha (Pristine®, BASF Ag Products, Research Triangle Park, NC) and Chlorothalonil at 1.75 l/ha (Bravo®, Syngenta Crop Protection, Inc., Greensboro, NC) were used in rotation at two-week intervals to prevent fungal diseases. The crops were irrigated twice a week with 3 cm of water using overhead sprinklers. Fertilizer 4-0-8 (N-P-K) at a rate of 2.2 Kg.ha⁻¹ was used once a week as an in-furrow band in all the fields and its use was initiated three weeks after planting. *Bacillus thuringiensis* based insecticides, Dipel DF® (var. kurstaki) at 1.1 Kg.ha⁻¹ and Xentari DF® (var. kustaki) at 1.2 L.ha⁻¹ (Valent Biosciences Corporation, Libertyville, IL) were used to control melon worms, *Diaphania hyalinata* (L.) and pickle worms, *D. nitidalis* (Stoll) in the field.

Within-Plant Distribution

Within-plant distribution of *F. schultzei* on cucumber was studied during three cropping seasons, fall 2008, spring 2009 and fall 2009 in two commercial fields (Field A & B) each season. The size of different fields in the study ranged from 0.05-0.5 hectares (ha). Selection of these study areas at various sites was to extract information on the distribution of *F. schultzei* in a wider area.

Season 1 (Fall 2008)

Study Area A: During the first cropping season, cucumber was planted on a 0.4 ha plot, which was a part of a commercial field in Homestead (N 25° 33’ W 80° 33’). The field was planted on Sep 10, 2008 and managed using cultural practices as described
above. The field was divided into four equal blocks, where each block consisted of 20 meter long, 40 rows of plants. Each block was divided into 10 plots, consisting of 20 m long 4 rows of cucumber plants. On 10th and 17th Oct, ten plants from each plot were randomly selected. Each plant was stratified into three sections: a freshly emerged terminal leaf bud (2-5 days old), a middle leaf (5th leaf from the top), a bottom leaf (8th fully-grown leaf from the top) and a flower with no preference for the site of flowers to be picked. Thus, from each plant, a newly emerged leaf bud, two leaves and a flower were collected at the time of sampling. The samples belonging to a stratum of a plant collected from one plot were placed in one ziplock® bag (17 X 22 cm) marked with the date, plot number, block number and sample type. All samples were transported to the Vegetable IPM laboratory, TREC, Homestead where samples were placed in a one quart plastic cup with 75% ethanol for 30 minutes to dislodge various life stages of thrips. The samples were carefully taken out of the cup leaving thrips in alcohol. The contents in alcohol were sieved using a 25 µm grating, USA Standard Testing Sieve (W. S. Tyler, Inc.) as per Seal and Baranowski (1992). The residue in the sieve was washed off with 75% alcohol in to a Petri dish and checked under a dissecting microscope at 12X to record various life stages of thrips.

**Study Area B:** The study area (N 25° 29' W 80° 29') was planted on Sept. 15, 2008 by direct seeding and managed following cultural practices as described above. A ~0.11 ha study area in the field was divided into 3 equal sized blocks, each consisting of 56 rows of cucumber plants each 15 m long. Blocks were divided into 14 equal sized plots consisting of 10 m long four adjacent rows. The field was sampled on Oct 18 and
Oct 26. In order to collect samples, 10 plants were randomly selected, and samples were collected and processed as above.

**Season 2 (Spring 2009)**

**Study Area A:** The study area (N 25° 30’ W 80° 28’) was planted on March 2nd and for sampling the designated study area was divided into four equal blocks, where each block consisted of 10 rows, 15 m long, covering a 0.08-ha area. The field was sampled on 11 and 17 April, 2009 during 6th and 7th week after planting. For sampling, twenty plants were randomly selected in each plot, stratified, sampled and processed as described above.

**Study Area B:** The study area (N 25° 29’ W 80° 31’) was planted by direct seeding on March 10, 2008. The study area ~0.2 ha was divided into six equal sized blocks measuring 0.03 ha, where each plot consisted of 4 rows of plants, 61 m long. The field was sampled on 8 and 17 April, 2009 during 4th and 5th week after planting. On each sampling date, 20 plants were randomly selected in each plot. Each plant was stratified to collect a flower, upper leaf, middle leaf and lower leaf sample. Samples collected from different plots were placed in separate bags (17 X 22 cm) marked with the date, plot number, block number and sample type. Samples were processed as discussed above.

**Season 3 (Fall 2009)**

**Study Area A:** During the second year, `Vlaspek’ cucumber was direct seeded on Sep1, in a commercial field (N 25° 36’ W 80° 29’) in which a 0.27 ha study area was designated (Figure 4-1). The study area was divided into four equal blocks consisting of 35 rows of cucumber plants, 15 m long. Each block was divided into seven-equal plots carrying 15 m long, 5 rows of cucumber plants. Ten plants were randomly selected in
each plot. Each plant was stratified and sampled as above from the beginning until the
end of flowering. Samples were collected on Sep 25, Oct 3, 10, 17, 24 and 29. All
materials and methods involved in collection and processing of samples were as
discussed above.

**Study Area B:** The study area B was in an adjacent field (N 25° 36’ W 80° 29’)
under the same management program. It was a 0.27 ha field, seeded on Sep 8. The
field was sampled on Oct 5, 12, 18, 24 and 31. Materials and methods for planting and
sectioning field, and sampling and processing samples were as discussed above.

**Statistical analysis**

Data were analyzed independently for each field and growing season. Data on the
abundance of larvae and adults from each field was averaged for all the samplings.
Because I wanted to determine the preferred plant parts by larvae and adult, the mean
number of larvae and adults per 10 crown, middle leaf, bottom leaf and flower per field
was analyzed using one way analysis of variance (ANOVA) (PROC GLM, SAS Institute
Inc. 2003). Data were transformed by log$_{10}$(x+1) to comply with model assumptions
before analysis. Untransformed means and standard errors are reported in figures.
Differences among means of larvae and adult on various plant parts were separated
using Tukey’s HSD (Honestly significant difference) test ($P < 0.05$).

**Spatial Distribution**

Spatial distribution of *F. schultzei* was studied during the fall cropping seasons of
2008 and 2009. Each season, three study areas within commercial fields were
employed to conduct these studies. `Vlaspek’ cucumber was directly seeded on flat
ground and fields were managed following standard cultural practices as described
above.
Season 1 (Fall 2008)

Study Area A: The study area measuring 0.24 ha and located at N 25° 24’ W 80° 32’ was divided into 64 plots. Each plot of area 37 m² consisted of 12.5 m long three rows of cucumber. From each of these plots, ten flowers (one flower/plant) were randomly collected and processed as described in the previous study to record thrips count. These plots were later pooled for analysis in various combinations forming variable sized plots for the study i.e., 74 m², 148 m², 296 m² and 592 m² corresponding to 2, 4, 8 and 16 combined plots.

Study Area B: The study was conducted in study area B (N 25° 29’ W 80° 29’) used for within plant distribution study in fall season (2008). The study area was divided into 42 equal sized plots of 23.33 m². Samples were taken and processed as above. Plots were pooled for analysis in a combination of 3, 7 and 14 plots forming bigger plots of size 70 m², 180 m² and 360 m², respectively.

Study Area C: The study was conducted in study area A (N 25° 33’ W 80° 33’) used for within plant distribution study in the fall of 2008. The field was divided into 40 equal sized plots of 100 m² and sampled as above. The plots were pooled in sets of 2, 4, and 10 forming plots of size 200, 400 and 1000 m² area for analysis.

Season 2 (Fall 2009)

In fall 2009, study areas A, B and C were located at N 25° 36’ W 80° 29’ and samples were collected from onset until conclusion of flowers in the crop. Figure 4-1 shows the arrangement of the three study area within a commercial field under the same management.
Study areas A and B were also used for within plant distribution study during fall 2009, although only flowers were used for the present study. Planting dates were Sep 1 and Sep 8, respectively. Sampling dates for the two study areas are given in Table 4-3 and Table 4-5.

Study Area C: The study area was planted on Sep 15. The study area was divided into four equal sections consisting of 15 m long 35 rows of cucumber plants. Each block was further divided into seven equal plots made up of 15 long, 5 rows of cucumber plants. Samples were collected on Sep 25 and Oct 3, 10, 17, 24 and 29 as discussed above.

**Statistical analysis**

Spatial distribution was determined separately for larvae and adults on flowers for each sampling using Taylor’s power law (Taylor 1961) and Iwao’s patchiness regression (Iwao 1968). In season 2008, distribution of *F. schultzei* was determined for only one time during the season. While distribution of *F. schultzei* in second season (Fall 2009) was determined at 4 and 5 different times in Field A and B, C, respectively. Taylor’s power law determines the relationship between variance ($s^2$) and mean density of larvae and adults per sample by means of linear regression model:

\[
\log s^2 = \log a + b \log x \tag{1-1}
\]

where, slope ($b$) signifies degree of aggregation and $\log a$, is a sampling factor related to variability in sampling size (Southwood, 1978). Iwao’s patchiness regression expressed as:

\[
m^* + \bar{x} = \alpha + \beta x \tag{1-2}
\]
is analogous to Taylors’s power law, where \( m^* \) (mean crowding index) = \( \frac{s^2}{\bar{X}} - 1 \), and \( s^2 \) and \( \bar{X} \) are the sample variance and mean respectively. The mean crowding index was given by Lloyd (1967) to express the degree of crowding by mobile animals and it was used by Iwao to derive the Iwao patchiness regression model. The intercept (\( \alpha \)) is an Index of basic contagion, which measures the tendency of insects towards crowding, and the slope (\( \beta \)) is the density contagiousness coefficient and is analogous to the b value in Taylor’s power law. The ‘b’ and ‘\( \beta \)’ value in Taylor’s power law and Iwao patchiness regression’s patchiness regression model respectively, when greater than 1.0 represent an aggregated distribution of population. While, b and \( \beta \) values significantly < 1.0 and not significantly > 1.0, indicate a uniform and random distribution, respectively. Regression parameters were estimated using general linear regression model (PROC GLM) of Statistical analysis system (SAS Institute Inc. 2003). The goodness of fit of data set from each field to both the linear models was evaluated by the \( r^2 \) value. Student t-test was used to determine whether slope (b and \( \beta \) values of these two models were significantly different from 1.0.

In addition to these two models, an Index of dispersion (ID) was calculated as:

\[
ID = \frac{s^2}{\bar{x}} \quad [1-3]
\]

where, \( s^2 \) is sample variance and \( \bar{x} \) is mean number of \( F. schultzei \) per sample. The distribution estimated using ID is said to be aggregated if ID > 1.0 and regularly distributed if ID approaches zero. Morista (1962) suggested that the distribution changes from aggregated to random with the change in the size of area occupied by insects. To address this, we determined spatial distribution of different sized plots in
each of the experimental fields. These plots were formed by adding up small sub plots at each field in different combination forming a range of different sized plots for analysis.

**Sample Size Requirements**

Sample numbers were evaluated at three levels of precision (0.10, 0.20 and 0.40) using Wilson and Room (1982) equation, given as:

\[
n = c^2 t^2 \frac{a}{X}^{b-2}
\]  

[1-4]

Where, \(a\) and \(b\) are coefficients from Taylor’s power law regression, \(c\) is the reliability, \(n\)= sample size, \(t\) is student t-value at \(n-1\) degree of freedom and \(X\) is the mean density.

The sample size was estimated for average cumulative thrips number from three experimental fields under study in fall 2009. Estimates were made for three levels of density of *F. schultzei* larvae \((\bar{X} = 0.5, 2 \text{ and } 5)\) per sample. These densities were determined based on various samples collected in three plots during the period of study. Estimation of sample size at three levels of thrips density will help scouting personnel or growers to collect right number of samples at different levels of infestation in the field and thus apply control measures accordingly.

**Seasonal Abundance**

Study on seasonal abundance of *F. schultzei* on cucumber was conducted for fall cropping season in year 2008 and 2009. In both the years, the study was conducted at two commercial fields, where all the designated study areas ranged between 0.25- 0.5 ha. `Vlaspek` cucumber seeds were planted in each field on different dates following standard cultural practices as described earlier (Olson and Santos 2010).
Season 1 (Fall 2008)

The first trial was conducted in two fields adjacent to each other (N $25^\circ$ 25' W $80^\circ$ 32'). 'Vlaspek' cucumber was directly seeded into a Rockdale soil in the first week of Sep (2008). Seeds were spaced 15 cm within the row and 91.4 cm between rows. Cucumber was irrigated twice a week with 3 cm of water using overhead sprinklers.

For sampling, each field was divided into 4 equal blocks. Each block consisted of 56 rows, 15 m long of cucumber plants. Blocks were then divided into 14 plots consisting of 15 m long four adjacent rows. In each plot, ten flowers (one flower/plot) were randomly selected and placed in separate ziplock® bags (17 cm x 22 cm), marked with the date and block number and transported to the Vegetable IPM laboratory, TREC where samples were placed in a one quart plastic cup with 75% ethanol for 30 minutes to dislodge various life stages of thrips. The samples were carefully taken out of the cup leaving thrips in alcohol. The contents in alcohol were sieved using a 25-µm grating, USA Standard Testing Sieve (W. S. Tyler, Inc.) as per Seal and Baranowski (1992). The residue in the sieve was washed off with 75% alcohol in to a Petri dish and checked under a dissecting microscope at 12X to record various life stages of thrips. Since, flower was the sampling unit in the study; sampling was initiated at the onset till conclusion of flowering. Samples were collected once a week for six weeks during the period of study (Table 4-10)

Season 2 (Fall 2009)

The second trial to study seasonal abundance of *F. schultzei* was conducted in 2009 at two study areas also used for spatial distribution study during fall 2009 (field A and B located at N $25^\circ$ 36’ W $80^\circ$ 29’). For sampling fields at both sites were divided into four equal blocks and each block consisted of 17 m long 30 rows of cucumber plants.
The block was divided into 10 plots with three 17 m long rows in each plot. Ten flowers (a flower/plant) were randomly selected in each of the plot and placed in separate zip lock bags, marked with the date and block number and transported back to the Vegetable IPM laboratory, TREC, where samples were processed and number of thrips was recorded as discussed for trial conducted in 2008. Sampling dates have been mentioned in table 4-10.

**Statistical Analysis**

Data were analyzed independently for each year. However, the number of adults and larvae from two fields in each year (2008, 2009) was averaged over various sampling dates. The data was transformed using the square-root of \((X + 0.25)\) to stabilize error variance prior to analysis of variance. The averaged number of larvae and adults per sampling over two seasons in each year was analyzed by one way analysis of variance (ANOVA) (PROC GLM, SAS Institute Inc. 2003). Differences between means of larvae and adult count for all the sampling dates were separated using the Tukey’s HSD test \((\alpha < 0.05)\) using SAS Institute Inc. 2003.

**Results and Discussion**

**Within-Plant Distribution**

The number of larvae and adults captured on flowers was significantly higher than the leaves sampled from various sections of a plant at study area- A in 2008 \((F = 224.45; \text{df} = 2, 117; p < 0.001\) for larvae \(F = 186.57; \text{df} = 2, 117; p < 0.001\) for adults) (Figure 4-2a). Similar results were obtained for Study area- B in 2008 (Figure 4-2b).

Leite et al. (2002) reported that *F. schultzei* prefers to feed on the upper leaves to middle and lower leaves of tomato plants. Similarly, Pinent and Carvalho (1998) reported on studies where they fed *F. schultzei* on tomato leaflets to study its biology.
and life cycle. Surprisingly we did not find *F. schultzei* feeding on tomato leaves during the past two years in Homestead (discussed in Chapter 2). Gonzalez et al. (2001), while sampling cucumber leaves to monitor *T. palmi* found *F. schultzei* on leaf samples collected in Cuba, which was contradictory to our results. In order to confirm these reports, the study was repeated in spring 2009 and fall 2009. At the two fields sampled in spring 2009, we did not find any difference in the feeding preference of this pest and the mean number of *F. schultzei* was significantly larger in flowers, with a few numbers on other plant parts (Figure 4-2c, d). Similar results have been documented from the studies conducted at two sites in fall 2009. The number of *F. schultzei* adults and larvae were significantly higher in flower than other plant parts (Figure 4-2e, f).

In addition to *F. schultzei*, *T. palmi* was captured on cucumber plants during the study. The majority of *T. palmi* was found infesting leaves of cucumber plants. The number of *T. palmi* on flower samples was low. Leaves of cucumber plants sampled at all the plots during the three season study were heavily infested with *T. palmi*. *Thrips palmi* has a wide host range and prefers host of family Cucurbitaceae and Solanaceae, where adults and larvae of *T. palmi* preferably feed on leaves of its host plant (Capinera 2000).

**Spatial Distribution**

Study area A and B (2008): At the two sites, larvae of *F. schultzei* exhibited an aggregated distribution. The slope (*b* and *β*) value from two linear regression models for the entire plot sizes were significantly > 1 (*P < 0.05, Table 4-1*). The coefficients of determinant (*r^2*) for Taylor’s power law and Iwao’s patchiness regression indicated a better fit by Iwao’s patchiness regression over Taylor’s power for all the plot sizes. Similar results were obtained for adult’s distribution in Study area A, where slope values
for Taylor’s power law and Iwao’s patchiness regression \((b \text{ and } \beta)\) ranging from 1.33 to 2.65 and 1.10 to 1.29, respectively were significantly > 1 \((P < 0.05)\) (Table 4-2). The results for distribution pattern of adults in the smallest plot from two regression models were not in agreement with each other, where Iwao’s patchiness regression model with higher \(r^2\) value provided a better fit to the data. The coefficient of determinant for Taylor’s power law for adults sampled in bigger plots of Study area A and Study area B was also lower than the coefficients of determinants for Iwao’s patchiness regression model. The low \(r^2\) value suggests that Iwao’s patchiness regression gave better fit than Taylor’s power law to the data and was more appropriate in explaining the distribution of \textit{F. schultzei} (Table 4-1, 2).

The values of Index of dispersion (ID) ranging from 5.00 to 17.00 for larvae and 3.46 to 5.86 for adults in all the plots at Study area A were significantly > 1 \((P < 0.05)\) (Table 4-1). Thus, ID was in agreement with Taylor’s power law and Iwao’s patchiness regression confirming an aggregated distribution of larvae and adult populations of \textit{F. schultzei} in the field. Milne (2006) also observed such aggregated behavior of \textit{F. schultzei} in his study. He suggested that, aggregation by \textit{F. schultzei} males on plant parts is primarily to attract conspecifics for mating, possibly by release of sex pheromones. In addition, there are several reports suggesting the clumped distribution of other thrips species of family Thripidae including, \textit{T. Flavus} Schrank, \textit{T. Major} Uzel, \textit{T. Atratus} Haliday, \textit{F. occidentalis} (Pergande) and a group of flower thrips (Arevalo and Liburd 2007, Morison 1957, Kirk 1985, Terry 1995, Terry and Dyreson 1996) on various plant parts. Besides reproduction as a factor inducing clumping of \textit{F. schultzei} population, there is not much information available on factors responsible for the
aggregates forming behavior of various thrips species. We speculate that aggregations in the area could be under the influence of plant phenology, flower aggregates, temperature, fertilizer, presence of natural enemies, reproduction, low dispersal by larvae, thigmotactic behavior etc. Lack of information on these aspects is an open challenge to researchers working on thrips and any knowledge on the interplay of these factors influencing distribution will open new prospects to exploit thrips biology for developing a sound management program.

Study area- C (2008): Dispersion pattern of *F. schultzei* was different from the other two fields. Lower $b$ and $\beta$ values (not significantly >1 ($P < 0.05$)) for larvae and adults distribution, suggested a random to regular distribution of the pest in the field (Table 4-1, 2). The Iwao’s patchiness regression with $r^2$ values ranging from 0.1 to 0.97 for larvae and adults distribution provided a moderate to good fit to the data (Table 4-1, 2). However, Taylor’s power law owing to its low $r^2$ values did not show good fit to the data. Such varied distribution pattern of *F. schultzei* in various fields is in agreement with other published studies on thrips species. Seal et al. (2006) in their study reported variability in the distribution pattern of chilli thrips in two fields sampled at the same time. The reason for fluctuating distribution pattern for thrips between fields in the same season is not known. Given that, *F. schultzei* infestation in field cucumber starts at onset of flowering in the fourth week after planting. I assume that, while we sampled the field in the seventh week after planting, thrips invading the field had enough time to infest the whole area and establish during the course of time. Thus, with increasing competition amongst conspecifics for food and space, there could have been local dispersion by various life stages leading to a more random distribution of the pest in the
infested area. However, these assumptions were made based on single sampling done in each of the three plots. Thus, in year 2009 we sampled another three cucumber fields beginning the flower initiation until the conclusion. Plots in each of the three study areas were pooled in a combination of seven and fourteen.

Study area- A (2009): The results from the first sampling suggested an aggregated distribution of larvae and adults in larger plots (1260 m$^2$) of the field. The slope ($b, \beta$) values for larvae and adults, from Taylor’s power law and Iwao’s patchiness regression model were, $b = 2.08, 6.51$ and $\beta = 1.50, 5.51$, respectively (Table 4-3, 4). In our field studies, we observed that $F. schultzei$ infestation begins from the edges of a field, with gradual dispersal inside the field. Thus, the aggregation observed in our plots during the first sampling could be due to the presence of thrips in the outer edges of the field. The $b$ and $\beta$ values for larvae during next three samplings were not significantly > 1 ($P < 0.05$) indicating a random to regular distribution pattern (Table 4-3). The $r^2$ values from Taylor’s power law and Iwao’s patchiness regression ranged from 0.73 to 0.99, indicating a good fit to the data collected from larger plots during these sampling. However, the coefficient of determination ($r^2$) for data from smaller plots for the two models was low, suggesting poor fit to the data (Table 4-3). The random distribution during the early three weeks could be attributed to the low thrips density in the field that reduced the chances of thrips captured during samplings (Southwood 1978).

High $b$ and $\beta$ values from samples collected during fifth week suggested an aggregation of thrips larvae in the field, while thrips population was at peak during this time of the cropping season. We assume that the aggregation of thrips larvae is due to the increase in population density of thrips in the area, which concurs with Morisita
(1962). The author from his study suggested that any change in distribution from random to aggregate or vice versa could results from change in the thrips density. Correspondingly, the distribution pattern of adults was also clumped in larger plots (1260 m$^2$) for remaining five samplings except fourth and sixth sampling during the study. The values of $b$ ranged from 1.66 to 5.32 for Taylor’s power law and from 1.11 to 6.05 for Iwao’s patchiness regression (Table 4-4). Apparently, adults exhibited an aggregated distribution in the area until the last sampling. The regular distribution of adults in the matured crop during last sampling was due to low density of adults in the field. The low population density could be due to the movement of adults to neighboring plots planted later in the season and offering more food resources (Figure 4-7).

Study area- B (2009): The low coefficient of determinants ($r^2$) suggested that the models did not fit well to larvae data. This could be due to low thrips density in the field, which was at initial stage of infestation (Table 4-5). However, in the next week with increase in larvae population, the two models gave a comparatively better fit to the data and slope values indicated a regular distribution of larvae (Table 4-5). Similarly, adults in the first week of sampling were aggregated owing to large thrips density at the edges of the field and were randomly distributed in the next week. Both larvae and adults during the subsequent weeks showed fluctuation in distribution pattern that could be due to the environmental conditions, which affected thrips population density in the season. The goodness of fit of Iwao’s patchiness and Taylor’s power law to the data improved with subsequent samplings for both larval and adult distribution. The two regression models were consistent in describing the aggregation pattern of thrips, while
Index of dispersion did not show agreement for larvae and adult distribution in this study.

Study area- C (2009): Slope values for larvae ($b = 1.40$ and $\beta = 1.14$) during the first week were not significantly $>1$ ($P < 0.05$), suggesting a random distribution of larvae in the field (Table 4-7). Conversely, on comparing the adults distribution we found that slope values ($b = 1.24$ and $\beta = 1.25$) were significantly $>1$ ($P < 0.05$) describing an aggregated distribution of adults. The aggregated distribution could be due to high adults count on the edges of the field. The three models used for describing larvae distribution were in agreement with each other having high $r^2$ values suggesting a good fit of the models to the data (Table 4-7). The Index of dispersion for the adult’s population described a regular distribution during the first two weeks. The results of ID were not in agreement with Taylor’s power law and Iwao’s patchiness regression. During the subsequent weeks (2$^{nd}$ and 3$^{rd}$) with an increase in thrips density, both larvae and adults were aggregated in their pattern of distribution. These results were well supported by high $r^2$ values for the two regression models (Table 4-7, 8). The Index of dispersion was in agreement with the two models for larvae and adults distribution during third and fourth week of sampling.

On comparing the overall pattern of *F. schultzei* between different fields in two years of study, we conclude that depending on thrips density, *F. schultzei* exhibited varied distribution patterns. During peak population densities, *F. schultzei* was found to be aggregated at all the fields, forming hot spots in the entire area under infestation. Between the two linear models used to estimate population distribution, high $r^2$ for Iwao’s patchiness suggested a better fit to the data using this model than Taylor’s
power law. This information will help to determine the distribution pattern of *F. schultzei* in the field relative to its population density and response of various models to species specific data to conduct selective management practices.

**Sample size Requirement**

The results for estimation of sample size based on various population densities are given in Table 4-9. We observed that the sample size increased with increase in levels of precision ranging from 0.30 to 0.20. At an average number of two larvae per flower in 0.27 ha field, the number of samples required ranged from 35 to 79 at 0.30 to 0.20 levels of precision, respectively. The number of samples required at this density for three levels of precision to inspect an infestation in a field is economical and practical. However, the large sample sizes like 273, required at 0.20 precision level when the predetermined population density was 0.5, is time consuming and economically unsound. Southwood (1978) suggested 0.25 as the recommended precision level to assess the population density; damage inflicted and control studies. At 0.25 level we determined 175 as the size of samples to be collected in a 0.27 ha field, which is feasible and non-destructive to the crop in field.

Since, the study was conducted with the aim to reduce the efforts of collecting large samples in the field by growers and scouting personnel. We assume that the estimates made on sample sizes for two levels of infestation will help growers collect minimum and adequate samples required to determine the correct threshold level of pest in fields.

**Seasonal Abundance**

In fall 2008, we found that density of adults and larvae during the growing season was inconsistent. *Frankliniella schultzei* is a flower thrips and thus the infestation in the
field began in a week after the flower initiation (Table 4-10). The adult populations peaked during the third (Oct. 8) and fifth (Oct. 22) week of sampling. The highest number of adults was reported during the fifth week with average number of 34 adults per ten flowers. The larvae population varied during the season. It grew rapidly in the second week of sampling and was highest during the fourth (Oct. 15) and fifth (Oct. 22) week of sampling. Both larvae and adults counts gradually started decreasing by the sixth week as the crop begins to senesce. Similarly, in fall 2009, the adult population increased with increase in flower number in cucumber crop. The population peak for adults was observed during the fifth week (Oct. 25) of sampling, with average number of 25 adults per ten flowers (Table 4-10). The larvae number also increased with the progression of time during the cropping season, where the highest number of larvae was recovered during the fifth week.

On comparing the average number of thrips obtained in 2008 and 2009, we found that thrips density was higher in 2009. This could be due to the high temperature during fall 2009, as speculated by Leite et al. (2002), while working on F. schultzei, a pest of tomato plants in Brazil. The average temperature (60 cm above ground) during 2009 cropping season was 78 ± 3.0 °F (Average temperature ± Std. dev) in comparison to 76± 5.0 °F during fall 2008 cropping season (Florida Automated Weather Network at http://www.fawn.ifas.ufl.edu). High temperature during fall 2009 may have increased the population growth rate and thus high thrips density during the season. Our observations on F. schultzei were in agreement with other studies suggesting that increasing temperatures leads to increased thrips development rate and population density (Lowry et al. 1992, Lewis 1997, Kirk 1997).
During the two cropping seasons, we observed the invasion of *F. schultzei* with the onset of flowering in the field. We assume that this could be due to the local dispersion by the thrips population from adjacent uncultivated crops to host plants. Chellemi et al. (1994) reported that, flower thrips belonging to the genus *Frankliniella* often colonize their host plant in large numbers during the flowering stage. However, colonization by these flower thrips lasts only for short period due to the short flowering period of vegetable crops (Salguero-Navas et al. 1991). Given that the flower initiation does not assure the immediate infestation of thrips populations as different thrips species vary in their timing of infestation, it is important to determine the species-specific population dynamics in the field. Through this study, we addressed the population dynamics of *F. schultzei* in fall cropping season and determined the peak population period during the growing season. The results from our study will help develop suitable sampling protocols for *F. schultzei* and will guide the scouting personnel and growers to time control measures effectively.
Table 4-1. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* larvae sampled in fall 2008

<table>
<thead>
<tr>
<th>Study area</th>
<th>N</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²  a   b</td>
<td>r²  α  β</td>
<td>ID</td>
</tr>
<tr>
<td>A</td>
<td>2</td>
<td>74</td>
<td>0.25 -1.83 2.21 Agg</td>
<td>0.95 -3.66 1.25 Agg</td>
<td>5.00 Agg</td>
</tr>
<tr>
<td></td>
<td>4</td>
<td>148</td>
<td>0.46 -1.72 2.50 Agg</td>
<td>0.87 -16.07 1.52 Agg</td>
<td>10.66 Agg</td>
</tr>
<tr>
<td></td>
<td>8</td>
<td>296</td>
<td>0.70 -2.69 3.16 Agg</td>
<td>0.81 -16.21 1.56 Agg</td>
<td>12.53 Agg</td>
</tr>
<tr>
<td></td>
<td>16</td>
<td>592</td>
<td>0.53 -3.11 3.53 Agg</td>
<td>0.64 -25.19 1.84 Agg</td>
<td>17.00 Agg</td>
</tr>
<tr>
<td>B</td>
<td>3</td>
<td>70</td>
<td>0.08 0.12 0.76 Reg</td>
<td>0.68 -1.99 1.41 Agg</td>
<td>1.67 Agg</td>
</tr>
<tr>
<td></td>
<td>7</td>
<td>180</td>
<td>0.35 -1.59 3.08 Agg</td>
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</tr>
<tr>
<td></td>
<td>14</td>
<td>360</td>
<td>0.99 -9.41 12.82 Agg</td>
<td>0.97 -21.63 4.55 Agg</td>
<td>2.10 Agg</td>
</tr>
<tr>
<td>C</td>
<td>2</td>
<td>200</td>
<td>0.10 1.65 -0.16 Reg</td>
<td>0.16 10.63 0.62 Reg</td>
<td>4.54 Agg</td>
</tr>
<tr>
<td></td>
<td>4</td>
<td>400</td>
<td>0.23 1.62 -0.01 Reg</td>
<td>0.56 5.91 0.82 Reg</td>
<td>3.52 Agg</td>
</tr>
<tr>
<td></td>
<td>10</td>
<td>1000</td>
<td>0.15 1.03 0.42 Reg</td>
<td>0.77 1.01 1.04 Ran</td>
<td>2.94 Agg</td>
</tr>
</tbody>
</table>

*N= Number of plots pooled. Agg, aggregated distribution, b significantly (P ≤0.05) >1; Reg, regular distribution, b significantly < 1 (P ≤0.05); Ran, random distribution, b not significantly different from 1 (P >0.05)."
Table 4-2. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* adult sampled in fall 2008

<table>
<thead>
<tr>
<th>Study area</th>
<th>N*</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²</td>
<td>a</td>
<td>b</td>
</tr>
<tr>
<td>2</td>
<td>74</td>
<td></td>
<td>0.19</td>
<td>-2.45</td>
<td>2.66</td>
</tr>
<tr>
<td>A</td>
<td>4</td>
<td>148</td>
<td>0.11</td>
<td>-0.09</td>
<td>1.33</td>
</tr>
<tr>
<td>8</td>
<td>296</td>
<td></td>
<td>0.36</td>
<td>-1.90</td>
<td>2.65</td>
</tr>
<tr>
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<td>592</td>
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<td>-0.49</td>
<td>1.77</td>
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<td>0.17</td>
<td>-0.17</td>
<td>1.25</td>
</tr>
<tr>
<td>B</td>
<td>7</td>
<td>180</td>
<td>0.44</td>
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<td>1.99</td>
</tr>
<tr>
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<tr>
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<td>1.16</td>
</tr>
<tr>
<td>C</td>
<td>4</td>
<td>400</td>
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<td>0.28</td>
<td>2.07</td>
<td>-0.73</td>
</tr>
</tbody>
</table>

*N= Number of plots pooled. Agg, aggregated distribution, b significantly (P ≤0.05) >1; Reg, regular distribution, b significantly < 1 (P ≤0.05); Ran, random distribution, b not significantly different from 1 (P >0.05).
Table 4-3. Taylor’s power law, Iwao’s patchiness regression parameters for distribution of *F. schultzei* larvae sampled in fall 2009 at Study area A on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²</td>
<td>a</td>
<td>b</td>
</tr>
<tr>
<td>Sep 27</td>
<td>7</td>
<td>630</td>
<td>0.19</td>
<td>-0.34</td>
<td>-2.62 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.83</td>
<td>-0.33</td>
<td>2.08 Agg</td>
</tr>
<tr>
<td>Oct 3</td>
<td>7</td>
<td>630</td>
<td>0.05</td>
<td>0.16</td>
<td>0.53 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.73</td>
<td>0.10</td>
<td>0.87 Ran</td>
</tr>
<tr>
<td>Oct 10</td>
<td>7</td>
<td>630</td>
<td>0.01</td>
<td>2.96</td>
<td>0.03 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.86</td>
<td>3.05</td>
<td>0.07 Reg</td>
</tr>
<tr>
<td>Oct 17</td>
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<td>2.89</td>
<td>1.10 Ran</td>
</tr>
<tr>
<td></td>
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<td>0.99</td>
<td>2.27</td>
<td>1.14 Ran</td>
</tr>
<tr>
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<td>0.90 Ran</td>
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<td>0.99</td>
<td>1.37</td>
<td>1.97 Agg</td>
</tr>
<tr>
<td>Oct 29</td>
<td>7</td>
<td>630</td>
<td>0.81</td>
<td>-0.89</td>
<td>2.20 Ran</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99</td>
<td>-1.09</td>
<td>2.32 Agg</td>
</tr>
</tbody>
</table>

*N= Number of plots pooled. Agg, aggregated distribution, b significantly (P ≤0.05) >1; Reg, regular distribution, b significantly < 1 (P ≤0.05); Ran, random distribution, b not significantly different from 1 (P >0.05).*
Table 4-4. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* adult sampled in fall 2009 at Study area A on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N*</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²  a  b</td>
<td>r²  α  β</td>
<td></td>
</tr>
<tr>
<td>Sep 29</td>
<td>7</td>
<td>630</td>
<td>0.91 0.13 5.12 Agg</td>
<td>0.90 -3.20 4.50 Agg</td>
<td>0.79 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.98 0.20 6.51 Agg</td>
<td>0.91 -4.11 5.51 Agg</td>
<td>0.74 Reg</td>
</tr>
<tr>
<td>Oct 3</td>
<td>7</td>
<td>630</td>
<td>0.79 0.04 1.66 Agg</td>
<td>0.65 -0.43 1.67Agg</td>
<td>1.75 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.95 -0.73 5.31 Agg</td>
<td>0.86 -7.7 6.05Agg</td>
<td>2.09 Agg</td>
</tr>
<tr>
<td>Oct 10</td>
<td>7</td>
<td>630</td>
<td>0.96 -2.8 3.89 Agg</td>
<td>0.98 -8.3 1.73 Agg</td>
<td>5.7 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99 -1.59 2.86 Agg</td>
<td>0.97 -5.59 1.56 Agg</td>
<td>5.50 Agg</td>
</tr>
<tr>
<td>Oct 17</td>
<td>7</td>
<td>630</td>
<td>0.57 -1.07 2.47 Agg</td>
<td>0.73 1.71 1.27Agg</td>
<td>8.72 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.90 0.82 1.08 Ran</td>
<td>0.98 7.02 1.03 Ran</td>
<td>8.79 Agg</td>
</tr>
<tr>
<td>Oct 24</td>
<td>7</td>
<td>630</td>
<td>0.10 0.98 1.03 Ran</td>
<td>0.38 9.61 1.03 Ran</td>
<td>11.63 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.91 0.62 1.28 Agg</td>
<td>0.99 6.70 1.11 Agg</td>
<td>10.79 Agg</td>
</tr>
<tr>
<td>Oct 29</td>
<td>7</td>
<td>630</td>
<td>0.11 1.34 0.62 Reg</td>
<td>0.38 9.45 0.89 Ran</td>
<td>8.91 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99 5.04 -2.5 Reg</td>
<td>0.99 39.58 -1.21Reg</td>
<td>9.07 Agg</td>
</tr>
</tbody>
</table>

*N* = Number of plots pooled. Agg, aggregated distribution, *b* significantly (*P* ≤0.05) >1; Reg, regular distribution, *b* significantly < 1 (*P* ≤0.05); Ran, random distribution, *b* not significantly different from 1 (*P* >0.05).
Table 4-5. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* larvae sampled in fall 2009 at Study area B on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N*</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²</td>
<td>a</td>
<td>b</td>
</tr>
<tr>
<td>Oct 5</td>
<td>7</td>
<td>630</td>
<td>0.01</td>
<td>-0.84</td>
<td>0.88 Ran</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.03</td>
<td>-1.14</td>
<td>1.01 Ran</td>
</tr>
<tr>
<td>Oct 12</td>
<td>7</td>
<td>630</td>
<td>0.56</td>
<td>0.07</td>
<td>0.93 Ran</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.87</td>
<td>0.11</td>
<td>-0.02 Reg</td>
</tr>
<tr>
<td>Oct 18</td>
<td>7</td>
<td>630</td>
<td>0.59</td>
<td>-2.72</td>
<td>5.24 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.95</td>
<td>-1.31</td>
<td>3.15 Agg</td>
</tr>
<tr>
<td>Oct 24</td>
<td>7</td>
<td>630</td>
<td>0.92</td>
<td>-12.6</td>
<td>9.7 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.96</td>
<td>3.01</td>
<td>-0.34 Reg</td>
</tr>
<tr>
<td>Oct 31</td>
<td>7</td>
<td>630</td>
<td>0.22</td>
<td>-1.15</td>
<td>2.25 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.97</td>
<td>5.62</td>
<td>-1.37 Reg</td>
</tr>
</tbody>
</table>

*N* = Number of plots pooled. Agg, aggregated distribution, *b* significantly (*P* ≤0.05) >1; Reg, regular distribution, *b* significantly <1 (*P* ≤0.05); Ran, random distribution, *b* not significantly different from 1 (*P* >0.05).
Table 4-6. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* adult sampled in fall 2009 at Study area B on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N*</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²  a   b</td>
<td>r²  α   β</td>
<td></td>
</tr>
<tr>
<td>Oct 5</td>
<td>7</td>
<td>630</td>
<td>0.86 0.10 1.42 Agg</td>
<td>0.79 -0.57 2.06 Agg</td>
<td>0.91 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.93 0.19 1.78 Agg</td>
<td>0.98 -0.82 2.43 Agg</td>
<td>0.84 Reg</td>
</tr>
<tr>
<td>Oct 12</td>
<td>7</td>
<td>630</td>
<td>0.19 -0.20 -0.67 Reg</td>
<td>0.10 0.79 0.05 Reg</td>
<td>0.81 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.98 -0.13 0.83 Ran</td>
<td>0.96 0.08 0.86 Ran</td>
<td>0.73 Reg</td>
</tr>
<tr>
<td>Oct 18</td>
<td>7</td>
<td>630</td>
<td>0.76 0.18 0.91 Ran</td>
<td>0.53 0.69 0.83 Ran</td>
<td>1.55 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99 0.18 1.31 Agg</td>
<td>0.98 -0.00 1.53 Agg</td>
<td>1.46 Agg</td>
</tr>
<tr>
<td>Oct 24</td>
<td>7</td>
<td>630</td>
<td>0.24 1.07 0.67 Ran</td>
<td>0.99 0.49 0.88 Ran</td>
<td>4.33 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.92 0.52 1.12 Ran</td>
<td>0.95 3.37 1.02 Ran</td>
<td>4.97 Agg</td>
</tr>
<tr>
<td>Oct 31</td>
<td>7</td>
<td>630</td>
<td>0.86 -1.1 2.51 Agg</td>
<td>0.87 -6.82 1.63 Ran</td>
<td>10.75 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.95 5.11 -1.85 Reg</td>
<td>0.98 0.94 -0.30 Reg</td>
<td>12.31 Agg</td>
</tr>
</tbody>
</table>

*N= Number of plots pooled. Agg, aggregated distribution, b significantly (P ≤0.05) >1; Reg, regular distribution, b significantly < 1 (P ≤0.05); Ran, random distribution, b not significantly different from 1 (P >0.05).
Table 4-7. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters distribution of *F. schultzei* larvae sampled in fall 2009 at Study area C on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N’</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²</td>
<td>a</td>
<td>b</td>
</tr>
<tr>
<td>Oct 13</td>
<td>7</td>
<td>630</td>
<td>0.98</td>
<td>0.28</td>
<td>1.13</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99</td>
<td>0.46</td>
<td>1.20</td>
</tr>
<tr>
<td>Oct 21</td>
<td>7</td>
<td>630</td>
<td>0.38</td>
<td>0.21</td>
<td>1.14</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.93</td>
<td>0.04</td>
<td>2.07</td>
</tr>
<tr>
<td>Oct 28</td>
<td>7</td>
<td>630</td>
<td>0.05</td>
<td>-0.56</td>
<td>1.71</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.86</td>
<td>3.06</td>
<td>2.45</td>
</tr>
<tr>
<td>Nov 5</td>
<td>7</td>
<td>630</td>
<td>0.65</td>
<td>-0.07</td>
<td>1.55</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99</td>
<td>0.51</td>
<td>1.67</td>
</tr>
</tbody>
</table>

*N= Number of plots pooled. Agg, aggregated distribution, b significantly (*P* ≤0.05) >1; Reg, regular distribution, b significantly < 1 (*P* ≤0.05); Ran, random distribution, b not significantly different from 1 (*P* >0.05).
Table 4-8. Taylor’s power law, Iwao’s patchiness regression and Index of dispersion parameters for distribution of *F. schultzei* adult sampled in fall 2009 at Study area C on various sampling dates

<table>
<thead>
<tr>
<th>Sampling date</th>
<th>N*</th>
<th>Plot size (m²)</th>
<th>Taylor’s power law</th>
<th>Iwao’s patchiness regression</th>
<th>Index of dispersion</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td></td>
<td>r²     a    b</td>
<td>r²   α      β</td>
<td></td>
</tr>
<tr>
<td>Oct 13</td>
<td>7</td>
<td>630</td>
<td>0.99 -0.04 1.35 Agg</td>
<td>0.81 -0.53 1.46 Agg</td>
<td>0.76 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.98 -0.12 1.24 Agg</td>
<td>0.92 -0.30 1.25 Agg</td>
<td>0.73 Reg</td>
</tr>
<tr>
<td>Oct 21</td>
<td>7</td>
<td>630</td>
<td>0.28 -0.19 0.17 Reg</td>
<td>0.08 0.48 0.13 Reg</td>
<td>0.83 Reg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.93 0.02 2.07 Agg</td>
<td>0.99 -1.05 2.11 Agg</td>
<td>0.77 Reg</td>
</tr>
<tr>
<td>Oct 28</td>
<td>7</td>
<td>630</td>
<td>0.97 -2.80 6.15 Agg</td>
<td>0.91 -4.42 2.45 Agg</td>
<td>1.61 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.99 -1.47 4.00 Agg</td>
<td>0.99 -3.62 2.22 Agg</td>
<td>1.62 Agg</td>
</tr>
<tr>
<td>Nov 5</td>
<td>7</td>
<td>630</td>
<td>0.76 -0.74 2.25 Agg</td>
<td>0.97 -4.85 1.78 Agg</td>
<td>5.78 Agg</td>
</tr>
<tr>
<td></td>
<td>14</td>
<td>1260</td>
<td>0.97 -1.40 3.15 Agg</td>
<td>0.98 -9.83 2.52 Agg</td>
<td>9.82 Agg</td>
</tr>
</tbody>
</table>

* N= Number of plots pooled. Agg, aggregated distribution, b significantly (P ≤0.05) >1; Reg, regular distribution, b significantly < 1 (P ≤0.05); Ran, random distribution, b not significantly different from 1 (P >0.05).
Table 4-9. Number of samples required for estimation of population density at three levels of precision

<table>
<thead>
<tr>
<th>Plot size (ha)</th>
<th>Levels of precision</th>
<th>Number of samples at two population densities</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td>$\bar{X}$ * = 0.5</td>
</tr>
<tr>
<td>0.27</td>
<td>0.20</td>
<td>273</td>
</tr>
<tr>
<td></td>
<td>0.25</td>
<td>175</td>
</tr>
<tr>
<td></td>
<td>0.30</td>
<td>122</td>
</tr>
</tbody>
</table>

$\bar{X}$ = number of larvae
Table 4-10. Seasonal abundance of larvae and adults on cucumber flowers sampled at two fields in 2008 and 2009

<table>
<thead>
<tr>
<th>Sampling Date</th>
<th>Number of Larvae (Mean ± SEM)</th>
<th>Number of Adult (Mean ± SEM)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Fall 2008</td>
<td></td>
</tr>
<tr>
<td>Sep 26</td>
<td>0.92 ± 0.17 c</td>
<td>1.32 ± 0.19 d</td>
</tr>
<tr>
<td>Oct 2</td>
<td>7 ± 0.76 c</td>
<td>4 ± 0.57 d</td>
</tr>
<tr>
<td>Oct 8</td>
<td>23.64 ± 1.65 b</td>
<td>32.53 ± 2.63 a</td>
</tr>
<tr>
<td>Oct 15</td>
<td>55.07 ± 4.61 a</td>
<td>25.35 ± 1.9 b</td>
</tr>
<tr>
<td>Oct 22</td>
<td>54.42 ± 4.82 a</td>
<td>34.39 ± 2.59 a</td>
</tr>
<tr>
<td>Oct 29</td>
<td>19.82 ± 2.31 b</td>
<td>12.21 ± 2.36 c</td>
</tr>
<tr>
<td></td>
<td>Fall 2009</td>
<td></td>
</tr>
<tr>
<td>Sep 30</td>
<td>1.03 ± 0.13 d</td>
<td>0.85 ± 0.15 c</td>
</tr>
<tr>
<td>Oct 5</td>
<td>1.5 ± 0.25 d</td>
<td>1.75 ± 0.35 c</td>
</tr>
<tr>
<td>Oct 12</td>
<td>48.75 ± 5.63 c</td>
<td>17.78 ± 1.92 b</td>
</tr>
<tr>
<td>Oct 18</td>
<td>66.42 ± 6.8 b</td>
<td>22 ± 2.65 ab</td>
</tr>
<tr>
<td>Oct 25</td>
<td>87.39 ± 8.7 a</td>
<td>25.89 ± 3.14 a</td>
</tr>
<tr>
<td>Oct 30</td>
<td>54.46 ± 6.22 bc</td>
<td>14.25 ± 2.09 b</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different (α = 0.05) by LSD test. Fall 2008 (Larvae: $F = 74.23$; df = 5, 162; Adults: $F = 79.45$; df = 5, 162) Fall 2009 (Larvae: $F = 56.24$; df = 5, 162; Adults: $F = 35.16$; df = 5, 162)
Figure 4-1. Pictorial view of the study areas used for within plant (A and B) and spatial distribution studies (A, B and C) during Fall 2009. (Source: www.maps.google.com)
Figure 4-2. Mean number of larvae and adults in various plant parts sampled in fall 2008 spring 2009 and fall 2009. (* and ** indicates significant difference in mean number of larvae and adults collected from various plant parts using ANOVA at $\alpha = 0.05$). A) Larvae: $F = 224.45; df = 2, 117; P < 0.001$; Adult: $F = 186.57; df = 2, 117; P < 0.001$. B) Larvae: $F = 117.30; df = 3, 164; P < 0.001$; Adult: $F$
= 67.90; df = 3, 164; P < 0.001; C) Larvae: F = 50.79; df = 3, 28; P < 0.001; Adult: F = 22.33; df = 3, 28; P < 0.001. D) Larvae: F = 64.8; df = 3, 60; P < 0.001; Adult: F = 173.82; df = 3, 60; P < 0.001; E) Larvae: F = 76.43; df = 3, 108; P < 0.001; Adult: F = 45.17; df = 3, 108; P < 0.001. F) Larvae: F = 108.54; df = 3, 108; P < 0.001; Adult: F = 34.96; df = 3, 108; P < 0.001.


Thrips palmi Karny is a polyphagous pest of vegetable crops in various parts of the world (CABI 1998). In North America, its distribution confines to Hawaii, Puerto Rico and southern parts of Florida. Since, its advent in south Florida in 1991, it has been reported as a serious pest of various greenhouse and field crops including, eggplants (Solanum melongena L.), pepper (Capsicum annum L.), potatoes (Solanum tuberosum L.), beans (Phaseolus vulgaris L.), and cucumber (Cucumis sativus L.) (Seal and Baranowski 1992). In 1993, T. palmi infesting pepper in Palm Beach County alone was responsible for the economic damage of over 10 million US $ (Nuessly and Nagata 1995). Besides, the feeding and oviposition damage, it is a vector of various plant viral diseases including Tomato Spotted Wilt Virus (TSWV) (Honda et al 1989).

Thrips palmi has a wide host range and prefers plants in the family Cucurbitaceae and Solanaceae (Capinera 2000). Adults and larvae of T. palmi preferably feed on leaves of its host plants leading to bronzing of leaves, which eventually dries and dies off. Heavy infestation of T. palmi on cucumber may lead to production of scarred, damaged or deformed fruit with no marketable value. In Homestead, T. palmi is one the major pests of field cucumbers and is a challenge to cucumber growers in the area (Personal observation).

In this region, invasion by a new adventive thrips species, Frankliniella schultzei (Trybom) in last two years on various vegetable crops has further aggravated the problem encountered by these growers. Frankliniella schultzei earlier known to make few encounters in flowers of ornamental plants in southern and central Florida (Funderburk et
al. 2007), has now established in southeastern Florida. In our preliminary study conducted to evaluate the abundance of *F. schultzei* on five different vegetable crops, Cucumber (*var.* Vlaspek*'), Pepper (*var.* King Arthur), Snap beans (*var.* Opus), Squash (*var.* Straightneck) and Tomato (*var.* Flora-Dade) we found that *F. schultzei* is a potential pest of cucumber. *Frankliniella schultzei* a key pest of tomato in several parts of South America has been reported as an important pest of cucumber also. Results from within plant distribution study, suggested that it is an anthophilous pest inhabiting flowers of its host crop. Thus, considering the pest status of *T. palmi* and damage potential of *F. schultzei*, it is important to develop a management program for the two thrips species affecting field cucumbers, an important vegetable crop grown in the County.

Chemical control has always been a primary mode of controlling thrips infesting various field crops (Morse and Hoddle 2006). However, the use of insecticides is not an absolute solution to thrips problem owing to its high cost of application, rapid selection for resistance by highly reproducing thrips and adverse effects on natural enemies and environment (Herron et al. 2007, Jensen 2000). These adverse effects of insecticides usage emphasize the need to introduce biological control agents for thrips including *F. schultzei* and *T. palmi*. Predators of the genus *Orius* (Heteroptera: Anthocoridae) are native natural enemies and have been shown to control western flower thrips, *F. occidentalis* (Pergande). The ability of *O. insidiosus* (Say) to feed on a wide range of thrips species makes it a promising biological control agent (Baez et al. 2004). Seal (1997) observed *O. insidiosus* to prey on *T. palmi*, and found that the first nymphal instar fed on 15-25 larvae of *T. palmi* each day. A study by Silveira et al. (2005) in Brazil showed that *O. insidiosus* was also effective in controlling *F. schultzei*. Other predators
used for control of various thrips species include phytoseiid mites belonging to genus *Amblyseius* (Phytoseiidae). The predatory mite, *A. swirskii* (Athias-Henriot) is receiving much attention and has been documented as a potential biological control agent of whiteflies and thrips. Another species in this genus, *A. cucumeris* (Oudemans) has been reported as an effective predator of onion thrips on greenhouse cucumber in Europe (Gillespie 1989, Van Houten and Van Stratum 1993). Furthermore, Van de Veire and Degheele (1995), and Jacobson (1997), found *A. cucumeris* to be efficient in regulating flower thrips in greenhouse conditions. Shipp and Wang (2003) used *A. cucumeris* and *O. insidiosus* as predators for the regulation of *F. occidentalis* in greenhouse tomatoes and found *A. cucumeris* to be more effective in reducing the pest population. However, Messelink et al. 2006 found *A. swirskii* to provide a better control of *F. occidentalis* than *A. cucumeris*. Considering the success of phytoseiid mites in regulating various thrips species, we evaluated the role of *A. swirskii* and *A. cucumeris* as a potential predator of *F. schultzei* and *T. palmi* inhabiting different microhabitats of the same crop. Presence of two thrips species on cucumber plants may affect the predatory behavior of the two mite species. Thus, we also investigated the persistence of predacious mites on leaves and flowers of cucumber in the presence of two thrips species.

**Materials and Methods**

**Predaceous Mites**

*Amblyseius swirskii* and *A. cucumeris* were obtained from Koppert Biological Systems Inc. (Romulus, MI). Upon arrival, mites were stored in a growth chamber maintained at 11± 2 °C, RH 60±5 %, and 14:10 h L:D until the day of release. The mites were shipped in plastic bottles of 50, 000 mites mixed with bran and bran mites as a food for the predatory mites and were stored for a maximum period of 3-4 days before release.
In order to release a desired number of mites per plant, mite number in a fixed volume of bran was estimated and standardized before release by repeatedly drawing a known volume and counting predatory mites under a stereoscopic microscope. The bottle containing bran with mites was shaken each time to ensure homogenous distribution of mites in the container before withdrawing bran for standardization. Individual estimates were made for *A. cucumeris* and *A. swirskii*. The bran used for estimation was the same commercial product to be used for release obtained from Koppert Biological Systems. The results suggested that 0.32 g and 1.70 g are the required amounts of bran to obtain 20 and 40 mites of both, *A. cucumeris* and *A. swirskii*.

**Crop Management**

Cucumber seeds (var. ‘Vlaspek’) were directly seeded on a flat ground of Krome gravelly loam soil, consisting of about 33% soil and 67% limestone pebbles (>2mm) on April 22, 2010. Seeds were spaced 15 cm within the row and 91.5 cm between rows. At planting, fertilizer 8-16-16 (N-P-K) was applied at 908 Kg/ha in furrow; and Halosulfuron methyl at 55 ml/ha (Sandea®, Gowan Company LLC., Yuma Arizona) was used as a pre emergence herbicide to control weeds. Pyraclostrobin at 0.8 l/ha (Pristine, BASF Ag Products, Research Triangle Park, NC) and Chlorothalonil at 1.75 l/ha (Bravo®, Syngenta Crop Protection, Inc., Greensboro, NC) were used in rotation at two-week intervals to prevent fungal diseases. The crops were irrigated twice a week using overhead sprinklers delivering 3 cm of water. Additional fertilizer 4-0-8 (N-P-K) was added once a week as an in-furrow band from the third week onwards after planting. *Bacillus thuringiensis* based insecticides, Dipel DF® (var. kurstaki) at 1.1 Kg/ha and Xentari DF® (var. kustaki) at 1.2 l/ha (Valent Biosciences Corporation, Libertyville, IL) were used in rotation to control
melon worms (Diaphania hyalinata L.) and pickle worms (D. nitidalis Stoll) in the experimental field.

**Field Trial**

The experiment was conducted at TREC, Homestead, FL. Phytoseiid mites were evaluated as a curative practice where the five treatments evaluated were 1) *A. cucumeris* (20 mites/plant), 2) *A. cucumeris* (40 mites/plant), 3) *A. swirskii* (20 mites/plant), 4) *A. swirskii* (40 mites/plant) and 5) control (no mites). The treatments in this study were arranged in a RCB design with four replications. Each replicate (=block) was carrying five equal sized plots, which represented a treatment. A buffer zone measuring 4.5 m was maintained between two adjacent blocks. Each plot in a block measuring 45 m² was also separated by a 4.5 m long buffer zone. The buffer zones between plots and blocks were planted to sunhemp plants, Crotalaria juncea L. to restrict the movement and mixing of predatory mites among different treatments (Figure 5-1). *In-situ* counts were done during the first week of flowering to check the abundance of thrips larvae. On detection of larvae in flowers, a single release of *A. swirskii* and *A. cucumeris* was made by the end of first week of flowering (May 27, 2010).

Releases of mites were done by using ziplock® bags filled with a standardized volume of bran + mites as described above. Each time before withdrawing bran from the bottles, it was shaken gently to ensure uniform spread of mites in the bran. In each treatment plot, these bags with an opening at its end were held upright above the plant canopy with a distance of 15 cm between plant and the bag, while I walked with a uniform speed between rows releasing fixed dosage of mites on each plant. Sampling was initiated on the sixth day after mites release (June 3, 2010) and the subsequent four samplings were done at four-day intervals during the study. Sampling units were
cucumber flowers and leaves to check the persistence and effectiveness of phytoseiid mites during the study. Ten flowers and five leaves (one flower and leaf per plant) were randomly selected from each treatment plot and placed in separate ziplock® bags (17 X 22 cm), marked with date, treatment and plot numbers. Bags were transported to the Vegetable IPM laboratory, TREC, Homestead where flower samples were placed in a one-quart plastic cup with 75% ethanol for 30 minutes to dislodge various life stages of thrips. The samples were removed from the cup leaving thrips in alcohol. The contents in alcohol were sieved using a 25 µm grating, USA Standard Testing Sieve (W. S. Tyler, Inc.) as per Seal and Baranowski (1992). The residue in the sieve was washed off with 75% alcohol in to a Petri dish and checked under a dissecting microscope at 12X to record various life stages of thrips. Thrips and mites present on leaf samples were directly counted under stereo microscope. Thrips collected from plant samples were identified to species level using thrips identification key (Nakahara 1994).

**Statistical Analysis**

Data was analyzed independently for flower and leaf samples collected. The mean number of thrips larvae, mites and mite eggs sampled on different dates from all the treatments were compared using one way analysis of variance (ANOVA) (PROC GLM, SAS Institute Inc. 2003). Data were transformed by $\log_{10}(x+1)$ to homogenize variance before analysis. Differences among treatment means for sampling dates were separated using the Tukey’s honestly significant difference (HSD) mean separation test ($\alpha = 0.05$). Linear regression analysis was done to test the relationship between thrips and mite density using PROC REG.

Mites and *T. palmi* density was expressed as accumulated mite x days and *Thrips palmi* x days per leaf, analyzed using ANOVA. Mite days and *Thrips palmi* days were...
calculated by averaging the count of mite and *T. palmi* over successive pairs of sampling
days per leaf and multiplied by four (number of intervening day between two samplings)
and accumulated over the entire study period:

\[
\left(\frac{x_n + x_{n+4}}{2}\right) \times t,
\]

[5-1]

Where \(x_n\) = number of mites or *T. palmi* at \(n^{th}\) sampling and \(t\) is number of intervening
days between two successive samplings. All the analysis for this study was done on

**Results**

**Effect of *A. swirskii* and *A. cucumeris* on *F. schultzei* populations:** Neither of
the two treatment rates of *A. swirskii* (20 and 40 mites/plant) or *A. cucumeris* (20 and 40
mites/plant) was effective in reducing the *F. schultzei* population inhabiting flowers of
cucumber plants. On the 6\(^{th}\) day after mite release (DAR), mean numbers of *F. schultzei*
in various treatment plots did not differ from the control plot (\(F = 0.62; \text{df} = 4, 15; P =
0.649\)) (Figure 5-2). Similar results were obtained for subsequent samplings done on the
10\(^{th}\) DAR (\(F = 0.43; \text{df} = 4, 15; P = 0.783\)), 18th DAR (\(F = 1.43; \text{df} = 4, 15; P = 0.248\)) and
22\(^{nd}\) DAR (\(F = 1.63; \text{df} = 4, 15; P = 0.197\)) except on the 14\(^{th}\) DAR (\(F = 5.22; \text{df} = 4, 15; P
= 0.003\)) during the study. The number of *F. schultzei* was higher in plots treated with *A.
cucumeris* than control plots on the 14\(^{th}\) DAR (Figure 5-2).

On observing the effect of four treatments on *T. palmi* after mite release, we found
that high rate of *A. swirskii* (40 mites/plant) was effective in reducing the *T. palmi* larvae
population on 6\(^{th}\) DAR (\(F = 4.53; \text{df} = 4, 95; P < 0.001\)) (Figure 5-3) and it was consistent
in suppressing *T. palmi* population during the entire cropping season. The number of *T.
palmi* in the plots treated with high rate of *A. swirskii* was significantly lower than the
control plots on 10\textsuperscript{th} DAR ($F= 14.98; df = 4, 95; P < 0.001$), 14\textsuperscript{th} DAR ($F = 12.66; df = 4, 95; P < 0.001$), 18\textsuperscript{th} DAR ($F = 9.74; df = 4, 95; P < 0.001$), and 22\textsuperscript{nd} DAR ($F=16.96; df = 4, 95; P < 0.001$) (Figure 5-3). However, the low rate of \textit{A. swirskii} was effective beginning the 14\textsuperscript{th} DAR and samples collected from these plots on 14\textsuperscript{th} DAR ($F = 12.66; df = 4, 95; P < 0.018$), 18\textsuperscript{th} DAR ($F = 9.74; df = 4, 95; P < 0.001$), and 22\textsuperscript{nd} DAR ($F = 16.96; df = 4, 95; P < 0.001$) had significantly lower thrips population than control plots (Figure 5-3). On comparing the thrips number obtained from plots treated with high and low rate of \textit{A. swirskii}, we found that thrips were significantly lower in high rate treated plots on 6\textsuperscript{th} and 10\textsuperscript{th} DAR than the plots receiving low rate of \textit{A. swirskii} (Tukey’s HSD test, $P < 0.05$).

There was no difference in thrips number in plots receiving the two rates of \textit{A. swirskii}, sampled on 14\textsuperscript{th}, 18\textsuperscript{th} and 22\textsuperscript{nd} DAR (Figure 5-3). The results from accumulated \textit{Thrips palmi} x days analysis suggested that differences among various treatments were significant ($F = 54.76; df = 4,295; P < 0.0001$), with least accumulated thrips days on plots receiving high rate of \textit{A. swirskii} and most observed on control plots (Table 5-4).

Unlike \textit{A. swirskii}, neither of the two rates of \textit{A. cucumeris} was effective in regulating \textit{T. palmi} larvae in our study. The two rates of \textit{A. cucumeris} showed reduction of \textit{T. palmi} on 18\textsuperscript{th} DAR ($F = 9.74; df = 4, 95; P < 0.001$) (Figure 5-3). The \textit{t} test suggested no significant difference in thrips density on leaves for the two different rates of \textit{A. cucumeris} on 18\textsuperscript{th} DAR ($t = 0.67; df = 38; P = 0.20$) (Figure 5-3).

**Population abundance of \textit{Amblyseius swirskii} and \textit{A. cucumeris} on cucumber flowers**: Average number of mites recovered from flowers samples during the study was small, with the highest number of mites captured from the two treatment plots on 18\textsuperscript{th} DAR (Table 5-1). Similar results were obtained for \textit{A. cucumeris}, where the number of
mites recovered from flower samples on various sampling dates was minimal. Maximum number of *A. cucumeris* mites was captured on 10th and 6th DAR from plots treated with low and high rate of mites, respectively (Table 5-1). There was no significant difference in the number of mites recovered from treated plots than control plots during the study ((6th DAR: \( F = 1.12; \text{df} = 4, 15; P = 0.36 \)), (10th DAR: \( F = 0.35; \text{df} = 4, 15; P = 0.84 \)), (14th DAR: \( F = 1.63; \text{df} = 4, 15; P = 0.19 \)), (18th DAR: \( F = 1.90; \text{df} = 4, 15; P = 0.14 \)), (22th DAR: \( F = 1.63; \text{df} = 4, 15; P = 0.19 \))) (Table 5-1).

**Population abundance of *Amblyseius swirskii* and *A. cucumeris* on cucumber leaves:** Mean number of mites recovered from the plots receiving *A. swirskii* on first sampling was low and no mites were recovered from leaves sampled from control plots (\( F = 6.61; \text{df} = 4, 95; P < 0.0001 \)) (Table 5-2). However, on the 10th DAR, there was an increase in number of mites recovered from plants treated with high rate of *A. swirskii*. Average number of mites captured on the 10th DAR from plots treated with high rate was significantly larger than the plots treated with low rate of *A. swirskii* (\( F = 81.68; \text{df} = 4, 95; P < 0.0001 \)) (Table 5-2). There was a sudden decrease in *A. swirskii* abundance on the 14th DAR. As the season progressed, mites captured from the plots treated with high rate of *A. swirskii* decreased with the lowest count observed on the 22nd DAR. The data suggests that mite’s population exhibited a decreasing trend in response to *T. palmi* population. In order to determine if thrips density was a factor affecting mite’s abundance, we performed linear regression on the mites and thrips collected on all the sampling dates, excluding 6th DAR. The \( r^2 \) value of 0.44 with \( P\)-value of 0.0005 indicated an association between thrips and mite density, suggesting that thrips density was an important factor affecting mite’s population in our study (Figure 5-4). In plots treated with
low rate of *A. swirskii*, there was a gradual increase in number of mites recovered from various samplings until 14\textsuperscript{th} DAR, after which decrease in mite number was observed (Table 5-2). The decreasing mite count with every sampling complies with decreasing thrips density as exhibited by high rate treatment plots. Given that large number of mites were recovered from low rate of *A. swirskii* treated plots, the accumulated mite days per leaf sampled from plots receiving high rate of *A. swirskii* was significantly greater than plots receiving low rate of *A. swirskii* (Tukey’s HSD test, *P* < 0.05) (Table 5-4).

The results from mite egg counts in high treatment plots suggest that the egg density followed the similar trend as that of mite’s density in the plots. Highest number of *A. swirskii* eggs from high treatment plots was counted on the 10\textsuperscript{th} DAR. The egg count was significantly higher than the eggs recovered from the low treatment plots on 10\textsuperscript{th} DAR (*F* = 60.76; df = 7, 95; *P* < 0.0001) (Table 5-3). The number of eggs decreased on the subsequent sampling dates in plots treated with higher rates. On comparing the results with plots treated with low rate of *A. swirskii*, we found that mean number of eggs/sample showed increasing trend with the progression of sampling period except on the 18\textsuperscript{th} DAR.

*Amblyseius cucumeris* populations on cucumber leaves fluctuated throughout the cropping period in plots treated with high rate of *A. cucumeris*, where the highest count was on 6\textsuperscript{th} DAR (Table 5-2). Although the number of mites recovered from plots increased numerically as the season progressed, but there was no marked increase in mite number (Table 5-2). The accumulated mite days per leaf from the plots receiving two rates of *A. cucumeris* was low and significantly different from plots receiving *A. swirskii* indicating low and constant reproduction rate of *A. cucumeris* during the season (Tukey’s HSD test, *P* < 0.05) (Table 5-4). There was no significant difference in the mites captured from the two
treatment plots on any sampling day ($P > 0.05$, Tukey’s HSD test) (Table 5-2). On comparing mite eggs count we found that, in plots treated with high rate of *A. cucumeris*, the eggs count was highest on the 6th day after release. The number of eggs counted on leaf samples for the two treatment plots on the 6th DAR was not different ($F = 1.03$, df = 4, 95; $P = 0.412$) (Table 5-3). Similar results were obtained for the two treatment plots on 10th DAR and 14th DAR, exhibiting no significant difference in the eggs counted on leaf samples ($P > 0.05$, Tukey’s HSD test). No eggs were found on leaf samples from low treatment plots sampled on 18th DAR and thereafter (Table 5-3).

**Discussion**

*Amblyseius cucumeris* and *A. swirskii* failed to control *F. schultzei* inhabiting cucumber flowers as there was no significant difference in thrips number between control plots and mites treated plots. These results are in agreement with Arevalo et al. (2009) who found that *A. cucumeris* was not effective in regulating flower thrips inhabiting blueberry flowers. In the present study, the number of mites recovered from flower samples from plots receiving *A. cucumeris* and *A. swirskii* was low. Based on this we assume that the inadequacy of mite’s populations persisting on cucumber flowers can be an important reason for the failure of thrips control in cucumber flowers. The low mite density in flowers could be the results of a behavioral preference for the microhabitat as it is possible that leaves offered better refuge and a better breeding area in comparison to flowers for the two mite species.

A biological control study by Chow et al. (2008) using *O. insidiosus* suggested that *O. insidiosus* often switched between its available preys on the crop. They reported that the minute pirate bug owing to its generalist feeding behavior exhibits preferred feeding on the easily available prey whether foraging on flowers or foliage. Similarly, we speculate
that the presence of highly abundant *T. palmi* population on cucumber leaves might have been an important factor resulting in low persistence of mites on flowers leading to failure of *F. schultzei* control. Furthermore, leaves might have offered an open arena for mites to feed on leaf feeding *T. palmi*. In order to confirm that mites were feeding on *T. palmi*, mites and *T. palmi* counts on leaf samples collected at the time of flower collection were recorded. The number of *T. palmi* among the plots treated with two rates of *A. swirskii* was found to be significantly lower than the control plots. High rate of *A. swirskii* was effective in regulating *T. palmi* beginning the sixth DAR and it continued to give suppression over *T. palmi* during the study. *A. swirskii* was recovered from sampled leaves and the large number of mite eggs on leaves suggested the successful reproduction of these mites.

The high rate of *A. swirskii* performed better than the low rate by providing suppression of *T. palmi* within a week of application. However, the low rate of *A. swirskii* was effective beginning 14 DAR. Thus, early application of this lower dosage before the threshold number of thrips population is reached can yield maximum suppression with lower resources usage. The early application of *A. swirskii* will give opportunity to these phytoseiid mites to adapt, reproduce and establish successfully on the host crop in order to suppress forthcoming high thrips abundance on the host. Thus, the application of low rate of *A. swirskii* during 2-3 weeks old crop with low thrips infestation can perform better in regulating thrips population than the application in six week old crop as in our study.

*Amblyseius swirskii* and *A. cucumeris* exhibited differential predation on *T. palmi* inhabiting cucumber leaves. One of the important reasons explaining varied predation by this mite species is differential feeding behavior, i.e., *T. palmi* as an unsuitable prey for *A.
cucumeris. In order to confirm this, we counted mites on the leaf samples to determine if mites are reproducing by feeding on another alternative food source. Results from the A. cucumeris mites and A. cucumeris eggs counts on leaf samples suggest that their density was low during most of the cropping season and it was significantly lower than A. swirskii (Tukey’s HSD test, $P < 0.05$) (Table 5-2,3). Amblyseius cucumeris exhibited little activity by reducing thrips number during the end of the cropping season, supported by high mite and mite eggs number during the end of the season. Such delayed response by A. cucumeris towards prey could be due to their slow rate of adaptability to the environment. Another probable reason for the decreased thrips count in the plots could be due to the invasion of A. swirskii in A. cucumeris treated plots. However, this possibility was overruled due to the absence of mites in flower and leaf samples collected from control plots of the study. Absence of mites in the control plots suggested that the sunhemp barriers were effective in limiting mites in their respective plots and there was no intermixing of mites among treatment plots. Thus, mites sampled from plots receiving A. swirskii were assumed to be A. swirskii and A. cucumeris to be A. cucumeris, sampled from respective plots in the study.

Another potential reason for the failure A. cucumeris in controlling T. palmi could be the low survival rate of these mites in the environment. Results from our study are in accordance with Arthurs et al. (2009) who reported that A. swirskii performed better than Neoseiulus cucumeris in regulating Scirtothrips dorsalis Hood on pepper plants under landscape conditions. This suggests that A. swirskii could be a better thrips predator in general than other phytoseiid species that have been studied. Relating our study to their report, we speculate that a possible reason for such differential behavior by these two
mite species in our study may be due to the better adaptability by *A. swirskii* to adverse environmental conditions. Owing to its Mediterranean origin (Moraes de et al. 2004), *A. swirskii* was tolerant to the hot climate of Homestead, where average temperature ranged from 18 °C (min) to 35 °C (max) during the period of study, exhibiting better performance when compared to *A. cucumeris*. Ferreira et al. (2008), suggested that plants having leaf domatia, offer a good refuge to phytoseiid mites by protecting them against unsuitable climatic conditions. However, in our study we found that irrespective of the absence of leaf domatia on cucumber plants (Arthurs et al. 2009) *A. swirskii* was efficient in reducing thrips population under adverse field conditions.

Thus, the effectiveness of *A. swirskii* on *T. palmi* inaugurates the perspective of this mite species as potential biological control agent when compared to the chemical control strategies generally used for the management of this pest. These chemical insecticides have been used on a calendar basis making it uneconomical for growers and inflicting the long-term ecological and environmental damage. In the past two decades, *T. palmi* has been known to exhibit reduced susceptibility towards various groups of insecticides including Spinosad (Seal 2010), demanding the introduction of biological control agents for this pest.

Very few studies have been conducted to evaluate mites as predator of thrips species in uncontrolled field conditions. Results from our study is an advancement over the previous available reports on phytoseiid mites as predator of thrips and adds one important thread to the study of Arthurs et al. (2009), who concluded these mites to be an effective predator of *S. dorsalis* on pepper. However, the success rate of phytoseiid mites as predators in field conditions or in nursery depends on various factors including
dissemination of this information among growers and educating them about the importance of these mites.
Table 5-1. Number of mites (mean ± SEM) per 10 flowers sampled on five sampling days from five treatment plots: 1) Low rate of *A. cucumeris* (20 mites/plant), 2) High rate of *A. cucumeris* (40 mites/plant), 3) Low rate of *A. swirskii* (20 mites/plant), 4) High rate of *A. swirskii* (40 mites/plant) and 5) Control

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<th>Treatments</th>
<th>6&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>10&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>14&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>18&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>22&lt;sup&gt;nd&lt;/sup&gt; DAR</th>
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<td>Low rate (20 mites/plant)</td>
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<td>0.16±0.10a</td>
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<tr>
<td><em>A. swirskii</em></td>
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<tr>
<td>Low rate (20 mites/plant)</td>
<td>0.50±0.20a</td>
<td>0.33±0.30a</td>
<td>0a</td>
<td>2.33±1.5a</td>
<td>0.50±0.34a</td>
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<td>High rate (40 mites/plant)</td>
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<td>1.00±0.68a</td>
<td>2.00±1.3a</td>
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Means in columns followed by the same letter are not significantly different (6<sup>th</sup> DAR: $F = 1.12$; df= 4, 15 ; $P = 0.36$; 10<sup>th</sup> DAR: $F = 0.35$; df= 4, 15 ; $P = 0.84$; 14<sup>th</sup> DAR: $F = 1.63$; df= 4, 15 ; $P = 0.19$; 18<sup>th</sup> DAR: $F = 1.90$; df= 4, 15 ; $P = 0.14$; 22<sup>nd</sup> DAR: $F = 1.63$; df= 4, 15 ; $P = 0.19$).
Table 5-2. Number of mites (mean ± SEM) per leaf sampled on five sampling days from treatment plots: 1) Low rate of *A. cucumeris* (20 mites/plant), 2) High rate of *A. cucumeris* (40 mites/plant), 3) Low rate of *A. swirskii* (20 mites/plant), 4) High rate of *A. swirskii* (40 mites/plant) and 5) Control

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<th>6(^{th}) DAR</th>
<th>10(^{th}) DAR</th>
<th>14(^{th}) DAR</th>
<th>18(^{th}) DAR</th>
<th>22(^{nd}) DAR</th>
</tr>
</thead>
<tbody>
<tr>
<td>Low rate (20 mites/plant)</td>
<td>0.53±0.19a</td>
<td>0c</td>
<td>0.06±0.06c</td>
<td>0.20±0.1b</td>
<td>0b</td>
</tr>
<tr>
<td>High rate (40 mites/plant)</td>
<td>0.93±0.18a</td>
<td>0.20±0.10c</td>
<td>0.46±0.10c</td>
<td>0.60±0.3b</td>
<td>0b</td>
</tr>
<tr>
<td>A. <em>cucumeris</em></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Low rate (20 mites/plant)</td>
<td>2.80±0.25a</td>
<td>12.53±2.0b</td>
<td>26.46±2.5a</td>
<td>24.60±7.5a</td>
<td>16.00±2.1a</td>
</tr>
<tr>
<td>High rate (40 mites/plant)</td>
<td>1.93±0.50a</td>
<td>67.40±4.74a</td>
<td>9.13±1.6b</td>
<td>11.93±2.8a</td>
<td>4.80±0.5b</td>
</tr>
<tr>
<td>A. <em>swirskii</em></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Control</td>
<td>0a</td>
<td>0c</td>
<td>0c</td>
<td>0.20±0.1b</td>
<td>0b</td>
</tr>
</tbody>
</table>

Means in columns followed by the same letter are not significantly different (6\(^{th}\) DAR: $F = 1.61$; df= 4, 95; $P < 0.37$; 10\(^{th}\) DAR: $F = 81.68$; df= 4, 95; $P < 0.0001$; 14\(^{th}\) DAR: $F = 26.82$; df= 4, 95; $P < 0.0001$; 18\(^{th}\) DAR: $F = 11.16$; df= 4, 95 $P < 0.0001$; 22\(^{nd}\) DAR: $F = 3.23$; df= 4, 95; $P < 0.0001$).
Table 5-3. Number of mite eggs (mean ± SEM) per leaf sampled on five sampling days from treatment plots: 1) Low rate of *A. cucumeris* (20 mites/plant), 2) High rate of *A. cucumeris* (40 mites/plant), 3) Low rate of *A. swirskii* (20 mites/plant), 4) High rate of *A. swirskii* (40 mites/plant) and 5) Control.

<table>
<thead>
<tr>
<th>Treatments</th>
<th>6&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>10&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>14&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>18&lt;sup&gt;th&lt;/sup&gt; DAR</th>
<th>22&lt;sup&gt;nd&lt;/sup&gt; DAR</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>A. cucumeris</em></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Low rate (20 mites/plant)</td>
<td>0.13±0.09a</td>
<td>0.20±0.10b</td>
<td>0.11±0.08b</td>
<td>0b</td>
<td>0b</td>
</tr>
<tr>
<td>High rate (40 mites/plant)</td>
<td>0.80±0.30a</td>
<td>0.20±0.10b</td>
<td>0b</td>
<td>0b</td>
<td>0.40±0.13b</td>
</tr>
<tr>
<td><em>A. swirskii</em></td>
<td></td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Low rate (20 mites/plant)</td>
<td>0.60±0.0a</td>
<td>5.40±1.20a</td>
<td>10.00±2.40a</td>
<td>6.00±2.00a</td>
<td>15.00±2.55a</td>
</tr>
<tr>
<td>High rate (40 mites/plant)</td>
<td>1.20±0.71a</td>
<td>16.93±1.56a</td>
<td>5.40±1.00ab</td>
<td>5.80±2.02a</td>
<td>2.80±0.68b</td>
</tr>
<tr>
<td>Control</td>
<td>0a</td>
<td>0b</td>
<td>0b</td>
<td>0b</td>
<td>0b</td>
</tr>
</tbody>
</table>

Means in columns followed by the same letter are not significantly different (6<sup>th</sup> DAR: *F* = 1.03; df = 4, 95; *P* = 0.41; 10<sup>th</sup> DAR: *F* = 60.76; df = 4, 95; *P* < 0.0001; 14<sup>th</sup> DAR: *F* = 9.98; df = 4, 95; *P* < 0.0001; 18<sup>th</sup> DAR: *F* = 7.24; df = 4, 95; *P* < 0.0001; 22<sup>nd</sup> DAR: *F* = 8.23; df = 4, 95; *P* < 0.0001).
Table 5-4. Number of cumulative *Thrips palmi* x days (mean ± SEM) and mite x days (mean ± SEM) per leaf on five sampling days from the following treatment plots: 1) Low rate of *A. cucumeris* (20 mites/plant), 2) High rate of *A. cucumeris* (40 mites/plant), 3) Low rate of *A. swirskii* (20 mites/plant), 4) High rate of *A. swirskii* (40 mites/plant) and 5) Control

<table>
<thead>
<tr>
<th>Treatment</th>
<th><em>Thrips palmi</em> x days (No./leaf)</th>
<th>Mite x days (No./leaf)</th>
</tr>
</thead>
<tbody>
<tr>
<td><em>A. cucumeris</em> (low rate)</td>
<td>947.73±53.8b</td>
<td>1.93±0.2c</td>
</tr>
<tr>
<td><em>A. cucumeris</em> (high rate)</td>
<td>1435.13±86.3a</td>
<td>4.63±0.6c</td>
</tr>
<tr>
<td><em>A. swirskii</em> (low rate)</td>
<td>800.07±48.9c</td>
<td>160.53±16.5b</td>
</tr>
<tr>
<td><em>A. swirskii</em> (high rate)</td>
<td>266±8.8d</td>
<td>282.90±14.0a</td>
</tr>
<tr>
<td>Control</td>
<td>958.17±56.3b</td>
<td>0.57±0.1c</td>
</tr>
</tbody>
</table>

Means in columns followed by the same letter are not significantly different when compared by Tukey’s (α = 0.05). *Thrips palmi* x days: $F = 54.76$; df = 4, 295; $P < 0.0001$; Mite x days: $F = 173.76$; df = 4, 295; $P < 0.0001$.
Figure 5-1. Pictorial view of the biological control field showing five solid rows of sunhemp (buffer) separating four blocks of treatment. A small plot of sunhemp separates each treatment plot. (Source: www.maps.google.com)
Figure 5-2. Number of *F. schultzei* larvae (mean ± SEM) per 10 flowers sampled on five sampling dates after the release of *A. swirskii* (high rate and low rate) and *A. cucumeris* (high rate and low rate). Mites were released on day 0 indicated by an arrow. On each day of sampling, treatments with no letters are not significantly different (*P* > 0.05, Tukey’s HSD test).
Figure 5-3. Number of *T. palmi* larvae (mean ± SEM) per cucumber leaf sampled on five sampling dates after the release of *A. swirskii* (high and low rate) and *A. cucumeris* (high and low rate). Mites were released on day 0 indicated by an arrow. On each day of sampling, treatments with same letter(s) are not significantly different (*P > 0.05*, Tukey’s HSD test).
Figure 5-4. Linear regression showing the number of *T. palmi* larvae and mites abundance in plots treated with *A. swirskii* (40 mites/plant) during the period of study.
CHAPTER 6
CONCLUSIONS

*Frankliniella schultzei* (Trybom) is one of the key pest of tomato and other economically important vegetable and ornamental crops across the globe. It is a pest of South America origin from where it dispersed to other parts of the world. *Frankliniella schultzei* earlier known to make few encounters in flowers of ornamental plants in southern and central Florida (Funderburk et al. 2007), has now established in southeastern Florida. One of the important factors supporting *F. schultzei* establishment is the broad polyphagy and ability to survive under intermittent suboptimal conditions.

*Frankliniella schultzei* is a new vegetable pest in this region and there is not much information on its distribution, seasonal abundance, and most importantly on their management. Considering the lack of information about this pest, and future needs of our industries as well as growers, I conducted various studies. The first study was to determine the abundance of *F. schultzei* on five economically important vegetable crops (cucumber, tomato, squash, pepper and beans) grown in Florida. We found that irrespective of high adult count of *F. schultzei* on tomato, number of larvae was significantly higher on cucumber than on the other hosts. Tomato, reported as one of the major hosts of *F. schultzei* in Cuba, Brazil and Paraguay, did not serve as primary host of this pest in Florida. This information will be useful to determine the reproductive host of this pest and crops largely at risk in this region.

Within-plant distribution study was conducted to determine the plant parts preferred by *F. schultzei* as feeding and oviposition sites. The study was conducted in field cucumber at three different sites for three seasons, and we found that *F. schultzei* feed and reproduces on flowers of its host plants. It is an anthophilous pest exhibiting
thigmotactic behavior and stays in tight secluded areas of the flower. Gonzalez et al. (2001) found adults of *F. schultzei* on cucumber leaves in Cuba, but in my study, *F. schultzei* was observed only on flower samples with insignificant number on other plant parts.

*Frankliniella schultzei* exhibited an aggregated distribution in the field. The distribution of larvae and adults in the field was studied in fall 2008 and fall 2009. The distribution pattern of larvae and adults varied in accordance to the thrips density. During peak population density, *F. schultzei* was aggregated and it formed hot spots in the area under infestation. However, irrespective of the low population density at the beginning of infestation, slope values significantly > 1, for the two models suggested the aggregated distribution. Such clumped pattern could be due to the presence of thrips on the outer edges of the field, as *F. schultzei* infestation begins from the edges of a field.

The density of *F. schultzei* larvae and adults during the cucumber cropping season in fall 2008 and 2009 was irregular. During the two cropping seasons, we observed the invasion of *F. schultzei* with the onset of flowering in fields. *Frankliniella schultzei* number increased with the progression of time during the cropping season, where the peak population was observed during the fifth week. The invasion of *F. schultzei* with the onset of flowering could be due to the local dispersion by the thrips population from adjacent uncultivated crops to host plants.

Between the two phytoseiid mites tested for biological control of *F. schultzei* in field cucumber, we found that none of the two treatment rates of *A. swirskii* and *A. cucumeris* was effective in reducing *F. schultzei* population inhabiting flowers of cucumber plants. Average number of *A. swirskii* and *A. cucumeris* recovered from
flower samples during the study was low in comparison to the number of mites released in plots. There was no significant difference in the number of mites recovered from various treatment plots.

A large number of *A. swirskii* mites were recovered from leaf samples collected from plots treated with high rate. Mites count from high rate treated plots was higher than plots treated with low rate of *A. swirskii* on 10th DAR. On comparing, the number of *T. palmi* between control plots and plots treated with high rate of *A. swirskii*, it was found that mites were effective in regulating *T. palmi* inhabiting leaves of cucumber plants. The high rate of *A. swirskii* significantly reduced *T. palmi* number by the 6th DAR. Mites recovered from leaves samples collected from plots treated with *A. swirskii* was significantly higher than plots treated with *A. cucumeris*. Mean number of *A. cucumeris* recovered from leaf samples collected from treatment plots was significantly lower than *A. swirskii* treated plots.
LIST OF REFERENCES


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Waring, S. M. 2005. Differential Predation by Orius Insidiosus (Say) on Frankliniella Occidentalis (Pergande) and Frankliniella Bispinosa (Morgan) in sweet pepper. MS Thesis/PhD Dissertation. Entomology and Nematology Department, University of Florida, Gainesville, FL.


Garima Kakkar was born in New Delhi, India in 1983. She received Bachelor of Science (hons.) in botany from Sri Guru Teg Bahadur Khalsa College, University of Delhi, India. After receiving her bachelor’s degree, she began working on a master’s degree in same school, studying agrochemicals and pest management. In 2005, she joined Indian Agricultural Research Institute as a Senior Research Fellow for two years in a project entitled “Standardization of Nitrification Inhibitory Principles of Neem (Azadirachta indica A Juss) and Neem Coated Urea”. In May 2008, she started master’s degree in the Entomology and Nematology Department, University of Florida under the supervision of Dr. Dakshina R. Seal. She studied population dynamics and biological control of F. schultzei, an invasive thrips species in Homestead. She also identified F. schultzei and various other commonly found thrips in vegetable crops of south Florida. In 2010, Garima married Vivek Kumar, a fellow entomology graduate of the University of Florida. She will begin with her PhD program on biology of termites at University of Florida in spring 2011.