LEGUMINOUS AND GRAMINACEOUS COVER CROPS FOR THE CONTROL OF INSECT PESTS IN ORGANIC SQUASH

By

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To my parents, Stanhope and Sonia, for encouraging my academic interests and my sense of scholarship, making this milestone possible

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Leguminous and graminaceous cover crops were evaluated for the control of aphids and whiteflies [*Bemisia argentifolli* (Bellows and Perring)] in organic summer squash. Two monoculture legumes, sunn hemp [SH] (*Crotalaria juncea* L.) and velvet bean [VB] (*Mucuna pruriens* (L.) DC. var. pruriens), two monoculture grasses, sorghum sudangrass [SSG] (*S. bicolor* x *S. sudanense*) (Piper) Stapf and pearl millet [PM] (*Pennisetum glaucum* (L.) R. Br.), and two legume-grass mixtures, sunn hemp/pearl millet [SH/PM] and sorghum sudangrass/velvet bean [SSG/VB] were evaluated during 2006 and 2007.

Results from unbaited yellow sticky traps showed that the monoculture grasses, specifically SSG, has the best potential for suppressing aphids and whitefly populations in organic summer squash. The diculture, legume-grass mixture consisting of SSG/VB also demonstrated potential to reduce pest numbers by enhancing the natural enemy populations' specifically parasitoids and predators. Sorghum sudangrass is especially known for harboring coccinellids. High numbers of the whitefly predator, *Delphastus pusillus* were recorded in SSG plots, which provided effective control of whiteflies.

During both years of study, there was good evidence to conclude that residual populations of natural enemies from incorporated cover crops can suppress pest populations in the

subsequent squash cash crop. However, alternate resources surrounding the field will need to be present to support natural enemy populations until the squash crop is established.

Overall, insect pest populations had a density dependent effect on squash yields following cover crop incorporation into the soil. Both legumes, VB and SH attracted more aphids and whiteflies compared with the weedy fallow and therefore would not be recommended for use prior to organic squash. The PM and SSG were able to increase squash yields, while suppressing key pest populations (aphids and whiteflies) in squash. Future research should involve analyzing both grass cover crops for chemical compounds that may be responsible for suppressing pests, with the goal that these compounds could be used for pest control in organic systems.

CHAPTER 1
INTRODUCTION

Organic farming is a method of crop and livestock production that emphasizes biological and ecological processes to improve soil health, manage soil fertility and optimize pest management while excluding the use of conventional commercial fertilizers, synthetic pesticides and synthetic antibiotics and growth hormones (Treadwell 2007, Gold 2008, Martin 2006, Bachmann 2002). Within the past decade, organic farming has gained significant momentum within the United States resulting in $13.8 billion USD in organic food sales in 2005 (Crawford 2007, Oberholtzer et al. 2005) and is estimated to continue to increase annually by 9-16% (Oberhotlzer et al. 2005). In 2005, 98,525 vegetable acres were certified organic in the U.S, which was more than twice the acreage certified in 1997 (Economic Research Service 2008). It is clear that fresh, organic vegetables are in demand. In particular, squash was ranked 4th among the top organic vegetables that were purchased in the U.S. (Oberholtzer et al. 2005).

Summer squash (*Cucurbita pepo* L.) is a major vegetable crop grown commercially in Florida for fresh market sales (Roberts and Pernezny 2001). It is also one of the few crops that is shipped out of Florida every month of the year to grocery stores nationwide (AgMRC 2007, Mossler and Nesheim 2001, Roberts and Pernezny 2001). During the 2007 squash field-season, Florida growers harvested approximately 11,000 acres of squash, with a value of ~ $53 million USD (NASS, 2008). In recent years the growth of organic squash production in Florida has increased (Juan Carlos Rodriguez personal communication).

With the continued growth of the organic squash market, insect pest management becomes exceedingly important. Squash plants are significantly damaged by squash bugs, *Anasa nistis* (DeGeer), seedcorn maggot, *Delia platura* (Meigen), wireworms (*Limonius spp.*and others), leafminers *Liriomyza* spp., western spotted (*Diabrotica undecimpunctata undecimpunctata* L.,) and stripped cucumber beetle, *Acalymma trivittatum* (Mannerheim), various armyworm *Spodoptera* spp., cabbage looper *Trichoplusia ni* (Hübner) and various forms of powdery mildew. However, more serious diseases are caused by viruses vectored by aphids [*Aphis gossypii* Glover, *Myzus persicae* (Sulzer)] and whiteflies, *Bemisia argentifolii* (Bellows and Perring), which are key insect pests of squash (Molinar et al. 2007, Kucharek and Purcifull 2001).

Squash crops are very susceptible to aphids and whiteflies (Frank and Liburd 2005, Kucharek and Purcifull 2001). Aphids transmit over 50 viruses in cucurbits resulting in loss of yield and extreme symptoms such as deformed seeds, flower necrosis, leaf chlorotic spotting and fruit bumps (Hooks et al. 1998, Kucharek and Purcifull 2001). Five of these viruses are present in Florida including the papaya ringspot virus, the watermelon mosaic virus, the zucchini yellow mosaic virus, the cucumber mosaic virus, and an unnamed potyvirus (Kucharek and Purcifull 2001). A sixth virus called the squash mosaic virus is not as frequent in Florida but is transmitted by the cucumber beetle *Acalymma vittatum* (Fabricius), not by aphids (Kucharek and Purcifull 2001).

Feeding by whiteflies, *Bemisia argentifolli* (Bellows and Perring) significantly damages squash plants via introduced toxins that cause chlorosis, distortion of new growth, and physiological disorders such as the squash silverleaf (SSL) disorder (Liburd and Frank 2007). The SSL is one of the most important physiological disorders in cucurbits caused by immature whitefly feeding. This disorder produces silvering of the leaf veins, which eventually progresses to cover the surface of the leaf. The yield of the silvered squash plant is consequently lower because of a reduction in photosynthesis. In severe cases of silvering the fruit, stem and flowers of the squash become bleached (McAuslane et al. 1996). In addition to transmitting viruses, aphids and whiteflies suck sap juices from plants and excrete honeydew, which promotes the growth of sooty mold (*Capnodium* spp.) that eventually reduces the vigor and marketability of the vegetable (Palumbo et al. 2000).

Justification

Generally, aphids and whiteflies are conventionally controlled by several broad- spectrum insecticides from classes including organophosphates, carbamates, formamidines and cyclodienes (Dewar 2007, Palumbo et al. 2001, Webb 2007). However, this method of control has become increasingly problematic over the years primarily because of the resistance of aphids and whiteflies to these chemicals. Some of these synthetic insecticides can enhance the spread of non-persistent, aphid-vectored viruses especially in cucurbits (Stapleton et al. 2002). In addition to pest resistance, insecticides have an overall negative effect on the environment by destroying non-target organisms including natural enemies and pollinators (Pimental et al. 1992). Florida experiences mild winters and heavy precipitation, which can encourage the persistence of pests year round. All these issues raise serious economic and environmental health concerns from growers, scientists and the general public. Therefore a more sustainable approach for insect pest control in organic systems is needed.

The use of cover crops is required in organic production. It is possible that their use for protecting and improving soils and suppressing weeds may affect the population of pest and beneficial insects in a cropping system. Interest in the use of leguminous and graminaceous cover crops for pest management have been continually re-evaluated over the past few years (Bugg et al. 1991, Creamer and Baldwin 2000, Tillman et al. 2004). Cover crops act as a source of food, and an excellent microhabitat for diverse communities of generalist predators to attack insect pests of vegetable crops, in addition to protecting and improving soils (Bugg et al. 1991, Lu et al. 2000, Sullivan 2003, Costello and Altieri 1995). In addition, the evaluation of various summer cover crops for use in vegetable production systems in North Carolina resulted in increased biomass within short periods of time, and increased soil fertility (in the case of legumes) (Creamer and Baldwin 2000).

Cool-season cover crops have been shown to increase the establishment of the predacious big-eyed bug, *Geocoris punctipes* (Say), via nectar and other arthropods thereby reducing egg masses of heliothine species on cantaloupe plants (Bugg et al. 1991). Cover crop combinations of leguminous and graminaceous species that establish quickly and increase soil nitrogen can have benefits by reducing weed pest populations in subsequent cash crops (Creamer and Baldwin 2000).

Goal and Hypotheses

The overall goal of my research was to evaluate the effects of summer cover crops on insect pests and beneficials in an organic system before and after the planting of a squash (*Cucurbita pepo* L.) cash crop. I hypothesized that 1) the establishment of cover crops, especially legume-grass mixtures, would enhance natural enemy populations in the organic field 2) residual populations of natural enemies following cover crop incorporation will suppress pest populations in the subsequent squash crop, and 3) cover crops would lead to increased squash yield as a result of reduced pest pressures.

Specific Objectives

- To study the population dynamics of aphids, whiteflies and beneficials (selected parasitoids and predators) in monoculture and diculture (mixed cover crop stands).
- To determine whether residual populations of beneficials from incorporated cover crops can still suppress pest populations in squash.
- To determine how pest populations from cover crop treatments affect squash yield.
- To compare several monitoring techniques for aphids and whiteflies for improvement of management techniques on organic farms.

CHAPTER 2
LITERATURE REVIEW

Aphids

There are over 4700 species of aphids in the world, but only about 100 from the family Aphididae are considered to be major pests of agricultural and horticultural crops (Powell et al. 2006, Dixon 1977, Blackman and Eastop 2007). Approximately 2 billion aphids per acre can live on above ground plant parts and an additional 260 million can live belowground. Their small size, high reproductive rates, dimorphic migratory form, and ability to transmit numerous viruses enhance their effectiveness as pests (Awmack and Leather 2007, Petterson et al. 2007, Dixon 1977). Aphids vector and transmit between up to 275 viruses in many plant species (Frank and Liburd 2005, Kucharek and Purcifull 2001, Zhang and Hassan 2003, Nebreda et al. 2004, Powell et al. 2006). Some of these viruses include the papaya ringspot virus type W, the watermelon mosaic virus 2, the zucchini yellow mosaic virus, the cucumber mosaic virus and an unnamed potyvirus (Kucharek and Purcifull 2001). These aphid-transmitted viruses have been major impediments to the consistent production of squash in Florida, and they produce symptoms that include plant stunting, leaf strapping, leaf chlorotic spotting, reduced weight, distorted stamens, interveinal and flower necorsis, green mottling, deformed seeds and ringspots (Kucharek and Purcifull 2001).

Biology and Behavior of Aphids

According to Dixon (1977) many aphids have complex life cycles. Aphids have a heteroecious or monoecious life cycle based on how they utilize their host plants (Williams and Dixon 2007). Heteroecious or host alternating aphids live on a primary host plant in the winter and a different secondary host plant in the summer and eggs are laid on the primary host after mating. However, monoecious aphids are associated with only one or very closely related host plants to complete their life cycle (Williams and Dixon 2007).

Aphids have main two types of reproduction that is used to complete their life cycle: hololytic and anhololytic. Hololytic (sexual) reproduction in aphids involves the production of eggs followed by parthenogenesis (the production of offspring without males) whereas in anhololytic (asexual) reproduction, female aphids reproduce parthenogenetically all throughout the year (Williams and Dixon 2007). Parthenogenesis doubles the intrinsic rate of population increase, thereby presenting a definite reproductive advantage by reducing the time for the aphid to become sexually mature (Dixon 1997). Short day length is known to induce a change from asexual to sexual reproduction in aphids (Dixon 1977). This response is measured by a biological clock within aphids, which correlates with a photoperiodic receptor within the mid dorsal region of the aphid's brain and causes this change.

Aphid dispersal is accomplished by two distinct morphs: a winged (alate) form and a wingless (apterous) form. Walking is the active mode of dispersal for apterous aphids, whereas for alate aphids, walking and flying are the main modes of dispersal. An aphid is able to move from one point to the other either voluntarily or involuntarily. Voluntary aphid movement is prompted by extrinsic factors such as food, mate, oviposition, escape responses from sensed attack of a natural enemy or the chemical composition of a deteriorating host plant (Irwin et al. 2007). Involuntary aphid movement is usually as a result of the force of gravity, air currents, farm machinery or animals.

Aphids will selectively choose their host plants via successive sensory and behavioral patterns. They respond phototactically to different plant-reflected wavelengths and volatiles, which trigger pre-alighting behavior. Aphids will walk back and forth across the leaf surface assessing surface cues before stylet insertion. Probing of the epidermis is followed by repeated stylet penetrations into the mesophyll, parenchyma and phloem sieve tubes to allow for sap extraction and ingestion (Powell et al. 2006). Following ingestion of sap juices, aphids excrete a sticky substance called honeydew as a waste product. Honeydew promotes the growth of black mold on the plant but also attracts honeydew-loving ants, which tend to the aphids and defend them from their natural enemies (Palumbo et al. 2000, Hooks et al. 1998). Some aphids can also inject toxic salivary secretions into plants, which cause deformed and discolored leaves, galls on leaves, stems and roots and overall stunted plant growth (Drees 1993).

Whiteflies

Whiteflies are economically destructive pests on a global scale causing great damage to horticultural crops, ornamentals, greenhouse plants, and vegetables such as *Brassica* spp. (McAuslane 2005, Simmons 2002, Nebreda et al. 2005, Simmons et al. 2002, Goolsby et al. 1998). In 1998, whiteflies were responsible for almost $1 billon USD worth of crop loss, job displacement and control measures (Goolsby et al. 1998). *Bemisia argentifolli*, the silverleaf whitefly, extracts large quantities of phloem sap and reduces yields by up to 50% in vegetables and cotton (Chu et al. 2001, Byrne and Bellows 1991). Female whiteflies lay eggs on the lower leaf surface and cause heavy infestations (Nebreda et al. 2005). Honeydew excretions from adults and nymphs promote the growth of sooty mold, and cause additional discoloration on parts of the plant that are used for food (Byrne and Bellows 1991, Nebreda et al. 2005).

In cucurbit crops such as squash, *B. tabaci* Biotype B causes severe physiological disorders such as the squash silverleaf disorder (McAuslane et al. 1996). Immature crawlers feed voraciously on squash causing the characteristic silvering of the adaxial leaf surface and blanching of the fruit in as little as 2 weeks (Frank and Liburd 2005).

Biology and Behavior of Whiteflies

Whiteflies are parthenogenetic and arrhenotokous (unmated females produce males, and mated females produce males and females), and the adults are sexually dimorphic (males are smaller than females) (McAuslane 2005, Byrne and Bellows 1991). Mating occurs once the wings of the whiteflies have expanded and hardened (McAuslane 2005). In fall and spring, mating occurs 3 days after eclosion and copulation can last up to 8 hrs in the summer months. Whiteflies are multivoltine with up to 6 generations per year and will continue to reproduce as long as temperatures permit (Bryne and Bellows 1991).

Oviposition by female whiteflies depends heavily on host plant quality and temperature. The female selects suitable host plants via color cues for alighting purposes, but the odor of crushed plants will also serve as an attractant (Byrne and Bellows 1991). Once the female whitefly lands on a plant, she goes to the lower surface, probes the leaf, and ingests a small amount of sap with her piercing mouthpart in order to test the suitability of the plant. Once the plant is deemed suitable, the female begins to feed and eventually will deposit eggs on the underside of the leaves depending on the temperature and quality of the host (McAuslane 2005, Byrne and Bellows 1991). Female whiteflies can produce between 48 to 394 eggs with an average of 252 eggs at a temperature of 28.5° C in the summer months (Byrne and Bellows 1991).

Once crawlers hatch, they rapidly walk over the leaf surface, moving within and between healthy feeding areas, eventually settling down and remaining sessile until they become adults (Smith 1999, McAuslane 2005, Byrne and Bellows 1991). The sessile nature of the crawlers makes them very susceptible to parasitoids, predators and pathogens (Smith 1999).

Once the adults emerge from a T-shaped slit in the last nymphal instar, they fly up the same plant or move to another plant (McAuslane 2005). Whitefly migrations tend to be short ranged, and in most cases dispersal within distances of more than a few hundred meters are aided by humans. Two dispersal morphs exist in populations of *B. argentifolli*: a migratory morph and a trivial-flying morph, with the latter being the most common (Byrne and Bellows 1991). Migratory morphs can travel up to 7 km whereas trivial-flying morphs can only disperse up to 150 m (McAuslane 2005, Byrne and Bellows 1991). Once food quality deteriorates, migration to another food source is enabled by wind currents, where whiteflies tend to be transported like aerial plankton. They will land on suitable hosts by chance. Short range migration allows for well-established colonies on closely spaced vegetation (Byrne and Bellows 1991).

Management of Insect Pests Using Natural Enemies

Coccinellidae (Ladybird beetles)

Predacious ladybird beetles are important in suppressing pest populations and have been more widely used in biological control programs compared with other taxa of predacious insects (Obrycki and Kring 1998). They are especially important predators of aphids, whiteflies, mealybugs, scales and mites (Pedigo 2002, Frank and Mizell 2004).

The whitefly predator, *Delphastus* spp. is predacious on the greenhouse and silverleaf whiteflies throughout their adult lives (Heinz et al. 1999). Both males and females are known to consume high numbers of whiteflies, the females have a high fecundity and a total generation time of 19 to 20 days. They work well with parasitoids to reduce whitefly numbers because of their selective nature for the type of whiteflies that they consume. These coccinellids prefer healthy whiteflies, and can distinguish between those that have already been parasitized (Heinz et al. 1999).

Coccinella septempunctata L. is an example of an aphidophagous coccinellid and is one of the most predacious beetles on different aphid species worldwide (Shannag and Obeidat 2006). Other aphidophagous coccinellids like *C. septempunctata* have responded to the presence of aphids on plants by depositing eggs near the prey and significantly reducing their populations where there is a buildup of aphid refuge (Obrycki and Kring 1998). Voracity, abundance, timing and length of coccinellid generation in relation to aphid population are factors which influence the efficacy of ladybird beetles as natural enemies (Shannag and Obeidat 2006).

Carabidae (Ground beetles)

In sustainable organic systems, carabids are well known as natural enemies for insect pests. They are polyphagous, have a long life cycle and are efficient at suppressing pest outbreaks. However, carabids are very sensitive to anthropogenic influences. Agrochemicals can significantly influence their composition, abundance and spatial distribution (Lövei and Sunderland 1996, Kromp 1999). As generalist predators, there is still some debate about the effectiveness of carabids as biocontrol agents in terms of host specificity (Symondson et al. 2002). There are some ground-dwelling beetles that are able to feed extensively on aphids as they move throughout the plant via wind or rain dislodgement (Brewer and Elliott 2004).

Many beetles exhibit iteroparity (the ability to breed and reproduce multiple times throughout their life cycle). Eggs can be laid singly or in batches with up to several hundred per female in one season (Lövei and Sunderland 1996). Eggs hatch as elongated larvae that taper towards the end. Mature beetle larvae will pupate in the soil. The life cycle usually takes 12 months to be completed, and adult ground beetles can live for as long as 3 years. Both larvae and adult beetles are known to be voracious predators on insect pests (Lyon and Purrington 2007).

Syrphidae (Syrphid Flies)

Syrphid flies are voracious on several species of aphids and become abundant during heavy aphid densities (Brewer 1995, Antonelli 2003). They have high reproductive rates and high mobility which allows them to take advantage of aphid colonies (Ambrosino et al. 2006). The adult flies are harmless and feed on honeydew, nectar (used as an energy source), and pollen (used as a source of protein for egg production) (Smith and Chaney 2007). However, the larval stage is the stage that is predacious on aphids, with one larva consuming as many as 500 aphids during its 3 week developmental period (Antonelli 2003). Once the adult females lay eggs in early summer within aphid colonies, larvae will emerge 3 to 4 days later. Syrphid flies are multivoltine and can produce 3 to 7 generations per year, going from egg to adult in 16 to 28 days depending on weather and location (Antonelli 2003).

Parasitoids

The use of parasitoids as natural enemies for insect pest management has been practiced for as long as 1600 years in China (Wei et al. 2005). Parasitoids occupy similar feeding guilds with predators such as coccinellids or carabids. Interestingly, these predators can affect parasitoid reproductive opportunities by feeding on parasitized insects (Taylor et al. 1998). In order to understand the efficacy with which parasitoids can suppress pest populations, it is necessary to understand the dynamic multitrophic (plant-herbivore-parasitoid/predator) interactions that occur (Kalule and Wright 2005). "Bottom up" factors such as plant nutrient quality affect the insect pest, and eventually the self-sustainability and efficacy of the natural enemy (Kalule and Wright 2005). It is also a common occurrence for generalist predators to disrupt the biocontrol of a pest species by a parasitoid (Kalule and Wright 2005, Symondson et al. 2002).

Braconids and ichnuemonids are the two most common groups of parasitoids that attack aphids. Aphids such as *Brevicoryne brassicae* (L.) and *Myzus persicae* (Sulzer) are both attacked and parasitized by the braconid *Diaeretiella rapae* (McIntoch) (Costello and Altieri 1995). This parasitoid specializes on cruciferous aphids but is also known for attacking other polyphagous aphids (Zhang and Hassan 2003, Costello and Altieri 1995). In living mulches, *D. rapae* consistently kept aphid populations at a minimum compared with clean cultivation (Costello and Altieri 1995).

Alternatively, the aphelinid parasitoid, *Encarsia formosa* Gahan, is well known for its control of whiteflies on greenhouse crops and vegetables (Hoddle et al. 2007). Eggs mature within 8 to10 days and the adults reduce whitefly populations by either consuming the hemolymph of their immature hosts or by ovipositing (Hoddle et al. 2007). Also, braconid parasitoids such as *Aphidius* spp. can also have a significant negative impact on aphid populations. With a complete life cycle of 10 days, females can attack as many as 300 aphids (Moschetti 2003).

Lepidopteran pests usually have egg–larval parasitoids such as *Ascogaster* spp. and *Chelonus* spp. that use polydnaviruses and venom to induce premature metamorphosis of the host, thereby allowing the parasitoid to emerge from the pre-pupal stage of the host (Beckage and Gelman 2004). Presently, the most well known egg parasitoids of lepidopteran pests come from 33 species in three families which include: Trichogrammatidae, Scelionidae and Encyrtidae. They attack a wide range of lepidopteran pests including *Helicoverpa* spp., *T. ni* (Hübner), *P. xylostella* L., *Manduca sexta* L., *P. rapae* L., *Ostrinia nubilalis* Hübner, and *Mamestra brassicae* L. (Sithanantham et al. 2001).

Insect pests attacked by parasitoids are usually parasitized and tend to die slowly. The life cycle of a parasitoid is complex. A single immature parasitoid or several hundred immatures may develop within or on one host and ultimately kill the host via different hormonal pathways before emerging as an adult (Hoffmann and Frodsham 1993, Pedigo 2002).

Parasitoid wasps have many methods of manipulating their host in order to create a medium that will support and promote the development of their offspring (Beckage and Gelman

2004). Parasitoids can be regulators or conformers. Regulators use hormonal or neurohormonal strategies to disrupt developmental pathways in their host, whereas conformers have similar endocrine signaling pathways with the host that simultaneously coordinates developmental activites such as molting (Beckage and Gelman 2004). The final instar of the host will eventually show disruption of feeding activites, molting and metamorphosis that was induced by the parasitoid (Beckage and Gelman 2004).

Management of Insects Using Cover Crops

The idea of developing a more environmentally friendly approach towards pest management in fruits and vegetables offers several benefits to the grower and is more sustainable. Two hypotheses have been proposed to explain how cover crops reduce herbivore damage (pest outbreaks) in complex, mixed crop communities.

The 'natural enemies' hypothesis proposes that more diverse food sources (nectar, pollen, prey host species) allow for the establishment of higher densities of predators, parasitoids and parasites, which regulate pest populations in diverse habitats. Alternatively, the 'resource concentration' hypothesis predicts that specialist herbivores will choose to remain in pure stands (monocultures) as opposed to more diverse plant habitats (Root 1973). As a result, there is decreased pest pressure in complex plant communities. Cover crops, which are an integral part of sustainable organic agriculture, provide an excellent potential alternative for managing economically important insect pests (Lu et al. 2000, Treadwell 2007, Creamer and Baldwin 2000).

Cover crops can be annual, biennial or perennial herbaceous plants grown singly or in mixed species stands throughout the year (Sullivan 2003). In temperate climates cover crops are usually winter annuals that are planted in late summer to give soil cover in the winter (Lu et al. 2000, Sullivan 2003). Barley, ryegrass and rye, for example, are excellent winter grass cover crops because of their hardiness (Lu et al. 2000). Legume cover crops such as hairy vetch, clovers, medics and field peas are also used. They all have high nitrogen content in addition to being hardy (Lu et al. 2000, Sullivan 2003).

Summer cover crops are warm-season species that are used to improve soil conditions and prepare the land for a subsequent crop. Leguminous summer cover crops include cowpeas, VB, guar, crotalaria, soybeans, sesbania, and annual sweetclover, whereas graminaceous summer cover crops include sorghum-sudangrass, millet and forage sorghum (Creamer and Baldwin 2000, Sullivan 2003).

Cover crops have a variety of roles including suppressing weeds, insects and nematode populations, holding soil particles together, providing nitrogen for subsequent crops, reducing erosion runoff and pollution of surface water, removing excess salt, preventing leaching of nitrates and influencing insect life cycles (Creamer 1999, Creamer and Baldwin 2000, Hanna et al. 1995, Lu et al. 2000). Cover crops also influence insect population dynamics. In orchards, well managed cover crops divert generalist pests, confuse specialist pests, reduce pest success by altering host plant nutrition, changing the microclimate and by increasing natural enemy abundance (Bugg and Waddington 1994).

Much interest has also been shown in combining leguminous and graminaceous cover crops in vegetable agroecosystems possibly because of the enhanced protection that can be gained from attracting a greater diversity of beneficial insects due to increased plant diversity (Creamer 1999). Mixing of leguminous and graminaceous cover crops can also combine the benefits only pure strands could provide (Creamer 1999).

Growers will not utilize cover crops if they are not economically feasible. Inputs costs such as fertilizer applications, use of plastics and reduced pesticides should amount to practical savings when compared with conventional systems (Lu et al. 2000). The cover crops that I focused on in my research included two legume speices: SH (*Crotalaria juncea*) and VB (*Mucuna pruriens*), and grass species: PM (*Pennisetum glaucum*) and sorghum-sudangrass (Piper) Stapf.

Leguminous Cover Crops

Sunn hemp [SH] (*Crotalaria juncea* L.): This plant is an herbaceous tropical legume that is tolerant to drought and grows well in high and low pH soils (Ragsdale 2006, Balkcom and Reeves 2005). It is fast growing (60 to 80 days) and can grow to approximately 3 m in height; thereby, acting as a good weed suppressant (Ragsdale 2006, Rich et al. 2003). It remains succulent for about 8 weeks after being planted and becomes more fibrous as it matures (Creamer 1999). Sunn hemp produces high biomass yields (2.4 to 3.4 tons of dry mass per ha) and nitrogen (381 to 504 kg per ha) for subsequent crops before the first frost period (Creamer 1999, Balkcom and Reeves 2005). High nitrogen fixation coupled with resistance against southern root-knot and sting nematodes makes it an excellent green manure and cover crop (Rich et al. 2003, Wang and McSorley 2004). However, SH seeds may require inoculation with the appropriate *Rhizobium* bacteria for nitrogen fixation. And, although seed prices are fairly high ($3.30 to $8.80 per kilogram / $1.50 to $4.00 per pound), high seeding rates produce more succulent stems (Ragsdale 2006, Creamer 1999). With these limitations, SH is still considered a very good cover crop for use in North-Central Florida because very few insect or diseases affect its establishment or production (Ragsdale 2006, Rich et al. 2003).

Velvet bean [VB] (*Mucuna pruriens* L.): This plant is native to Asia and is an annual tropical legume that has spread throughout the US as a grazing and green manure cover crop (Rich et al. 2003). It grows vigorously in droughty, sandy and infertile soils and requires a long frost-free growing season (100 to 250 days to reach maturity) (Rich et al. 2003, Creamer 1999).

The stem reaches up to approximately 2.8 m in vine cultivars, the pods are hairy and the leaves are trifoliate with large, ovate leaflets (Creamer 1999). This cover crop adds a considerable amount of nitrogen to the soil and is known to be resistant and antagonistic towards nematodes (Rich et al. 2003). It also produces high biomass yield (2 to 2.8 tons per ha); thereby, supplying the subsequent cash crop with 570 to 790 kilograms of fixed nitrogen (Ragsdale 2006). Although the VB caterpillar, *Anticarsia gemmatalis* (Hübner), is a major pest of this cover crop, it tends to be generally free of diseases and other insect pests (Rich et al. 2003).

Graminaceous Cover Crops

Pearl millet [PM] (*Pennisetum glaucum* (L.) R. Br.): This grass is the most important millet for food and feed on the world market accounting for > 55% of global millet production (ICRISAT). This cover crop is a fast growing, high-yielding, high-quality grass that can be widely grown throughout Florida (Chambliss and Adjei 2006). This warm-season annual is able to grow up to 3.6 m in height, and grows best on well drained soils, but is tolerant of drought and acidic soil conditions (Chambliss and Adjei 2006). Planting usually takes place between April and July and the crop matures in 60 to 70 days (Creamer 1999). Pearl millet also has a fast growing, extensive network of roots that takes advantage of moisture and nutrients. The plant is able to accumulate nitrates during a drought if nitrogen was previously applied before the drought (Chamblis and Adjei 2006). Although PM grows slowly as a seedling, it competes well with late-emerging weeds and has an average productivity of 128 kg per ha (Myers 2002). This grass does not demand a large supply of nutrients to grow well. As a graminaceous cover crop, it is unable to fix nitrogen in the soil, but its nitrogen needs can be obtained from animal manure, a leguminous cover crop, or nitrogen fertilizer (Myers 2002). The only significant disease problem associated with this cover crop is downy mildew caused by *Sclerospora graminicola* (Sacc.) J. Schrot. Chinch bugs and the European corn borer are common insect pests that pose occasional problems for this plant but the only important pest is the European corn borer (ICRISAT, Myers 2002).

Sorghum sudangrass [SSG] (*Sorghum bicolor* x *S. sudanense* (Piper) Stapf): This grass is well known and has been frequently used in Florida for decades (Ragsdale 2006). It is a cross between grain sorghum and sudangrass, and is a warm season cover crop that produces 2 to 2.8 tons of biomass per ha (Ragsdale 2006, Creamer 1999). It is an excellent suppressor of weeds, insects and nematode populations, helps to control erosion, grows rapidly, and has allelopathic properties (Creamer 1999). A high C N ratio is present in large fibrous stems that act to reduce decomposition and prevent leaching (Ragsdale 2006). It does well when planted in mixtures but often attracts armyworms and corn silk flies which can harm vegetable crops (Ragsdale 2006).

CHAPTER 3
POPULATION DYNAMICS OF APHIDS, WHITEFLIES AND THEIR ASSOCIATED NATURAL ENEMIES IN MONOCULTURE AND DICULTURE COVER CROP SYSTEMS

The production of summer squash (*Cucurbita pepo* L.) in Florida had a value of $53 million USD in 2007 (USDA NASS 2008). Over the last decade, the number of small and medium-sized farms involved in organic squash production has significantly increased (Juan Carlos Rodriguez personal communication) due to consumer demands for organic vegetables over conventionally grown produce (Bonina and Cantliffe 2005). Regardless of production methods (conventional or organic) squash production is currently threatened by major economic pests such as aphids and whiteflies. If not effectively controlled, these pests are capable of causing millions of dollars in damage and lost yield (Palumbo et al. 2001, Simmons et al. 2000, McAuslane 2002).

Aphids have extremely high growth and developmental rates between temperatures of 78 and 80° C (Awmack and Leather 2007, Dixon 1977, Webb 2006). Typically, aphids start reproducing 7 to 10 days after egg hatch, and many aphids have telescoping generations, which enables an adult female to have offspring inside her that are simultaneously producing their own daughters by parthenogenesis (Powell et al. 2005, Metcalf and Metcalf 1993, Awmack and Leather 2007). This rapid reproductive behavior can result in severe crop damage if no effective management is employed. Furthermore, some aphids are able to survive in the soil for 48 days at 3° C and eggs are known to survive temperatures lower than - 40° C (Dixon 1985). Such longevity and persistence, increases their importance as economic pests.

Whiteflies are generally multivoltine having 2 to 6 generations per year. They can continue to reproduce as along as temperatures are favorable (Bryne and Bellows 1991). Their life-cycles can be as short as 2 weeks under warm temperatures and the adult female can produce 252 eggs at 28.5° C (Byrne and Bellows 1991, Webb 2006). Both aphids and whiteflies suck sap juices from plants and excrete honeydew, which promotes sooty mold growth and transmits viral diseases, all of which reduce the vigor and marketability of the vegetable.

Generally, aphids and whiteflies are conventionally controlled by several broad-spectrum insecticides from classes including organophosphates, carbamates, formamidines and cyclodienes (Webb 2007, Palumbo et al. 2001, Dewar 2007). Although neonicotinoids are a newer class of insecticides that are effective against aphids and whiteflies, many aphid species have become resistant to other insecticides (Dixon 1985). Some of these synthetic insecticides can enhance the spread of non-persistent viruses especially in cucurbits (Stapleton et al. 2002). Similarly, for whiteflies, all stages of the life cycle are resistant to numerous insecticidal sprays (Peet 1995, Palumbo et al. 2001). These insecticides also threaten non-target organisms including natural enemies, and have a negative effect on the environment by entering waterways via runoff or drift, or depositing on vegetation that may be later contacted or eaten by wildlife (Pimentel et al. 1992).

Organic farms use cultural and biological control techniques to manage key insect pests. Cover crops are an integral component of organic farming and sustainable vegetable culture, and can be used in several ways for insect management practices. For instance, cover crops act as a sink and source when intercropped with a cash crop by attracting and further enhancing the number of natural enemies for eventual release into the cash crop following senescence of the cover crop (Bugg 1991, 1992). Also, in terms of cultural control cover crops are used to reduce the colonization, dispersal and reproduction of pests on vegetable crops. This is accomplished in several ways: 1) acting as a sink for various pests, 2) by visually or olfactorally confusing pests 3) by having nutritional effects on host plants, or 4) by reducing the success of the pest via microclimatic changes (Bugg 1992). Parajulee et al. (1997) demonstrated that the intercropping of cotton with several cover crops enhanced the conservation of cotton aphid predators. Recently, Tillman et al. (2004) also showed that crimson clover (a cover crop used extensively) was responsible for the transfer of big-eyed bug predators onto cotton, but also reported an increase in several ladybird species. Earlier, Bugg et al. (1991) recorded an increase in big-eyed bug predators and successfully transferred these predators from subterranean clover to cantaloupe, which housed sentinel egg masses of the fall armyworm.

Effective organic management strategies for aphid and whitefly pests in cucurbits warrant attention in light of increasing organic production and a general public drive to reduce the use of chemical pesticides. The implementation of cover crops will depend on the grower's primary needs, the structure and condition of the landscape, and the subsequent cash crop (Sarrantonio 1998). A leguminous cover crop will add nitrogen to the soil that can be available for the subsequent cash crop. Alternatively, grass cover crops scavenge for nutrients and take up nitrogen from the soil, but some species produce large amounts of residue that can exceed the amount of organic matter that leguminous cover crops can provide (Bowman et al. 1998). Legume-grass mixtures may be more effective than using a legume or a grass independently. The benefits of legumes and grass mixtures include attracting a greater range of beneficial insects. However, one disadvantage of using mixtures is that they are complicated to manage and must be mixed suitably to respond to soil, weather and pest conditions (Bowman et al. 1998). Additionally care should be taken when selecting the best cover crop because they can harbor both beneficial and harmful arthropods (Bugg 1991). It is therefore important to evaluate different cover crops to determine which one could provide the greatest benefit in terms of pest suppression.

The primary goal of this research was to study the population dynamics of aphids, whiteflies and beneficials in monoculture and mixed cover crop stands in an organic system to assess their likely impact on a subsequent cash crop. My hypothesis was that mixed cover crop cultures would increase the number of beneficial arthropods and potentially suppress aphids and whitefly pests.

Materials and Methods

The experiment was conducted at the University of Florida Plant Science Research and Education Unit in Citra, FL in 2006 and repeated in 2007. The experimental design was a completely randomized block with four replicates of seven treatments. Seven treatments were evaluated, which included two legumes, sunn hemp [SH] (*Crotalaria juncea* L.) and velvet bean [VB] (*Mucuna pruriens* (L.) cv. GA Bush); two grasses, sorghum sudan grass [SSG] (*S. bicolor* x *S. sudanense* cv. Brown-Midrib) and pearl millet [PM] (*Pennisetum glaucum* L. cv. Tifleaf3); two legume-grass mixtures, sunn hemp/pearl millet [SH/PM] and sorghum sudangrass/velvet bean [SSG/VB], and a weedy fallow plot (control). Each treatment plot measured 12 m x 12 m. Each plot was separated by mowed alleys that were 12 m wide, which was intended to reduce the movement of pest and beneficial arthropods between plots. The size of the field measured 7.2 acres, but the total cultivated area was 2.05 acres.

Both legumes, SH and VB, were broadcast at a rate of 40 lbs/ acre (45 kg / ha) and 100 lbs/ acre (112 kg / ha) respectively, while PM and SSG were each planted in rows approximately 18 cm apart at the rate of 25lbs / acre (28 kg / ha) and 40 lbs / acre (45 kg / ha) respectively. The SH/PM mixture was planted using a Sukup 2100 planter at ½ of the recommended seed rate of SH, 20 lbs / acre (22 kg / ha) plus 2/3 the recommended seed rate of PM, 17 lbs / acre (19 kg /ha).

Prior to the beginning of the experiment in 2006, the experimental area was treated with spent mushroom compost (Quincy Farms, 190 Mannic Counn Road) at 10 tons / acre to enhance soil nutrient content. Southern cowpea cover crop, *Vigna unguiculata* L. cv. White Acre, (Alachua Seed and Lumber, Alachua, FL) was incorporated in the soil using a John Deere 6615 tractor (Marion Tractor, Inc., Ocala, FL) at 50 lbs / acre (56 kg / ha) at a depth of 5 cm prior to establishing the cover crop treatments to improve soil and nutrient fertility.

In 2007, 3 weeks prior to cover crop planting, elemental sulfur (Tiger 90) was applied to the entire field at 8lb/plot with a check drop spreader (Newton Crouch Inc., Griffin GA) to reduce the alkalinity of the soil. One application of sulfomag (8.2lbs / plot) was applied to all treatment plots 24 hrs before cover crop planting using a Catch drop spreader.

Assessment of Aphids, Whiteflies and Natural Enemies with Unbaited Yellow Sticky Traps

In 2006 and 2007 aphids, whiteflies and natural enemies (selected parasitoids and predators) were monitored using one unbaited yellow sticky trap placed in the center of each plot (Great Lakes IPM, Vestaburg, MI). Yellow sticky traps were left in the field for 48 hours then brought back to the lab where they were wrapped in clear plastic and stored in a refrigerator until they were ready to be processed. Ground predators and lepidoptera were assessed separately.

Aphids and whiteflies on yellow sticky traps were assessed by using a sub-sampling technique as outlined by Finn (2003) due to the high aphid and whitefly densities. This sub-sampling technique eliminates counting errors and reduces the time spent per trap. Each yellow sticky trap was overlaid with a gridded transparent paper divided into 63, 1- inch squares. Forty-eight of these squares were colored in, while 15 were left transparent (Figure 3-1). The area under these 15 transparent 1- inch squares were examined under a X 40 dissecting microscope and the number of aphids and whiteflies were recorded. Parasitoid numbers were low, therefore, the parasitoids in all 63 squares on the sticky traps were counted and recorded for each unbaited yellow trap.

Assessment of Ground Predators with Pitfall Traps

Carabid (ground) beetles were monitored using locally made pitfall traps. These traps were left in the field for approximately 6 days. A 25% antifreeze solution (Zerex ® G-05, Ashland Inc. Lexington, KY) was poured into the pitfall trap until the solution was just below the wire-meshed holes. The antifreeze solution was responsible for killing the insects and preventing rapid decomposition before the traps were serviced each week. Each week the contents from the intact cup was emptied into a plastic container and taken back to the laboratory for analysis. In 2007 when the experiment was repeated a 5% detergent solution (Colgate Palmolive Co. New York, NY) was used instead of the 25% antifreeze solution as a more environmentally friendly approach.

Assessment of Lepidopteran Pests

In 2006 and 2007, cover crops were sampled weekly for lepidopteran larvae, and levels of parasitism. Two sampling techniques were used to sample for lepidopterans. They included:

- Drop-cloth method (for lepidopteran larvae)
- Bucket trap method (for lepidopteran adults)

The first sampling technique was the drop-cloth method, which involved placing a 45 cm x 45 cm white cotton fabric on the ground and vigorously shaking 7 randomly selected plants from the center of each treatment plot 5 times. All larvae which fell on the drop cloth were counted, collected and brought back to the laboratory for identification. Dead larvae were placed into 70% ethanol for storage and identification. Recovered live larvae were placed into rearing cups containing synthetic diet (Shaver and Raulston 1971) so that development of hosts or parasites could be completed (Browning et al. 1985). All specimens were placed into an environmental chamber (Percival, Boon, IA) (28° C, photoperiod of 14:10 [L:D] h) and allowed to complete development.

The second sampling technique that was used for capturing adult lepidoptera involved the use of tri-colored (yellow, white and green) bucket traps. Bucket traps were baited with the floral volatiles phenylacetaldehyde and linalool. Vapor-strips were placed at the bottom of the traps to kill the lepidopterans entering the traps. One bucket trap was placed in the middle of each block (totaling 4). Adult lepidoptera were collected weekly by emptying the buckets and bringing them back to the laboratory for identification.

Statistical Analysis

The data for aphids, whiteflies, parasitoids and the whitefly predator, *D. pusillus* (yellow sticky trap counts) were square- root transformed and analyzed using repeated measures analysis (PROC MIXED, SAS Institute 2003) to examine the interaction effect between treatment and time (sampling week). Values for least square means were assessed and those means were compared to determine the effects of the cover crop treatments.

Data from ground arthropods in pitfall traps as well as lepidopteran larvae and adult counts were analyzed using analysis of variance (ANOVA) followed by mean separation using least significant difference (LSD) test (SAS Institute 2003).The data were square root transformed where necessary to meet the assumptions for the ANOVA. Results were considered significant if $P \leq 0.05$.

Results

During 2006, some cover crops were affected by diseases. In particular, PM was infected with the Pyricularia leaf spot disease, SSG had Southern blight disease and SH was infected with Leaf margin necrosis. The monoculture SSG had small patchy sections only at the edges of the field compared with other treatments and about 40% of the VB plots contained weeds. In 2007, the horizontal and vertical alleyways (between cover crop treatments) were covered with weeds during the first few days of cover crop establishment. Following the removal of the weeds from the alleyways, approximately 60% of the VB treatment plot still contained weeds. Additionally, plants in the VB plots showed yellowing leaves and were fairly dry. Generally, the overall growth of the cover crop treatments during 2007 was good.

There were several groups of natural enemies that were collected from the sticky traps, which included various parasitic diptera, aphidophagous syrphid flies (Diptera: Syrphidae) such *Toxomerus marginatus* (Say) and *T. geminates* (Say), various coccinellid species that included the whitefly predator, *Delphastus pusillus* (LeConte), the convergent ladybird beetle, *Hippodamia convergens* Guérin-Méneville, the seven spot ladybird beetle, *Coccinella septempunctata* L. and the multicolored Asian ladybird beetle, *Harmonia axyridis* Pallas.There was no significant difference in natural enemy numbers between treatments with the exception the coccinellids, which will be discussed in more detail further on.

There were also several groups of pest species collected via sticky trap sampling. These included aphids, whiteflies, leafhoppers and plant hoppers. The aphid species caught depended on the type of cover crop treatment in the field. The legumes, SH and VB mainly harbored the black legume aphid, *Aphis craccivora* Koch, the green citrus aphid, *Aphis spiraecola* Patch, the melon aphid, *Aphis gossypii* Glover and the green peach aphid *Myzus persicae* (Sulzer), whereas the grasses, PM and SSG, harbored the corn leaf aphid, *Rhopalosiphum maidis* (Fitch), the yellow sugar cane aphid, *Sipha flava* (Forbes), and *Hysteroneura setariae* (Thomas). Both sets of mixtures, SH/PM and SSG/VB, harbored aphids that were common to grasses and legumes. The weedy fallow (control) plots also had aphid species from both grasses and legumes.

Bemisia argentifolii (Bellows and Perring), the silverleaf whitefly was the main species found in the treatment plots. The only natural enemies of whiteflies that were observed included *Delphastus pusillus* LeConte, which was present in both 2006 and 2007.

Population of Aphids and Whiteflies within Cover Crop Treatments (Yellow Sticky Traps)

Aphids 2006. During 2006, the SH had a 90% decline in adult aphid populations over the 3 week sampling period when cover crops were in the field. There was no significant difference in the number of aphids in the SH compared with PM or the corresponding SH/PM mixture (Table 3-1). However, all three cover crop treatments did have significantly fewer aphids than the weedy fallow control on the 3rd (final) week of sampling (F = 2.93; df = 3, 6; P = 0.0357). Overall, there was no interaction between treatment and time. Alternatively, aphid populations in SSG and the SSG /VB mixture were consistently low (< 1 aphid per plot) throughout the sampling period. Only at the end of the sampling period did SSG have significantly fewer aphids than the weedy fallow control and the VB (F = 2.93; df = 3, 6; P = 0.0357). Overall there was no interaction between treatment and time (Table 3-1).

2007. During 2007, SH had significantly more aphids than SH/PM mixture and the weedy fallow control (F = 4.02; df = 3, 6; P = 0.01) during the first week of sampling (Table 3-2). Similarly, VB had significantly more aphids the weedy fallow control (F = 4.02; df = 3, 6; P = 0.01). However, at the end of the season, there were no significant differences in aphid populations among the treatments (Table 3-2). Fluctuations in the number of aphids per week was due to the significant time effect (F =7.11; df = 2, 6; P =0.0262).

There was also a significant interaction between cover crop and time (sampling period) (F = 3.64; df = 12, 54; P = 0.0005). There were only treatment differences in the 1st of the 3 week sampling.

Whiteflies 2006. During the first week of sampling, the SH treatment had significantly higher populations of whiteflies (~ 290 per plot) compared with SH/ PM mixture (~ 48 whiteflies per plot) or single stand PM (~ 12 whiteflies per plot) (F = 2.69; df = 3, 6; P = 0.048) but was not significantly different from the weedy fallow control (Table 3-3). Although whitefly populations in SH declined ~55% by the end of the sampling period, whitefly populations was still significantly higher than PM or the SH/PM mixture (F = 5.44; df = 3, 6; P = 0.0023). Both PM and the SH/PM mixture had significantly fewer whiteflies throughout the sampling period compared to the weedy fallow control.

The VB treatment had a significantly higher number of whiteflies than SSG in the 1st (F = 2.69; df = 3, 6; P = 0.0480), 2nd (F = 7.14; df = 3, 6; P < 0.0001) and 3rd week (F = 5.44; df = 3, 6; P = 0.0023), but was not significantly different from the weedy fallow control in both of those weeks (Table 3-3). At the end of the sampling period, although there was a 60% decrease in the number of whiteflies in VB, this plot still had 4 times as many whiteflies when compared to SSG or the SSG/VB mixture. There was no significant difference between SSG and its corresponding mixture. However, both plots had significantly fewer whiteflies than the control. There was also a significant time effect for the treatments (F = 10.94; df = 2, 6; P = 0.01). There was a significant decline in whitefly numbers in the PM treatment between the 1st and 3rd week of sampling. Also, between the 2nd and 3rd week of sampling there was a decline in the number of whiteflies in the VB and the SSG/VB mixture. There was no significant interaction between treatment and time for the 2006 field-season.

2007. During the 2007 field-season, SSG and the SH/PM mixture consistently maintained a low population of whiteflies throughout the season (Table 3-4). Significantly fewer whiteflies in the 1st week (F = 4.62; df = 3, 6; P = 0.0052) and 3rd week (F = 4.65; df = 3, 6; P =

0.0051), were observed in both monoculture grasses (PM and SSG) and the legume-grass mixtures (SH/PM and SSG/VB) compared with the weedy fallow control (Table 3-4). There was a significant interaction between treatment and time (sampling period) (F = 2.80; df = 12, 54; P = 0.0048) . Treatment differences were observed in the 1st and 3rd week of sampling.

Parasitoids 2006. The majority of the natural enemies identified within the cover crop treatments were parasitoids. These included: Aphelinidae: *Aphelinus* sp., Braconidae: *Aphidius* sp., *Chelonus* sp., and *Lysiphlebus testaceipes* (Cresson), Icheumonidae, Bethylidae, Sclelionidae: *Telenomus* sp., Eucoilidae, Mymaridae, Chalcididae, Eulophidae: Tetrastichinae, Trichogrammatidae, and Encyrtidae: *Metaphycus* sp. Many of the braconids were aphid parasitoids. The family Aphelinidae may have included whitefly parasitoids, but no whitefly parastioids were specifically identified.

During 2006, in the first week, SH and PM monocultures had significantly fewer parasitoids compared with the weedy fallow control (F = 2.68; df = 3, 6; P = 0.0491) [Table 3-5], but the SH/PM mixture had high numbers of parasitoids that were not significantly different compared with the weedy fallow (F = 2.68; df = 3, 6; P = 0.0491). Overall, there was an 82% increase in the number of parasitoids in PM over the 3 week sampling period. Alternatively, there was a 67% decrease in parasitoid numbers in the weedy fallow control over the same sampling time (Table 3-5).

Similarly, the SSG/VB mixture had significantly higher numbers of parasitoids than VB (F = 2.68; df = 3, 6; P = 0.0491) [Table 3-5] but was not significantly different from SSG or the weedy fallow control. Overall, there was a significant interaction between treatment and time (F = 2.20; df = 12, 54; P = 0.0245). Treatment differences were found in the 1st of the 3 sampling periods.

2007. In 2007, SH had significantly higher numbers of parasitoids in the 2nd week compared with all other treatments (F = 3.21; df = 3, 6; P = 0.0480) [Table 3-6]. There was no significant time effect or interaction between treatment and time.

Coccinellids (*Delphastus pusillus*) 2006. During the first week of sampling, SSG and the SSG /VB mixture had significantly more *D. pusillus* all other treatments except PM (F = 2.96, df = 3, 6; P = 0.0346) [Table 3-7]. By the end of the sampling period, SSG and the SSG/VB mixture, had significantly more coccinellids (F = 8.97; df = 3, 6; P < 0.0001) [Table 3-7] than most other treatments; however, PM was not significantly different than the SSG/VB treatment.

There was a significant interaction effect between treatments and time (F = 2.20; df = 12, 54; P = 0.0246).

2007. In the 1st and 2nd weed of sampling there were no significant differences among the treatments. However in the 3rd week of sampling SSG had significantly higher number of *D. pusillus* compared with all other treatments (F = 2.79; df = 3, 6; P = 0.0425) [Table 3-8]. The number of whitefly predators in 2007 was 10 times lower than in 2006. There was also a significant (F = 1.98; df = 12, 54; P = 0.0443) interaction between treatment and sampling period. Treatment differences were observed in 3rd week of sampling.

Assessment of Ground Arthropods from Pitfall Traps

Data from 2006 and 2007 field-seasons showed that Formicidae comprised the majority (~ 90%) of the arthropods that were collected from the pitfall traps (Figure 3-2a). Overall, there was a larger diversity of predators collected in 2006 (~ 8 families) compared with ~ 5 families in 2007 (Figure 3-2). Scarabs were collected in 2006, but were not found in 2007 (Figure 3-2b).

During 2006 when the pitfall trap data were pooled across sampling dates and examined according to each treatment, VB treatment had the highest numbers of predatory arthropods compared with all other treatments except PM (F = 6.74; df = 3, 6; P = 0.0007) [Table 3-9]. In

2007, the weedy fallow control had the highest number of predators compared with the other treatments except the two legume-grass mixtures, SH/PM and SSG/VB (F = 9.32; df = 3, 6; P < 0.0001) [Table 3-9].

Assessment of Lepidopteran Pests

During 2006 and 2007, the adult lepidopteran populations, which were monitored by block using floral volatile baited bucket traps produced six common species that include: VB caterpillar, *Anticarisa gemmatalis* (Hübner), *Mocis* spp., the bella moth, *Utetheisa ornatrix* L., the fall armyworm, *S. frugiperda* (J. E. Smith), the beet armyworm, *S. exigua* (Hübner), and the southern armyworm, *S. eridania* (Stoll). The population of VB moth adults was significantly higher than that of all other moth species collected via bucket traps in 2006 (F = 6.78; df = 5, 3; P = 0.0017) and 2007 (F = 2.87; df = 5, 3; P = 0.0519) (Table 3-10). Other species that were found periodically included: the corn earworm *Helicoverpa zea* (Boddie), the soybean looper, *Pseudoplusia includens* (Walker), the melonworm *Diaphania hyalinata* L., the sharp-stigma looper, *Ctenoplusia oxygramma* (Geyer), the golden looper *Argyrogramma verruca* (F.), and other miscellaneous noctuid species. There was only one incidence of an armyworm larva being parasitized by a tachniid fly.

The majority of the lepidopteran larvae collected from drop cloth samples by treatment were VB caterpillars. In 2006, a total of 47 lepidopteran larvae were collected from drop cloths over the 3 week period. Of these, 31 survived and were successfully reared to adults in the laboratory, all of which were VB caterpillars. There were significantly more VB caterpillars in the SH and VB monoculture compared with the other treatments (F = 8.04; df = 3, 6; P = 0.0003) (Table 3-11).

In 2007 a total of 21 lepidopteran larvae were collected from the drop cloth method. Of these, 20 were VB moths and one was a fall armyworm. There were significantly more VB larvae collected from SH compared with the other treatments ($F = 5.52$; df = 3, 6; $P = 0.0021$) (Table 3-11).

Discussion

During the 2006 and 2007 field-seasons, SSG had significantly fewer aphids and whiteflies compared with the control and other cover crop treatments. This grass monoculture has the best potential to prevent the buildup of key pests (aphids and whiteflies) in an organic field and consequently could diminish the number of pest problems that could become established on subsequent cash crops. My results were contrary to Bugg and Ellis (1990) who reported that sorghum grass, *Sorghum bicolor* L., harbored high numbers of aphids and coccinellids. The hybridization of sorghum and sudangrass could perhaps have resulted in increased resistance to aphids as well as other pests. Valenzuela and Smith (2002) found cultivars of SSG hybrids, which have resistance to diseases, insects and nematodes. Also, SSG hybrids have been reported to possess allelopathic properties that can suppress the growth of weeds (Valenzuela and Smith 2002), thereby potentially reducing the number of available hosts for aphids and other pests.

Low parasitoid populations in 2006 corresponded to higher aphid numbers (~ 2.6 aphids) per plot, whereas higher parasitoid populations in 2007 corresponded to fewer aphids numbers (~ 0.8 aphids) per plot. *Aphidius* spp. and *L. testaceipes* are the two main aphidiine parasitoids that were captured. These parasitoids have high fecundities (~ 300 to 1800 eggs per female) [Völkl et al. 2007] and take 14 to 21 days depending on temperature to complete their life cycle (Ferguson 2005). Both parasitoid species have a wide range of aphid hosts from several species (Rodrigues and Bueno 2001). Many of these host aphids were captured with the yellow sticky traps. Therefore, these parasitoids may have had some effect in regulating aphid populations.

With regards to predators, during 2006, adult syrphid flies were captured with yellow sticky traps, but these numbers were extremely low, and there were no differences among any of the treatments. During the 2007 field-season, blue pan traps were placed in the cover crop plots to enhance the capture of syrphid flies that were observed in the previous year, but none were captured. Therefore, aphidophagous syrphid flies were believed to play a minor role in regulating aphid populations.

There were no specific whitefly parasitoids that were recorded from the field, therefore, I assumed that the differences in whitefly numbers among treatments were probably due to coccinellids. In 2006 and 2007, SSG had between 2 to 47 times and 2 to 16 times more whitefly predators respectively, compared with other treatments. *Delphastus pusillus* was the principal predator found in the SSG treatment. It is a voracious predator of whiteflies. Hoelmer et al. (1993) reported that *D. pusillus* fed on all whitefly stages and consumed almost 1000 whitefly eggs before it pupating.

Other cover crop treatments were also able to suppress aphid and whitefly populations. For instance, SH and PM also had lower aphid populations than with the control. However, PM may have a slight advantage over SH in terms of its applicability because this grass treatment had higher numbers of parasitoids and ground arthropods (compared with SH). In 2007, the addition of sulfur to the soil may have reduced the number of soil arthropods in the PM, as well as other treatments. Cárcamo et al. (1998) has shown that sulfur contamination can negatively affect soil macroinvertebrates such as carabid beetles, staphilinids and spiders, most of which were found in the pitfall traps in 2007. Pearl millet may also be a good choice for suppressing whitefly numbers possibly due to the high number of predators and parasitoids that this cover crop harbored. Both VB and the weedy fallow control had significantly higher whitefly numbers than any other cover crop treatments. Although VB is able to fortify the soil with nitrogen for subsequent cash crop, the high numbers of whiteflies found in these treatments strongly suggests that it is not a good cover crop to establish before planting cucurbits, which is susceptible to whiteflies.

In 2006, both dicultures, SH/PM and SSG/VB had significantly fewer aphids than the weedy fallow control, however in 2007, they were significantly different to the control, which had high aphid populations. However, the SSG/VB diculture was able to maintain significantly fewer whiteflies than the SH/PM mixture in both years. Throughout both field seasons, there was no significant difference between the SSG/VB diculture and the SSG monoculture. However, the diculture always had significantly fewer whiteflies than the VB monoculture. Mixed cultures had more predaceous ground arthropods including carabid beetles, scarabid beetles, dermapterans and ants (based on pitfall trap counts). The greater number of beneficials recovered from the diculture plots supports the natural enemies hypothesis by Root (1973) who proposed that higher plant diversity leads to an increase in the number of natural enemies and potentially more regulation in pest population. In 2007, the weedy fallow had the highest number of ground arthropods because it was the most diverse in terms of the number of different species of weeds present.

Several of the carabid species found in the pitfalls were *Scarites* spp. Carabid beetles are known to feed on agricultural pests such as aphids (Kromp 1999, Mundy et al. 2000). They feed on aphids early in the season when their densities are still low and consume aphids that have fallen off the plants unto the ground (Kromp 1999). Laub and Luna (1992) concluded that cover crop mulches provided a suitable habitat for carabid predators including *Pterostichus* spp. and *Scarites* spp.

Velvet bean (VB) moths and larvae were the most abundant Lepidoptera recovered from bucket traps, and reared from larval samples in both 2006 and 2007. This insect is a serious pest of legumes and grasses (Gregory et al. 1998). The floral lures used in this study can attract for a relatively long distance (bigger than the 12 x 12 m plots). Also, these lures were chosen because previous research (Meagher and Landolt 2008) showed that VB caterpillars were attracted to these lures.

Although the SH and VB treatments harbored many VB caterpillars, they also harbored many parasitoids including Eulophidae and Braconidae or egg parasitoids including Trichogrammatidae, which are known for suppressing lepidopteran pests and providing some control. Demapterans and various species of ants are also predators of the VB caterpillar (Barbara 2000). Ants were the largest group of ground arthropods recovered from the pitfall traps in both 2006 and 2007. The diversity of the pitfall trap captures further supports the fact that there was great potential for the adequate suppression of the pestiferous lepidopteran in the plots.

Overall, the results from this study have shown that the monoculture grasses, specifically SSG, have the best potential for hosting lower populations of aphids and whiteflies in an organic system. Some of these monoculture grasses hold potential to be used as cover crops when subsequent cash crops that are susceptible to aphids and whiteflies. The SSG/VB diculture is also worthy of mention because it had the best potential for enhancing the natural enemy population. These results can help to develop sustainable pest management practices for organic growers, but also shows exceptional promise for the development of future organic pest management programs.

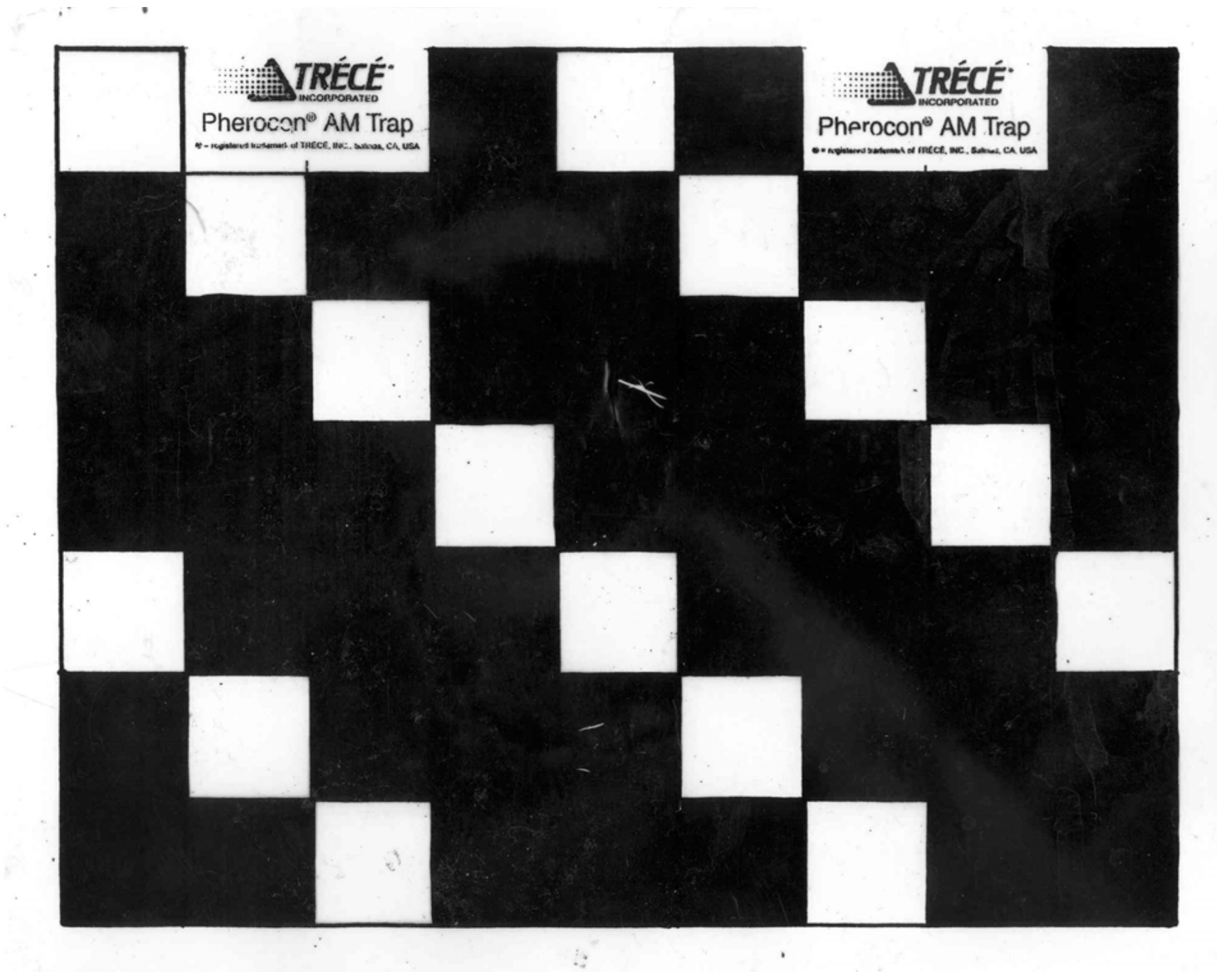


Figure 3-1. Gridded transparency used to count aphids and whiteflies on unbaited yellow sticky traps.

Table 3-1. Aphid population dynamics in monoculture and diculture treatments in 2006

Treatment	Mean ± SEM aphid numbers in 2006		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	3.50 ± 2.87	0.00 ± 0.00	0.25 ± 0.25c
PM	0.00 ± 0.00	0.25 ± 0.25	0.00 ± 0.00c
SH + PM	0.25 ± 0.25	0.25 ± 0.25	0.00 ± 0.00c
SSG	0.00 ± 0.00	0.25 ± 0.25	0.00 ± 0.00c
VB	4.00 ± 3.67	1.50 ± 1.19	1.75 ± 0.75ab
SSG + VB	0.25 ± 0.25	0.00 ± 0.00	0.50 ± 0.28bc
Weedy fallow (Control)	3.00 ± 1.91	0.75 ± 0.75	2.00 ± 1.08a

Aphid data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 0.81$; df = 3, 6; $P = 0.5725$

[b] $F = 1.09$; df = 3, 6; $P = 0.4048$

[c] $F = 2.93$; df = 3, 6; $P = 0.0357$

Table 3-2 Aphid population dynamics in monoculture and diculture treatments in 2007

Treatment	Mean ± SEM aphid numbers in 2007		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	5.75 ± 1.25a	0.00 ± 0.00	0.00 ± 0.00
PM	3.00 ± 1.58abc	0.25 ± 0.25	0.00 ± 0.00
SH + PM	0.75 ± 0.47cd	0.00 ± 0.00	1.75 ± 1.75
SSG	0.50 ± 0.50cd	1.00 ± 0.40	0.75 ± 0.75
VB	4.25 ± 1.87ab	0.25 ± 0.25	1.75 ± 0.85
SSG + VB	1.25 ± 1.25bcd	0.25 ± 0.25	1.25 ± 0.47
Weedy fallow (Control)	0.00 ± 0.00d	0.00 ± 0.00	4.00 ± 1.77

Aphid data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 4.02$; df = 3, 6; $P = 0.0100$

[b] $F = 2.33$; df = 3, 6; $P = 0.0764$

[c] $F = 1.59$; df = 3, 6; $P = 0.2072$

Table 3-3. Whitefly population dynamics in monoculture and diculture treatments in 2006

Treatment	Mean ± SEM whitefly numbers in 2006		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	289.50 ± 28.23a	76.50 ± 10.25a	115.75 ± 36.79a
PM	11.75 ± 6.34c	1.25 ± 0.94c	1.25 ± 1.25d
SH + PM	47.75 ± 32.17c	4.75 ± 2.17bc	2.25 ± 1.60cd
SSG	81.25 ± 72.91c	3.00 ± 2.04bc	3.50 ± 2.59cd
VB	242.25 ± 78.98ab	57.25 ± 13.60a	56.25 ± 11.07b
SSG + VB	95.25 ± 30.26bc	19.25 ± 14.07b	20.50 ± 6.43c
Weedy fallow (Control)	169.25 ± 128.02abc	67.00 ± 10.34a	68.00 ± 36.14ab

Whitefly data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 2.69$; df = 3, 6; $P = 0.0480$

[b] $F = 17.99$; df = 3, 6; $P < 0.001$

[c] $F = 5.44$; df = 3, 6; $P = 0.0023$

Table 3-4. Whitefly population dynamics in monoculture and diculture treatments in 2007

Treatment	Mean ± SEM whitefly numbers in 2007		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	57.00 ± 18.81ab	4.25 ± 1.43	47.75 ± 13.74a
PM	2.50 ± 1.89c	2.00 ± 0.57	1.25 ± 1.25d
SH + PM	5.50 ± 2.61c	2.25 ± 0.75	5.25 ± 2.78dc
SSG	2.00 ± 1.41c	1.50 ± 0.64	3.25 ± 1.37dc
VB	11.50 ± 2.95bc	1.00 ± 1.00	15.50 ± 6.91bc
SSG + VB	8.50 ± 3.22bc	0.50 ± 0.28	3.00 ± 1.47dc
Weedy fallow (Control)	158.00 ± 106.67a	8.25 ± 7.26	26.50 ± 11.98ab

Whitefly data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 4.62$; df = 3, 6; $P = 0.0052$

[b] $F = 0.91$; df = 3, 6; $P = 0.5117$

[c] $F = 4.65$; df = 3, 6; $P = 0.0051$

Table 3-5. Parasitoid population dynamics in monoculture and diculture treatments in 2006

Treatment	Mean ± SEM parasitoids in 2006		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	1.50 ± 1.19bc	0.75 ± 0.47b	1.25 ± 0.25
PM	1.00 ± 0.71bc	2.00 ± 1.22ab	5.50 ± 2.66
SH + PM	5.00 ± 1.58ab	4.50 ± 1.19a	2.75 ± 1.03
SSG	3.00 ± 0.91abc	0.75 ± 0.47b	1.25 ± 0.75
VB	0.25 ± 0.25c	2.00 ± 0.71ab	3.00 ± 0.57
SSG + VB	5.00 ± 1.47ab	1.75 ± 0.85ab	2.50 ± 1.55
Weedy fallow (Control)	10.75 ± 5.34a	1.25 ± 0.62b	3.50 ± 0.86

Parasitoid data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).
[a] $F = 2.68$; df = 3, 6; $P = 0.0491$
[b] $F = 2.19$; df = 3, 6; $P = 0.0922$
[c] $F = 1.35$; df = 3, 6; $P = 0.2861$

Table 3-6. Parasitoid population dynamics in monoculture and diculture treatments in 2007

Treatment	Mean ± SEM parasitoids in 2007		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	13.25 ± 4.36	10.75 ± 3.35a	25.75 ± 12.75
PM	4.75 ± 1.03	2.75 ± 0.47b	6.00 ± 2.34
SH + PM	6.25 ± 2.17	2.75 ± 0.85b	11.75 ± 3.66
SSG	6.75 ± 1.49	2.50 ± 1.19b	8.25 ± 1.54
VB	11.00 ± 5.14	5.00 ± 1.58b	13.25 ± 6.94
SSG + VB	7.50 ± 2.02	4.25 ± 0.63b	13.75 ± 6.00
Weedy fallow (Control)	3.50 ± 0.64	3.00 ± 1.47b	7.25 ± 2.32

Parasitoid data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 1.79$; df = 3, 6; $P = 0.1580$

[b] $F = 4.25$; df = 3, 6; $P = 0.0077$

[c] $F = 1.87$; df = 3, 6; $P = 0.1465$

Table 3-7. *Delphastus pusillus* population dynamics in monoculture and diculture treatments in 2006

Treatment	Mean ± SEM whitefly predator numbers in 2006		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	0.75 ± 0.25b	0.25 ± 0.25c	0.00 ± 0.00c
PM	2.75 ± 1.75ab	2.25 ± 0.63c	2.75 ± 1.18bc
SH + PM	1.25 ± 0.63b	1.00 ± 0.40c	0.25 ± 0.25c
SSG	6.25 ± 2.42a	21.75 ± 4.95a	19.00 ± 4.91a
VB	0.25 ± 0.25b	0.50 ± 0.28c	0.50 ± 0.28c
SSG + VB	6.00 ± 2.27a	12.75 ± 5.00b	9.25 ± 0.25b
Weedy fallow (Control)	0.50 ± 0.29b	0.75 ± 0.47c	1.25 ± 0.75c

Means for *Delphastus pusillus* data are untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 2.96$; df = 3, 6; $P = 0.0346$

[b] $F = 11.01$; df = 3, 6; $P < 0.0001$

[c] F = 8.97; df = 3, 6; P = 0.0001

Table 3-8. *Delphastus pusillus* population dynamics in monoculture and diculture treatments in 2007

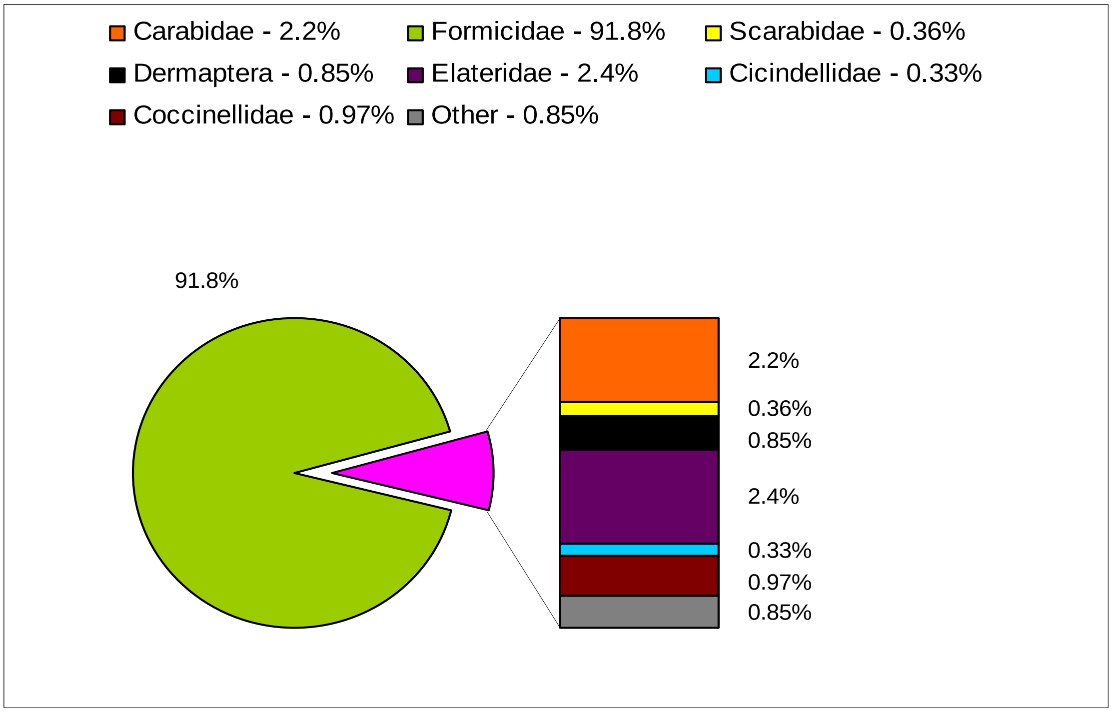
Treatment	Mean ± SEM whitefly predator numbers in 2007		
	Week 1[a]	Week 2[b]	Week 3[c]
SH	0.00 ± 0.00	0.25 ± 0.25	0.00 ± 0.00b
PM	0.50 ± 0.28	0.25 ± 0.25	0.25± 0.25b
SH + PM	0.00 ± 0.00	0.00 ± 0.00	1.25 ± 1.25b
SSG	1.25 ± 0.47	0.00 ± 0.00	2.75 ± 0.47a
VB	0.25 ± 0.25	0.00 ± 0.00	0.00 ± 0.00b
SSG + VB	1.00 ± 0.71	0.00 ± 0.00	0.75 ± 0.47b
Weedy fallow (Control)	0.50 ± 0.29	0.25 ± 0.25	0.50 ± 0.28b

Delphastus pusillus data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 1.53$; df = 3, 6; $P = 0.2248$

[b] $F = 0.69$; df = 3, 6; $P = 0.6589$

[c] F = 2.79; df = 3, 6; P = 0.0425



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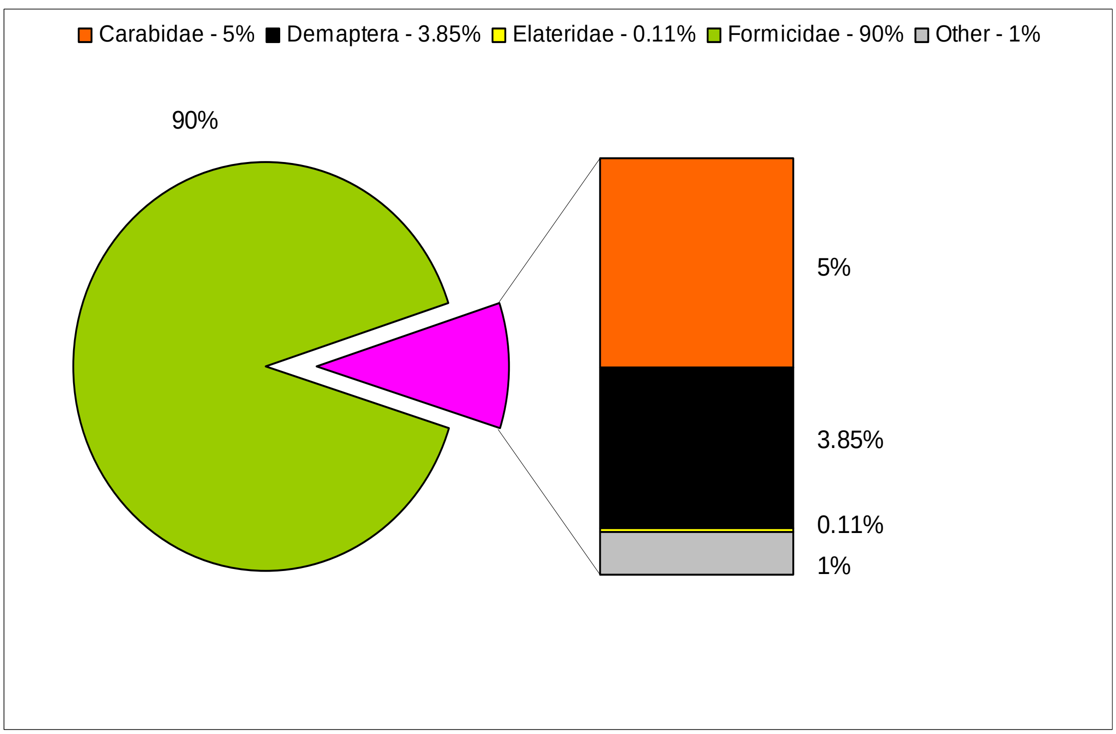


B.

Figure 3-2. Percentage of total natural enemy population from pitfall traps averaged across all cover crops.

Table 3-9. Mean number of ground arthropods captured from pitfall traps in 2006 and 2007

Treatment	2006[a]	2007[b]
SH	2.00 ± 0.71d	4.00 ± 0.71cd
PM	13.00 ± 1.77ab	1.75 ± 0.75d
SH + PM	4.75 ± 2.25cd	23.00 ± 3.18ab
SSG	3.75 ± 1.54cd	7.25 ± 1.31c
VB	21.00 ± 4.30a	16.50 ± 2.32b
SSG + VB	8.25 ± 3.47bc	21.5 ± 5.95ab
Weedy fallow (control)	7.00 ± 1.47bcd	28.00 ± 6.16a

Ground arthropod data were square root transformed before analysis, but the means shown represent untransformed values. Also, means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 6.74$; df = 3, 6; $P = 0.0007$

[b] $F = 9.32$; df = 3, 6; $P = 0.0001$

Table 3-10. Mean number of adult lepidoptera captured from floral volatile baited bucket traps in 2006 and 2007

Moth species	2006[a]	2007[b]
Anticarsia gemmatalis (VB moth)	36.00 ± 13.54a	5.00 ± 1.77a
Mocis sp.	8.00 ± 3.34b	1.25 ±0.94b
Spodoptera frugiperda (Fall armyworm)	0.25 ± 0.25b	0.00 ± 0.00b
Spodoptera eridania (Southern armyworm)	1.00 ± 0.71b	0.50 ± 0.50b
Spodoptera exigua (Beet armyworm)	1.25 ± 0.94b	1.50 ± 1.50b
Utetheisa ornatrix (Bella moth)	0.25 ± 0.25b	0.50 ± 0.50b

Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 6.78$; df = 5, 3; $P = 0.0017$
[b] $F = 2.87$; df = 5, 3; $P = 0.0519$

Table 3-11. Mean number of lepidopteran larvae recorded from drop cloths in 2006 and 2007

Treatment	2006[a]	2007[b]
SH	4.25 ± 1.10a	8.00 ± 2.97a
PM	0.25 ± 0.25b	1.00 ± 1.00b
SH + PM	0.00 ± 0.00b	2.00 ± 0.41b
SSG	1.00 ± 0.40b	0.00 ± 0.00b
VB	5.75 ± 1.65a	2.00 ± 1.00b
SSG + VB	0.00 ± 0.00b	0.75 ± 0.47b
Weedy fallow (control)	0.50 ± 0.28b	0.25 ± 0.25b

Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 8.04$; df = 3, 6; $P = 0.0003$
[b] $F = 5.52$; df = 3, 6; $P = 0.0021$

CHAPTER 4
INSECT POPULATION DYNAMICS IN ORGANIC SQUASH FOLLOWING INCORPORATION OF COVER CROPS

The United States is the fifth largest supplier of squash and pumpkins in the world (Economic Research Service 2004). The number of squash-producing farms has increased over the past 30 years and in 2006, the U.S. produced 430,100 metric tons (MT) of squash valued at $229 million USD (Smith 2008). Florida currently ranks as the leading producer of summer squash (*Cucurbita pepo* L.) with a value of $53 million USD (USDA NASS 2008). Many cucurbits including summer squash are severely affected by a wide range of key agricultural pests including aphids and whiteflies.

Many aphid species affect squash plants; some of the most pestiferous being the green peach aphid (*Myzus persicae* Sulzer), the cotton aphid (*Aphis gossypii*, Glover) and the cowpea aphid (*Aphis craccivora* Koch) (Mossler and Nesheim 2001). Aphids cause plant damage by directly feeding on phloem (Pettersson et al. 2007). The injected saliva causes twisting and curling of leaves which ultimately causes gradual wilting, discoloration and death (Mossler and Nesheim 2001). Aphids also transmit 275 viruses to squash plants (Nault 1997) usually in a non-persistent manner. This allows for rapid spread of the virus in a given area (Frank and Liburd 2005).

The silverleaf whitefly, *Bemisia argentifolli* (Bellows and Perring) also feeds on squash plants causing the squash silverleaf (SSL) disorder. This disorder causes dramatic reductions in marketable crop yield (Hooks et al. 1998).

In order to avoid total crop loss, farmers have used various broad-spectrum chemical insecticides to control aphids and whiteflies. The widespread use of these insecticides has led to increased resistance within key pest populations. Additionally, some pesticides may have increased the spread of virus diseases in cucurbits, possibly because of increased aphid movement (Webb 2007). These issues warrant the investigation of sustainable pest control strategies that are environmentally friendly.

Several methods of cultural control involve manipulation of the environment by increasing plant diversity (Wratten et al. 2007). Many studies have focused on intercropping several cover crop species (monocultures and dicultures of legumes and grasses) with a cash crop, and have successfully shown that intercropping can result in a build-up of natural enemies, which in turn are relayed to the cash crop to provide pest control (Parajulee et al. 1997, Tillman et al 2004). However, in some cases the intercropped system did not reduce pest densities (Bugg et al. 1991). In these studies, the key idea was the conservation of natural enemy populations for pest suppression. However, to-date there has been no studies which document the effects of incorporating cover crops into the soil to provide on pest suppression in a subsequent cash crop.

My goal was to determine if the incorporation of cover crops into the soil could lead to pest suppression due to increase in the number of natural enemies or potentially better plant nutrition. In this case it is presumed that the natural enemies will migrate to nearby refugia of secondary hosts until the cash crop is planted. After the cash crop is established, it is hypothesized that the natural enemies will leave the refugia in preference for pests on the cash crop.

Materials and Methods

Field experiments were conducted on organically certified land at the University of Florida's Plant Science Research and Education Unit in Citra, FL. Prior to the beginning of the experiment, the area was treated with spent mushroom compost (Quincy Farms, 190 Mannic Counn Road) at 10 tons / acre to enhance soil nutrient content and amend soil pH. Southern cowpea cover crop, *Vigna unguiculata* L. cv. White Acre, (Alachua Seed and Lumber, Alachua, FL) was planted in each plot at 50 lbs / acre (56 kg / ha) prior to establishing cover crop treatments to improve soil and nutrient fertility, and also to increase nematode populations. Experimental design was a completely randomized block with four replicates of seven treatments. The seven cover crop treatments included a weedy fallow plot which served as the control, two legumes, sunn hemp [SH] (*C. juncea*) and velvet bean [VB] (*M. pruriens var. pruriens*), two grasses, sorghum sudangrass [SSG] (*S. bicolor* x *S. sudanense*) and pearl millet [PM] (*P. glaucum*), and two sets of legume-grass mixtures: 1) sunn hemp and pearl millet combination [SH/PM], and 2) a sorghum sudandgrass and velvet bean combination [SSG/VB]. The legume-grass mixtures were selected because they are suppressive to rootknot nematodes and produce large amounts of biomass for weed suppression.

Each treatment plot measured 12 m x 12 m. Each plot was separated by mowed alleys that were 12 m wide, which was intended to reduce the movement of pest and beneficial arthropods between plots. Both legumes, VB and SH, were broadcast, whereas both grasses, PM and SSG were planted with a Sukup tractor (CHAPTER 3). In 2006, after 8 weeks, the cover crop treatments were flail-mowed and incorporated into 25.4 cm into the soil by disking (New Holland Flail Mower, Purdy Tractor and Equipment Inc., Hillsdale, MI). Yellow squash (Cougar F1 cultivar, Harris seeds) was direct seeded into each treatment plot on October 19, 2006; 3 weeks after cover crop incorporation. However, due to the poor germination of the squash plants, areas with no emergence had to be re-seeded. Re-seeding was done on October 30, 2006. This delayed arthropod sampling in squash for 7 weeks after cover crop incorporation. In 2007, squash plants were direct seeded on October 10, 2007; 7 days after cover crops were incorporated. Insect sampling in squash began 4 weeks after planting.

Key pests and beneficials in squash were sampled using unbaited yellow sticky traps, blue pan traps, *in situ* counts, leaf disc counts and pitfall traps. Adult lepidopteran populations were sampled using bucket traps.

Assessment of Key Pests and Natural Enemies with Unbaited Yellow Sticky Traps

In 2006 and 2007 natural enemies in each squash plot were monitored using one unbaited yellow sticky trap (Great Lakes IPM, Vestaburg, MI) and one blue pan trap (Packer Ware Bowls, Gainesville, FL). The inner two rows of the squash plots were sampled as to eliminate edge effects. Traps were left in the field for 48 hours then brought back to the lab where they were wrapped in clear plastic wrap and stored in a refrigerator until they were ready to be processed. The number of parasitoids on each sticky trap were counted and recorded.

Blue Pan Trap Captures

Blue water pan traps were supported by tomato wire cages (45 cm x 15 cm x 20 cm), filled with water and detergent [5% detergent solution] (Colgate Palmolive Co. New York, NY) to break the surface tension (Webb et al. 1994). One blue pan trap was placed diagonally across from an unbaited yellow sticky trap on the opposite end on the plot to monitor syrphid flies. Traps were left in the field for one week. At the end of the week, the contents from each trap were emptied and into a small plastic container, labeled accordingly and taken back to the lab to be analyzed. The aphids and natural enemies from the blue traps were counted under a 40 X dissecting microscope and the contents placed in 15 x 45 mm vials (Fisherbrand®, Fisher Scientific, Pittsburgh, PA) with 70% ethanol for storage.

Visual and Leaf Disc Counts

Each week visual (*in situ*) counts were conducted in the field to record the number of aphids (alate and apterous), whiteflies, as well as other insects. All insects were visually assessed by randomly examining the leaves of eight squash plants from each of the two inner rows for ~1 minute.

Circular leaf discs were also used to estimate the number of whitefly nymphs and eggs on squash plants (Frank and Liburd 2005). Every two weeks, eight plants were randomly chosen from the two inner rows and one leaf was randomly picked from each of the 8 plants and taken back to the lab. Two circular (2.5 cm diameter) sections were cut from each leaf using a cork borer. Leaf discs were then examined under a 40 X dissecting microscope for whitefly eggs and immatures (Frank and Liburd 2005).

Assessment of Lepidopteran Pests

In 2006 and 2007 field plots were also sampled weekly for adult lepidopterans using tri-colored bucket traps. Bucket traps were baited with floral volatiles, phenylacetaldehyde and linalool and vapor-strips were placed at the bottom of the traps to kill the lepidopterans entering the traps. One bucket trap was placed in the middle of each block (each block had 4 replicates). Adults were collected weekly by emptying the buckets and brought back to the laboratory for identification.

Sentinel Squash Plants and Peripheral Sampling

During the 2007 field-season two potted sentinel squash plants were placed on the edge of each plot 2 hours following cover crop incorporation into the soil to monitor the dispersal of aphids, whiteflies and natural enemies. Sentinel plants were placed in the plots on 3 October 2007 and taken out of the field on 15 October 2007. For each sentinel squash plant, aphids, whiteflies and natural enemies were assessed via *in situ* counts once per week for two weeks. On the third week, the sentinel plants were removed to allow for direct seeding of squash into the treatment plots.

Sentinel squash plants were raised and potted in the Small Fruit and Vegetable greenhouse at the University of Florida and transported to the field on the day of cover crop incorporation. Sentinel squash plants were approximately 30 cm in height when placed in the field. Plants were placed diagonally across from each other (approximately 15 m apart) within in each treatment plot.

In addition to sentinel squash plants, 20 unbaited yellow sticky traps were placed on the periphery of the field to assess residual natural enemy populations from incorporated cover crop treatments. Yellow sticky traps were placed along the periphery of the field on the 3rd, 6th and 9th October 2007. Traps remained in their peripheral location for 48hrs. They were then covered with plastic wrap and brought back to the lab for processing. Traps were approximately 50 m apart. The vegetation bordering the field was divided into three types: 1) fruit trees (orange) 2) woods and 3) grass/weeds.

Squash Silver Leaf Disorder Rating

In 2007, we noticed squash silver leaf (SSL) disorder in the field and recorded the severity of the disorder based on the following scale 0 = no silvering (0% silvering), 3 = moderate silvering (50% silvering), 5 = complete silvering (100% silvering). Four plants from the middle of the inner two rows were sampled and rated based on the above scale. Sampling was done 2 weeks after planting and at the end of the field-season (8 weeks after planting).

Statistical Analysis

Data from aphids, whiteflies and natural enemies from all the sampling techniques along with the SSL disorder rating were square root transformed and analyzed using analysis of variance (ANOVA) PROC GLM (SAS Institute 2003) because there was no interaction between treatment and time. Treatment means were separated using the least significant difference (LSD) separation procedures. Results were considered significant if $P \leq 0.05$. Regression analysis

(PROC REG, SAS Institute 2003) was also carried out to assess the relationship between the number of whitefly immatures and the presence of SSL disorder.

Parasitoid data from peripheral traps were analyzed using repeated measures analysis (PROC MIXED, SAS Institute 2003) to examine interaction effect between the different habitats and time (sampling weeks). Key pest (aphid and whitefly) data from peripheral yellow sticky traps were analyzed using analysis of variance (ANOVA) PROC GLM (SAS Institute 2003) because there was no interaction between habitat and time (sampling weeks).

Results

The aphids that were observed via *in situ* counts on squash plants included the melon aphid, *Aphis gossypii* Glover and the green peach aphid *Myzus persicae* (Sulzer). *Bemisia argentifolii* (Bellows and Perring), the silverleaf whitefly was the main species found in the treatment plots. The natural enemies observed were the convergent ladybird beetle, *Hippodamia convergens* Guérin-Méneville, and the multicolored lady beetle *Harmonia axyridis* (Pallas). Aphid mummies were observed on squash leaves about 3 weeks into sampling. The parasitoids that emerged from mummies included *Aphidius* sp. and *L. testaceipes* (Cresson).

Visual Counts of Insect Pests on Squash

Aphids. In 2006, squash planted in PM, SSG, VB and SSG/VB plots had significantly lower aphid populations compared with the weedy fallow (F = 6.21; df = 3, 6; P = 0.0011) [Table 4-1]. Squash in the SHPM and SH plots had high aphid numbers that were not significantly different compared with the weedy fallow (Table 4-1).

In 2007, squash planted in the SH treatment had the lowest aphid population compared with all other treatments including the weedy fallow (F = 40.5; df = 3, 6; P < 0.0001). Squash planted in treatments with PM had significantly fewer aphids than SSG (F = 40.5; df = 3, 6; P < 0.0001), but neither was significantly different from the control. Squash plants in the weedy fallow control had significantly fewer aphids compared with squash planted in treatments that had both legume-grass mixtures (Table 4-2).

Whiteflies. In 2006, squash planted in PM and SSG monoculture treatments had significantly fewer whiteflies compared with the weedy fallow control (F = 7.61; df = 3, 6; P = 0.0004), but whitefly numbers on squash in SSG plots were not significantly different than whitefly numbers from SH and SSG/VB treatments. Squash planted in treatments where VB was incorporated into the soil had significantly more whiteflies compared with the control. Squash planted in treatments where SH/PM mixture was incorporated into the soil had significantly fewer whiteflies than the weedy fallow control (Table 4-1).

In 2007, squash growing in the weedy fallow control and in areas where SH/PM mixture was incorporated into the soil had significantly fewer whiteflies compared with corresponding monocultures of squash from the SH and the PM treatment plots (F = 3.10; df = 3, 6; P = 0.0289) (Table 4-2). Squash planted in areas where SSG/VB mixture was incorporated into the soil also had significantly higher whiteflies compared with the weedy fallow control (F = 3.10; df = 3, 6; P = 0.0289). Squash planted in treatments where SSG and VB monocultures were incorporated into the soil were not significantly different from the weedy fallow control (Table 4-2).

Melonworm. Melonworms were recorded only in 2007 on squash plants. Melonworms were significantly lower on squash plants in treatments where PM was incorporated in the soil compared with the weedy fallow control (F = 3.30; df = 3, 6; P = 0.0228) (Table 4-2).

Natural enemies. In 2006, squash planted in plots where VB and SH/PM mixture were incorporated into the soil had significantly more ants than the weedy fallow control, but were not significantly different compared with the monocultures PM and SSG or the SSG/VB mixture (F

= 2.36; df = 3, 6; P = 0.0740) [Table 4-3]. There was no significant difference in the number of spiders, parasitoids, or ladybird beetles among any of the treatments (Table 4-3).

Similarly, in 2007, ant populations on squash plants where SH/ PM mixture was incorporated into the soil was significantly higher than all other treatments including the weedy fallow control (F = 6.46; df = 3, 6; P = 0.0009). Squash plants growing in areas where the SH treatment was incorporated had the lowest numbers of ants compared with other treatments including the control (Table 4-4). Although there were no differences in the number of spiders observed in each treatment, there was a significant difference in the number of parasitoids and aphid mummies among treatments. Squash planted in areas where SSG monoculture was incorporated into the soil had significantly more parasitoids than all other treatments including the weedy fallow control (F = 11.01; df = 3, 6; P < 0.0001). Squash plants in the SH treatment had the lowest numbers of parasitoids, but was not significantly different from PM, VB or the control (F = 11.01; df = 3, 6; P < 0.001) (Table 4-4). Aphid mummies were significantly higher on squash planted in areas where PM, VB and SSG/VB mixture were incorporated into the soil (F = 12.02; df = 3, 6; P < 0.0001). Squash plants from the weedy fallow control treatment had the lowest number of aphid mummies, but these were not significantly different from those plants growing in areas where SH/PM mixture or SSG was incorporated into the soil (Table 4-4).

Leaf Disc Count

Whitefly eggs. In 2006, there was no difference between whitefly eggs on squash plants from any of the treatments (Table 4-5).

However, in 2007, squash plants growing where VB was incorporated in the soil had significantly higher number of whitefly eggs compared with all other treatments (F = 10.06; df = 3, 6; P < 0.0001). Also, squash plants growing in areas where SSG and SH treatments were incorporated, as well as both legume-grass mixtures, SH/PM and SSG/VB, had significantly lower number of whitefly eggs than the weedy fallow control (F = 10.06; df = 3, 6; P < 0.0001) (Table 4-5).

Whitefly immatures. In 2006, squash plants from both grass treatments, SSG and PM, as well as the SH/PM mixture had significantly fewer whitefly immatures compared with the weedy fallow control (F = 3.04; df = 3, 6; P = 0.0312) (Table 4-6).

In 2007, squash plants from the PM and the SH/PM mixture had significantly more whitefly immatures compared all other treatments (F = 14.44; df = 3, 6; P <0.0001). Squash plants from the SSG, VB, SH monocultures and the SSG/VB mixture did not differ in the number of whitefly immatures (Table 4-6).

Blue Pan Trap Results

The blue water pan traps captured aphid species that included Aphelinidae: *Aphelinus* sp., Braconidae: *Aphidius* sp., *Chelonus* sp., and *Lysiphlebus testaceipes* (Cresson), Icheumonidae, Bethylidae, Sclelionidae: *Telenomus* sp., Eucoilidae, Mymaridae, Chalcididae, Eulophidae: Tetrastichinae, Trichogrammatidae, and Encyrtidae: *Metaphycus* sp., *Aphidius* spp. and *Lysiphlebus testaceipes* (Cresson). The family Aphelinidae does include whitefly parasitoids, but no whitefly parastioids were specifically identified. These water traps also captured the parasitoids and hyperparasitoids that were similar to those captured with yellow sticky traps (Figure 4-1). Several species of aphidophagous syrphid flies (Diptera: Syrphidae) were found in the blue pan traps including *Toxomerus* spp., *Platycheirus* spp. *Allograpta* spp. and *Sphaerophoria* spp. (Figure 4-2). Predaceous whitefly predators were not captured during the field-season.

During 2006, squash planted in areas where the SSG /VB mixture was incorporated in the soil had significantly higher number of syrphid flies than all the other treatments including the weedy fallow control (F = 6.48; df = 3, 6; P = 0.0009) (Table 4-7). However, squash planted in soil with the two monocultures, SSG and VB, had low syrphid fly numbers which was not significantly different from the weedy fallow control (Table 4-7).

However during 2007, squash plants in the VB treatment had a significantly higher number of syrphid flies than all other treatments except PM (F = 3.50; df = 3, 6; P = 0.0180) (Table 4-7).

Periphery Yellow Sticky Trap Results

Parasitoids. In the first week, there were significantly more parasitoids in the grass/ weeds area compared to the fruit tree region (F = 4.51; df = 2, 4; P = 0.0489) [Table 4-8]. The grass/weeds area had almost twice as many parasitoids than the fruit tree area. At the end of the three week sampling period, there was no significant difference between the weeds/grass area and the woods, but the woods had 1.5 times more parasitoids than the fruit tree region (F = 4.22; df = 2, 4; P = 0.0559) (Table 4-8). The fluctuations in parasitoid numbers during each week were significant (F = 11.37; 2, 8; P = 0.0046) and there was also a significant interaction between the grass, woods and fruit trees (oranges) habitats and the time (sampling periods) (F = 3.34; df = 2, 24; P = 0.0260). There were significant differences among habitats in the 1st and 3rd week.

Key Pests (Aphids and Whiteflies). There were significantly more aphids found towards the woods habitat compared with the other habitats (F = 9.08; df = 2, 4; P = 0.0087) [Table 4-9]. There was no difference between aphids found towards the orange fruit trees and the grassy area. Whiteflies were also significantly higher in the woods area compared with the fruit tree or grass area (F = 47.20; df = 2, 4; P < 0.0001). There was no significant difference in whitefly numbers between the fruit tree and grass habitat (Table 4-9).

Unbaited Yellow Sticky Trap Results

Parasitoids. In 2006, towards the end of the sampling period, SH had significantly more parasitoids than the weedy fallow control (F = 2.69; df = 3, 6; P = 0.0352). Although there was no significant difference between SH, PM and its corresponding mixture, however, there was an increasing trend in the number of parasitoids recorded in the SH/PM mixture. There was also a significant time effect (F = 6.54; df = 4, 12; P = 0.0049).

During 2007, there were no significant differences in parasitoid numbers among treatments.

Squash Silvering

The rating for SSL disorder was highest on squash plants growing in areas where PM, SH/PM and weedy fallow vegetation was incorporated into the soil. The ratings with all other treatments did not differ significantly (F = 21.82; df = 3, 6; P < 0.0001) (Figure 4-3).

Overall, the SSL disorder ratings did increase with the number of whitefly immatures. There was a significant and strong (P = 0.0081, R^2 = 0.78) correlation between whitefly abundance and SSL disorder rating (Figure 4-4).

Adult Lepidoptera Results

In 2006, there was no significant difference in the number of lepidopteran species that were found in the field (Table 4-10).

However, in 2007, the VB moth had significantly higher numbers than any other species (F = 59.90; df = 3, 3; P < 0.0001). The melon worm and the southern army worm had very low populations. The number of *Mocis* sp. captured was significantly less than the VB moth, but significantly more than the melon worm and the southern armyworm (F = 59.90; df = 3, 3; P < 0.0001) (Table 4-10).

Discussion

In this experiment we evaluated the effect of summer crop treatments on pest and beneficial insect populations in subsequent squash plants. Sentinel squash plants placed in each treatment immediately after cover crop incorporation yielded no parasitoids, and there was no difference in the number of ants, spiders or syrphid flies among treatments. However, the presence of parasitoids on yellow sticky traps from peripheral areas closely surrounding the treatment plots strongly suggests that natural enemies dispersed out of the treatment plots into the surrounding field. When squash plants were established a couple of weeks later, weekly observations of aphid and whitefly populations did not appear to show any pest suppression. However, pooled data from *in situ* counts of key pests and beneficials showed that the monoculture grass cover crop treatments, particularly SSG, were able to reduce aphid and whitefly numbers when compared with the weedy fallow control. We concluded that this reduction could have been a result of the residual population of natural enemies.

The peripheral vegetation that the parasitoids dispersed into were divided into three distinct areas 1) a fruit tree area to the west of the field, 2) a wooded area to the north of the field and 3) a grassy/weedy area to the east and south of the field. It is not surprising that the majority of the parasitoids were found in the grass/weedy area. This border acted as refuge for the parasitoids, providing alternate prey, nectar/pollen resources from the flowering weeds or oviposition sites. The aphid densities found in the weedy/grass areas may have increased the number of parasitoids (density dependent), which later migrated back into the squash plots, where they helped to reduce pests throughout the season. Parasitoids and hyperparasitoids could have been attracted back into the field by attractive hosts (abundance or aphids and whiteflies) or the honeydew secreted from aphids and whiteflies, which acts as an infochemical for foraging parasitoids and hyperparasitoids (Buitenhuis et al. 2004). However, hyperparasitoids are not attracted to honeydew from healthy aphids. Therefore, squash plants from SH or PM treatment, which had low aphid and parasitoid numbers may have had a higher number of hyperparasitoids than squash in other treatment plots.

Grasses such as SSG and PM may have caused a reduction in pest numbers by 1) altering environmental conditions (change in soil nutrient composition) 2) suppressing nematode activity and 3) helping to enhance natural enemy populations.

Environmental conditions are known to modulate plant volatile emissions (Paré and Tumlinson 1999). Therefore, incorporation of various cover crops into the ground may have resulted in differences in soil nutrient content with each treatment. Slightly stressed squash plants that are not infested by pests may be able to produce more volatile chemicals that effectively attract natural enemies of that pest. For instance, squash plants from grass treatments may not have sufficient nitrogen (grasses are known to be nitrogen scavengers and have high C:N ratios). Therefore, when these plants are infested with aphids and whiteflies, elevated levels of volatile chemicals may be produced compared with infested but non-nitrogen stressed squash plants. These higher volatile emissions may serve to attract more natural enemies or pests into those plots. There have been numerous studies showing how predators and parasitoids are more highly attracted to plants that are infested and stressed as a result of compromised environmental conditions (Paré and Tumlinson 1999).

Sorghum sudangrass (SSG) hybrids are also well known to have allelochemical properties that suppress many nematode species (Bowman et al. 1998). For instance, the root knot nematode is one of the most important nematode species affecting squash (Schooley 2005). Therefore, squash plants in the SSG treatment may have had less root knot nematodes than squash in other plots which may have allowed for greater resistance to other insect pests and diseases.

Also, these allelochemical properties displayed by incorporated SSG hybrids would go to suppress weeds once the squash was planted in the fall. Weed suppression potentially causes a reduction in the number of pests because the weeds would not be available to act as secondary hosts for these pests.

In my study, SSG had more coccinellids (*Coccinella septempunctata* L., *Hippodamia convergens* Guérin-Méneville, and *Harmonia axyridis* (Pallas) than all the other treatments. Relay intercropping studies by Parajulee et al. (1997) and others (Robinson et al. (1972) and Burleigh et al. (1973) showed that cotton cash crop associated with sorghum cover crop also had greater numbers of predators than cotton associated with some other cover crops. Predaceous whitefly coccinellids (*Delphastus pusillus*), which were observed in the cover crop treatments (CHAPTER 2) were not captured during the squash cash crop. Usually, these predaceous coccinellids require very high populations of whiteflies to maintain reproduction (Hoelmer et al. 1993). *D. pusillus* larvae are known to consume ~167 eggs per day and up to 1000 eggs before pupating (Hoelmer et al. 1993). From my studies, on average, there were not more than 120 whitefly eggs and immatures in 2006 and not more than 20 whitefly eggs and immatures in 2007. Therefore, there may not have been a sufficient number of whiteflies to allow the predator to successfully reproduce.

Whitefly eggs in squash were overall much higher in 2006 than in 2007. In 2006, it took ~ 5 weeks after cover crop incorporation to establish the squash crop due to initial crop failure, but in 2007, no problems were encountered with planting schedules and squash crop establishment (took place in only 3 weeks). The longer period of time in 2006, combined with decreasing temperatures may have reduced natural enemy populations to the point where they would be insufficient to reduce pest numbers. This may explain the limited pest suppression in 2006. The SSG had significantly lower whitefly eggs and immatures during 2006 and 2007 when compared with the weedy fallow. Like other studies have shown, this grass appears to have potential for suppressing whitefly numbers.

When squash silver leaf (SSL) disorder was observed in 2007, SSG and SH monocultures had the lowest number of whiteflies immatures and significantly lower SSL ratings compared with the weedy fallow. Comparatively, the high numbers of whitefly immatures in PM and SSG also corresponded to high incidences of SSL disorder, which supported the theory that the immatures are the cause of the SSL disorder.

Squash plants from SSG/VB mixture had significantly more syrphid flies and mummified aphids than the weedy fallow control. The emergence of *Aphidius* spp. from these mummies, in addition to *L. testaceipes* that we captured from unbaited yellow sticky traps suggest that these parasitoids may be responsible for suppressing aphid populations in the field. It is possible that the nitrogen supplied by the VB was scavenged by the SSG, causing the emission of more parasitoid-attracting volatiles due to a nitrogen stressed squash plant.

We were also able to see evidence of relationships between pest and beneficial populations. High numbers of aphids from squash plants in VB plots during 2007 and high whitefly populations from the SSG/VB treatment corresponded to the low parasitoid numbers that were observed in those treatments. Conversely, squash plants had low aphid numbers with SSG and correspondingly to high parasitoid populations.

Although, we observed the potential of the SSG/VB mixture to enhance natural enemy populations, squash plants in the SH/PM mixture had significantly more aphids and whiteflies in both field-seasons compared with the weedy fallow. Therefore we would recommend that the SH/PM mixture is not suitable for pest suppression on organic farms. This is not a surprising find, because not all cover crop mixtures enhance natural enemy populations, in fact increasing the vegetational diversity of agroecosystems have variable results (Bugg 1992, Corbett 1998).

A large number of syrphid flies were also captured from squash plants in the SSG /VB treatment. Although this treatment had the lowest aphid population when sampled, the reduction cannot be attributed to the high syrphid population because Bugg et al. (1992) noted that syrphids tended to discriminate against older larger aphid colonies in favor of smaller "promising" colonies. In 2007, the large number of syrphid flies found on squash plants in VB plots may be due to the greater number of weeds in those plots compared with other cover crop plots. Aphidophagous syrphids respond well to nectar and pollen produced by flowering weeds (Häni et al. 1998).

We captured only a small number of melon worm moths, the larvae of which are serious pests of cucurbits. Squash plants from the PM had significantly fewer number of melon worms, *Diaphania hyalinata* L., compared with other plots. This may be related to the allelochemical activity of PM which indirectly affects the plant's resistance to herbivory (Khan et al. 2005). Parasitoids such as Trichogrammatidae, Chalcididae and other egg parasitoids such as Scleionidae may also have been responsible for reduced numbers of lepidopteran pests.

Overall, there is good evidence to conclude that residual populations of natural enemies from incorporated cover crops and allelochemicals from selected crops are able to suppress pests in a subsequent cash crop. This pest suppression will, however, depend on the diversity of the peripheral vegetation surrounding the field because alternate food sources need to be present to maintain natural enemy populations until the squash crop is established.

Table 4-1. Aphid and whitefly means from *in situ* squash leaf counts from various cover crop treatments in 2006

Treatment	Mean ± SEM	
	Aphids[a]	Whiteflies[b]
SH	157.00 ± 7.71ab	241.50 ± 30.44bc
PM	129.75 ± 6.90bc	169.50 ± 21.40d
SH + PM	176.25 ± 1.79a	196.00 ± 11.00cd
SSG	130.50 ± 14.5bc	182.25 ± 9.63cd
VB	119.50 ± 4.97c	344.00 ± 33.49a
SSG + VB	122.00 ± 10.87c	223.00 ± 13.22bcd
Weedy fallow (Control)	168.00 ± 8.68a	260.00 ± 24.77b

Aphid and whitefly data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 6.21$; df = 3, 6; $P = 0.0011$
[b] $F = 7.61$; df = 3, 6; $P = 0.0004$

Table 4-2. Aphid and whitefly means from *in situ* squash leaf counts from various cover crop treatments in 2007

Treatment	Mean ± SEM		
	Aphids[a]	Whiteflies[b]	Melon worm[c]
SH	161.75 ± 6.82d	65.25 ± 7.18abc	2.50 ± 0.95bc
PM	216.00 ± 10.31c	95.57 ± 9.46ab	1.75 ± 1.43c
SH + PM	346.25 ± 14.67a	70.25 ± 6.51dc	5.25 ± 2.49bc
SSG	250.75 ± 10.81b	80.25 ± 8.46abcd	14.50 ± 4.27a
VB	339.00 ± 13.34a	78.00 ± 6.25 bcd	8.50 ± 3.47ab
SSG + VB	326.00 ± 18.04a	103.50 ± 14.31a	5.00 ± 2.54bc
Weedy fallow (Control)	224.00 ± 3.48bc	63.00 ± 5.11d	6.75 ± 1.93ab

Aphid, whitefly and melon worm data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 40.5$; df = 3, 6; $P < 0.0001$

[b] $F = 3.10$; df = 3, 6; $P = 0.0289$

[c] $F = 3.30$; df = 3, 6; $P = 0.0228$

Table 4-3. Mean number of natural enemies from *in situ* squash counts from cover crop treatments in 2006

Treatment	Mean ± SEM			
	Ants[a]	Spiders[b]	Parasitoids[c]	Lady beetles[d]
SH	6.25 ± 2.56c	0.00 ± 0.00	0.25 ± 0.25	0.00 ± 0.00
PM	18.00 ± 3.58ab	1.00 ± 0.57	0.25 ± 0.25	0.00 ± 0.00
SH + PM	26.00 ± 9.74a	0.75 ± 0.25	0.25 ± 0.25	0.00 ± 0.00
SSG	15.00 ± 2.67abc	0.00 ± 0.00	0.50 ± 0.28	0.50 ± 0.28
VB	20.75 ± 3.35a	0.75 ± 0.47	0.00 ± 0.00	0.00 ± 0.00
SSG + VB	18.50 ± 4.50ab	0.00 ± 0.00	0.25 ± 0.25	0.25 ± 0.25
Weedy fallow (Control)	7.75 ± 1.81bc	0.00 ± 0.00	0.25 ± 0.25	0.00 ± 0.00

Beneficial arthropod data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 2.36$; df = 3, 6; $P = 0.0336$

[b] $F = 2.49$; df = 3, 6; $P = 0.0620$

[c] $F = 0.43$; df = 3, 6; $P = 0.8503$

[d] $F = 2.05$; df = 3, 6; $P = 0.1108$

Table 4-4. Mean number of natural enemies from *in situ* squash leaf counts from cover crop treatments in 2007

Treatment	Mean ± SEM			
	Ants[a]	Spiders[b]	Parasitoids[c]	Aphid mummy[d]
SH	5.57 ± 1.65c	1.00 ± 0.57	2.00 ± 1.15c	10.75 ± 1.31b
PM	15.50 ± 2.17b	0.75 ± 0.47	4.25 ± 0.75bc	18.75 ± 3.94a
SH + PM	37.50 ± 8.10a	0.25 ± 0.25	6.75 ± 1.88b	9.25 ± 2.25bc
SSG	17.75 ± 4.80b	0.25 ± 0.25	15.50 ± 0.64a	4.75 ± 2.83bc
VB	11.75 ± 1.31b	0.25 ± 0.25	3.77 ± 1.65bc	22.50 ± 2.95a
SSG + VB	13.50 ± 2.25b	0.25 ± 0.25	6.50 ± 2.06b	22.25 ± 2.13a
Weedy fallow (Control)	12.50 ± 1.93b	0.25 ± 0.25	4.75 ± 1.70bc	3.55 ± 1.55c

Beneficial arthropod data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 6.46$; df = 3, 6; $P = 0.0009$

[b] $F = 0.89$; df = 3, 6; $P = 0.5213$

[c] $F = 11.01$; df = 3, 6; $P < 0.0001$

[d] $F = 12.02$; df = 3, 6; $P < 0.0001$

Table 4-5. Mean number of whitefly eggs recorded from squash leaf disc counts during 2006 and 2007

Treatment	Mean ± SEM whitefly eggs	
	2006[a]	2007[b]
SH	142.50 ± 3.30	1.25 ± 0.63c
PM	153.00 ± 12.04	16.5 ± 3.32b
SH + PM	121.50 ± 19.56	2.25 ± 1.31c
SSG	170.25 ± 8.16	1.00 ± 0.71c
VB	174.25 ± 19.73	25.25 ± 5.57a
SSG + VB	94.75 ± 15.78	2.75 ± 1.25c
Weedy fallow (Control)	178.00 ± 41.12	16.00 ± 5.88b

Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 2.41$; df = 3, 6; $P = 0.0690$

[b] $F = 10.06$; df = 3, 6; $P < 0.0001$

.

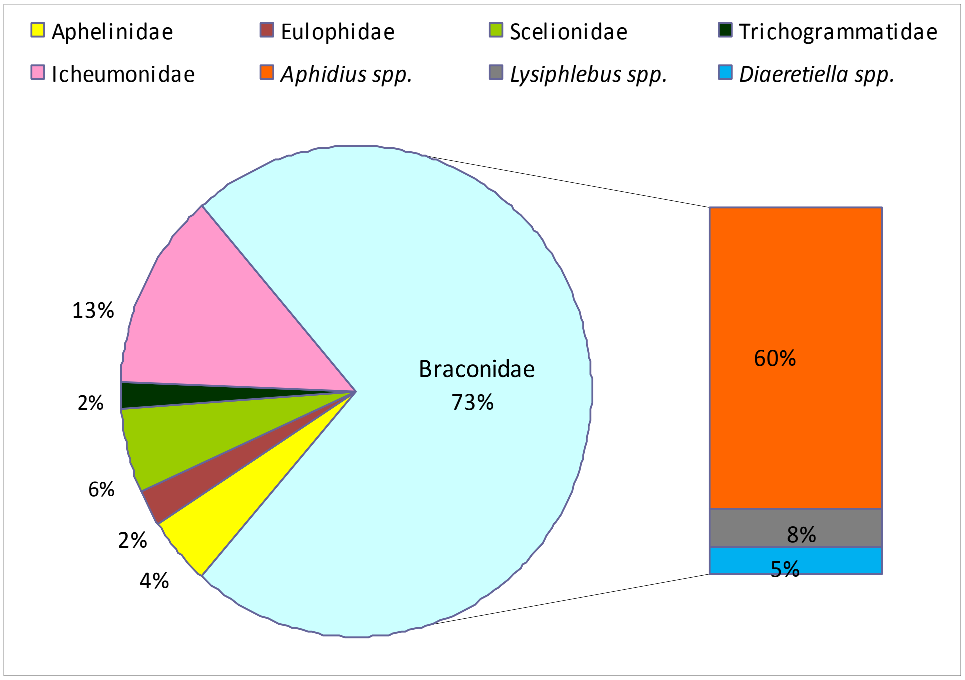
Table 4-6. Mean number of whitefly immatures recorded from squash leaf disc counts during 2006 and 2007

Treatment	Mean ± SEM whitefly immatures	
	2006[a]	2007[b]
SH	16.00 ± 1.29ab	1.75 ± 1.18b
PM	12.00 ± 4.41bc	16.75 ± 1.93a
SH + PM	4.75 ± 1.65c	14.25 ± 2.86a
SSG	9.50 ± 3.30bc	1.75 ± 1.18b
VB	14.00 ± 5.01abc	2.75 ± 1.88b
SSG + VB	12.75 ± 3.11abc	3.00 ± 2.12b
Weedy fallow (Control)	25.00 ± 4.02a	5.75 ± 1.03 b

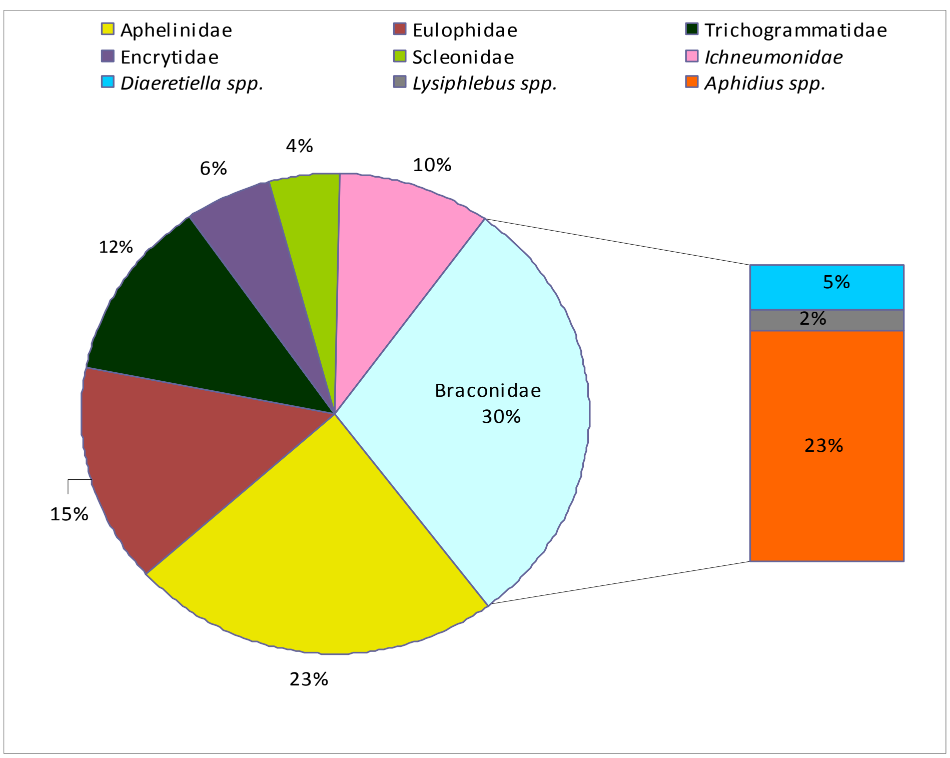
Whitefly immature data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different P = 0.05 (LSD)

[a] F = 3.04; df = 3, 6; P = 0.0312

[b] F = 14.44; df = 3, 6; P < 0.0001

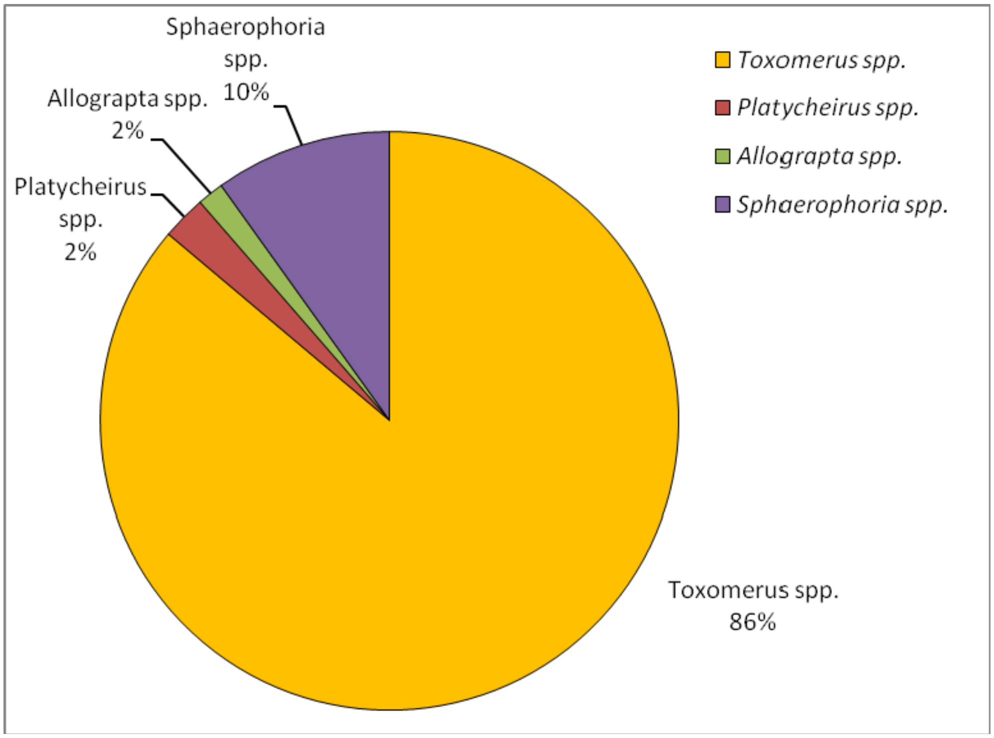


A.

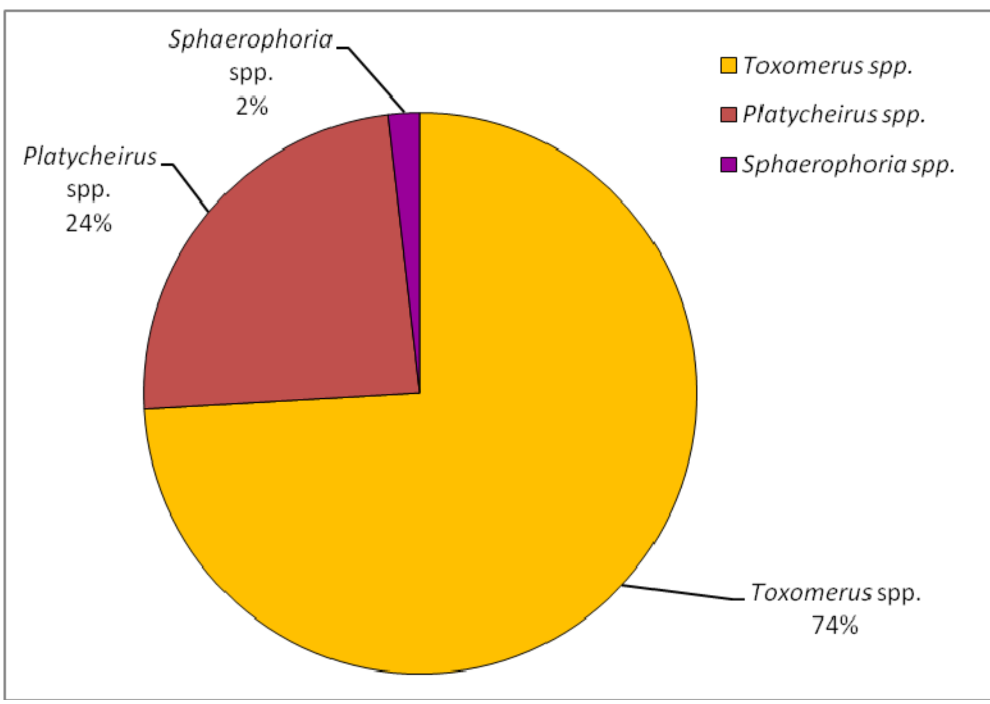


B

Figure 4-1. Major parasitoid families in an organic squash field.



A.



B.

Figure 4-2. Syrphid fly populations in an organic squash field in A) 2006 and B) 2007.

Table 4-7. Syrphid fly populations from each treatment in 2006 and 2007 from blue pan traps

Treatment	Mean ± SEM syrphid flies in squash	
	2006[a]	2007[b]
SH	10.00 ± 1.77b	1.25 ± 0.63b
PM	9.75 ± 1.54b	3.00 ± 0.91ab
SH + PM	9.25 ± 1.65b	1.50 ± 0.28b
SSG	4.75 ± 1.54c	2.00 ± 0.81b
VB	4.75 ± 1.37c	5.00 ± 0.82a
SSG + VB	17.50 ± 3.71a	2.00 ± 0.41b
Weedy fallow (control)	2.75 ± 0.85c	1.25 ± 0.63b

Syrphid fly data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 6.48$; df = 3, 6; $P = 0.0009$

[b] $F = 3.50$; df = 3, 6; $P = 0.0180$

Table 4-8. Parasitoid population dynamics from peripheral habitats surrounding an organic field

Treatment	Mean ± SEM parasitoid numbers in 2007		
	Week 1[a]	Week 2[b]	Week 3[c]
Grassy/weedy area	52.60 ± 11.46a	38.80 ± 6.74	16.00 ± 4.24ab
Fruit tree (oranges)	23.20 ± 4.39b	34.20 ± 6.68	8.00 ± 2.02b
Woods	41.00 ± 5.00ab	21.80 ± 1.82	25.80 ± 4.98a

Parasitoid data were square root transformed before analysis, but the means shown represent untransformed values. Means followed by the same letters are not significantly different ($P = 0.05$ according to least square means test following repeated measures analysis, LS).

[a] $F = 4.51$; df = 2, 4; $P = 0.0489$

[b] $F = 1.72$; df = 2, 4; $P = 0.2396$

[c] $F = 4.22$; df = 3, 6; $P = 0.0559$

Table 4-9. Population dynamics of key pests from peripheral habitats surrounding an organic field in 2007

Habitats	Mean ± SEM key pests	
	Aphids[a]	Whiteflies[b]
Fruit tree (oranges)	10.80 ± 0.66b	7.00 ± 0.94b
Grass	9.40 ± 1.91b	3.00 ± 0.71b
Woods	19.20 ± 2.65a	32.40 ± 4.05a

Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 9.08$; df = 2, 4; $P = 0.0087$
[b] $F = 47.20$; df = 2, 4; $P < 0.0001$

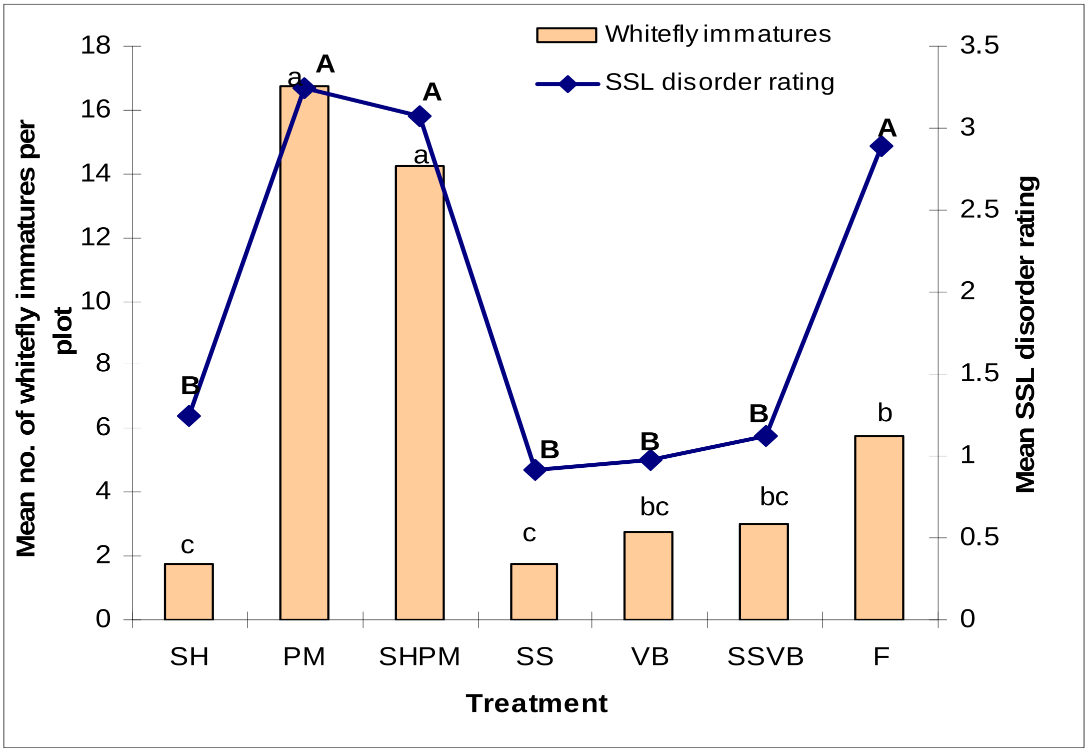


Figure 4-3. Populations of whitefly immatures with squash silver leaf disorder rating in 2007

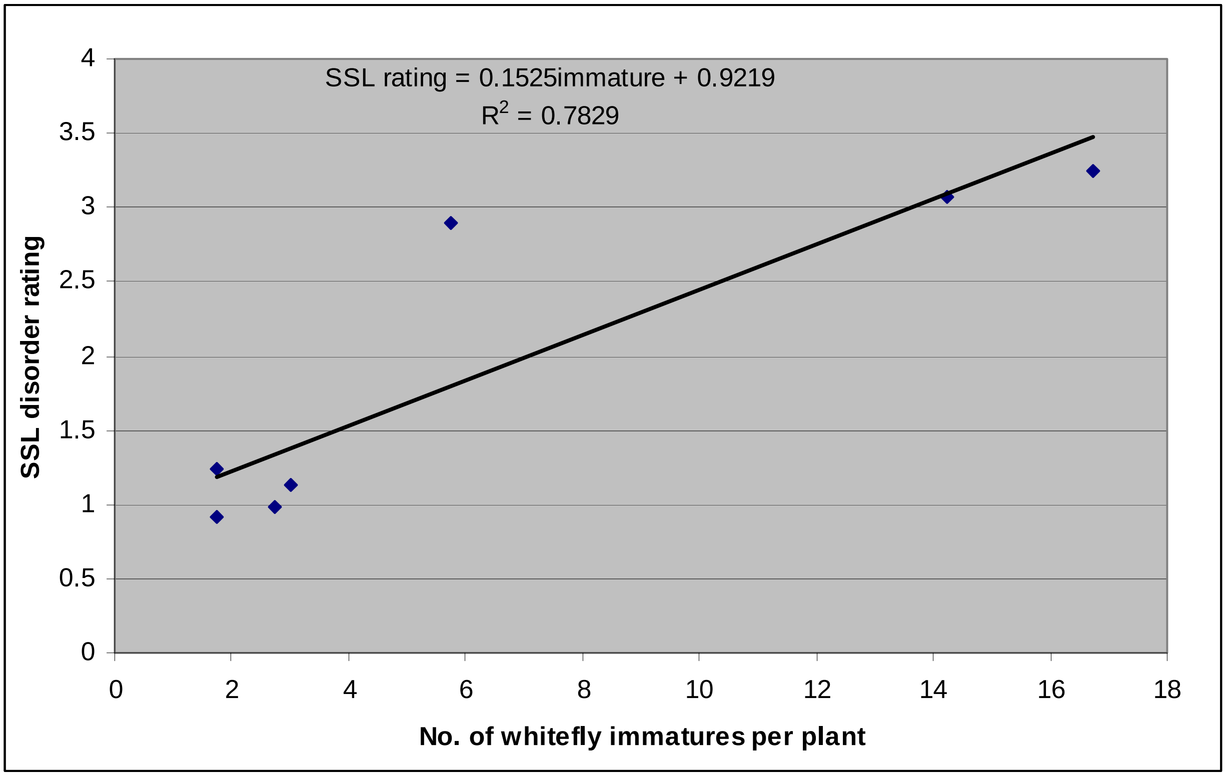


Figure 4-4. Relationship of immature whiteflies with squash silverleaf disorder rating

Table 4-10. Mean number of adult lepidoptera captured from pheremone baited bucket traps in squash during 2006 and 2007

Moth species	Mean ± SEM adult lepidoptera from squash	
	2006[a]	2007[b]
Anticarsia gemmatalis (VB moth)	0.75 ± 0.47	25.00 ± 2.25a
Mocis sp.	1.75 ± 1.03	9.75 ± 2.25b
Diaphania hyalinata (Melonworm)	0.00 ± 0.00	0.75 ± 0.47c
Spodoptera eridania (Southern armyworm)	0.25 ± 0.25	0.50 ± 0.50c

Adult lepidopteran data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 1.39$; df = 3, 3; $P = 0.3089$

[b] $F = 59.90$; df = 3, 3; $P < 0.0001$

CHAPTER 5
INFLUENCE OF PEST POPULATIONS FROM COVER CROPS ON TOTAL AND MARKETABLE YIELD OF SUMMER SQUASH

Economically, summer squash (*Cucurbita pepo* L.) is one of the most highly profitable vegetables to produce in the world (Kemble et al. 2005). In 2007 Florida's squash production was estimated at 10,900 acres with production of 101 million pounds valued at approximately $52.9 million USD (NASS 2008). Aphids and whiteflies are highly destructive pests on summer squash because they transmit diseases and injure the plant by sucking sap from the leaves thereby greatly reducing the yield and profitability of the crop. It is not uncommon for a squash plant to have three or four different aphid-transmitted viruses, which consequently causes poor yields (Molinar et al. 1999). Whiteflies also have caused a reduction in root dry weight of other cucurbits including pumpkins (McAuslane et al. 2004).

Cover crops can have positive and negative effects on plant yield. Generally, cover crops improve plant yields by enhancing soil health. Cover crops promote biologically active soil, which helps crops to resist pest pressures and ultimately increases yield (Phatak 1998, Sarrantonio 1998). Cover crops can also indirectly improve crop yield by reducing or eliminating the use of pesticides, which favors healthy populations of natural enemies of pestiferous insects (Phatak 1998). Tillman et al. (2004) have reported that cotton yields were significantly higher when cotton was interplanted with a number of cover crops (crimson clover, rye, a legume mixture and a combination of rye and the legume mixture) but that this was not as a result of pest suppression. Conversely, Robertson et al. (1991) reported that there was no significant effect on corn, peanut or cotton yield with the addition of a cover crop.

In addition to yield, cover crops help to speed up the infiltration of excess surface water, relieve compaction and improve soil structure, enhance nutrient cycling, and increase soil organic matter that further encourages beneficial soil microbial life (Bowman et al. 1998). For instance, legume cover crops such as VB and SH have nitrogen-fixing bacteria in their root nodules, and are able to add nitrogen to the soil. This improves the growth of the subsequent cash crop especially if they are used as a green manure (Caamal-Maldonado et al. 2001, Rich et al. 2003). Additionally, when leguminous cover crops are digested by soil microbes, polysaccharide by-products are produced that hold small soil particles together as aggregrates, which provides good soil aeration and less compaction (Sarrantonio 1998).

Alternatively, cover crops can negatively impact crop yields. The addition of a cover crop to a field may reduce the amount of water available for the following cash crop. Reduced water levels increase plant stress making it more susceptible to insect pests and diseases, which ultimately causes a reduction in yield (Robertson 1999). Some cover crops are susceptible to nematodes and some may be more attractive to key pests than beneficials. Also, graminaceous cover crops can result in high C:N ratios that can result in less nitrogen being available for crop growth during establishment. Additionally, it is also difficult to manage cover crop residues to prepare a good seed bed.

My goal was to determine how pest populations from cover crop treatments can affect total and marketable yield in summer squash. Knowing which cover crops to use to improve plant yield is an excellent starting point for successful crop management.

Methods

Field Experiments

Field experiments were conducted at the University of Florida, Plant Science Research and Education Unit in Citra, FL. The experimental setup was a randomized complete block design with four replicates of seven treatments. Cover crop treatments included two legumes (sunn hemp [SH] and velvet bean [VB]), two grasses (pearl millet [PM] and sorghum sudangrass [SSG]), and two legume-grass mixtures (sorghum sudangrass/velvet bean [SSG/VB] and sunn hemp/pearl millet [SH/PM]). Summer squash (Cougar F1 untreated) was direct seeded with a monosem planter into 8 m x 12 m plots with 4 rows. The distance between the 4 rows was 6 feet (1.8 m) whereas the spacing between each squash plant was 18 inches (0.46 m). An organic fertilizer, Nature Safe 10-2-8, (Griffin Industries Inc. Diamond Company, FL) was applied through a drop spreader at 33 lbs/plot for the squash seedlings on 26th October 2006 and 9th October 2007. Squash was direct seeded on 19 October 2006. Squash fruit was harvested 6 times throughout the 2006 field-season; on the 14th, 19th, 26th of December 2006, and on the 2nd, 8th, and 9th of January 2007. During the 2007 field-season, squash was direct seeded on 10th October, and was harvested 8 times: 21st, 26th, 28th November and 3rd, 6th, 10th, 12th, and 14th of December 2007.

Aphids and Whitefly Sampling

Each week visual (*in situ*) counts were conducted in the field for aphids and whiteflies. These key pests were visually assessed by randomly surveying the leaves of eight squash plants from each of the two inner rows. Each squash plant was surveyed for approximately one minute. All other insects encountered on the leaves were also recorded. Leaf samples on which aphid mummies were discovered were brought back to the laboratory. Parasitoids were identified upon emergence.

Unbaited yellow sticky traps (treated area 406.45 cm^2, Great Lakes IPM, Vestaburg, MI), and blue water pan traps (Packer Ware Bowls, Gainesville, FL) with a 5% detergent solution (Colgate Palmolive Co. New York, NY) to break the surface tension (Webb et al. 1994) were used primarily for natural enemy counts. Both traps were placed diagonally across for each other in the middle rows of each treatment plot.

Yield Sampling

All four rows within each squash plot were harvested by hand and total and marketable yields were assessed in the 2006 and 2007 field-seasons. Squash was harvested 1 to 2 times per week when the fruit reaches 7 cm or more, weighed with a hand scale, and the data were recorded. Squash is not a cool season crop, and protection of the crop was required in both 2006 and 2007. Meteorological data for Citra, FL were constantly monitored, and for days that were likely to have frost, the squash was harvested 2 or 3 days before signs of frost. Frost blankets were also used to prevent frost damage.

Unmarketable fruits were distinguished from marketable fruits based on size, color, and deformities. Unmarketable fruits were less than 5 inches in length, had dark brown discolorations on the surface of the fruit, showed signs of insect frass contamination and / or insect dug holes. Fruits that displayed physiological disorders or a dark colored dry rot especially at the bottom end of the fruit (known as blossom-end rot), were also characterized as unmarketable. Fruits that did not show signs as those described above were considered marketable. Once the squash was harvested and weighed, the total and marketable yield from each treatment plot was calculated.

Statistical Analysis

Data from squash numbers and squash yield were analyzed using analysis of variance (ANOVA) with means separated by LSD test ($\alpha = 0.05$). Means were considered significantly different from each other when P-value ≤ 0.05.

Results

Insect Population

Aphids. In 2006, both grasses, PM and SSG had significantly lower aphid populations compared with the weedy fallow ($F = 6.21$; df = 3, 6; $P = 0.0011$). The VB and the SSG/VB mixture also had significantly less aphids than the weedy fallow control. The SH and SH/ PM mixture were not significantly different than the control (Figure 5-1a).

In 2007, PM had significantly fewer aphids than SSG (F = 40.5; df = 3, 6; P < 0.0001), but neither was significantly different compared with the control. The weedy fallow control had significantly fewer aphids compared with both mixtures, but had significantly more aphids than SH (Figure 5-1b).

Whiteflies. In 2006, the trend for whiteflies was similar to aphids. Both grasses, PM and SSG, had significantly fewer whiteflies compared with the weedy fallow control (F =7.61; df = 3, 6; P = 0.0004). The VB treatment had significantly more whiteflies compared with the control, but the SH was not significantly different than the control. Of the two legume-grass mixtures only SH/PM had significantly fewer whiteflies compared with the weedy fallow control. The other mixture was not significantly different than the control (Figure 5-2a).

In 2007, the weedy fallow control and SH/ PM mixture and had significantly fewer whiteflies compared with the SH and the PM monocultures (F = 3.10; df = 3, 6; P = 0.0289) (Figure 5-2b). However, the other mixture, SSG/VB had significantly higher whiteflies compared with the weedy fallow control (F = 3.10; df = 3, 6; P = 0.0289). Both corresponding monocultures, SSG and VB were not significantly different from the weedy fallow control (Figure 5-2b).

Total Squash Yield

In 2006, total squash yields were significantly higher with the PM treatment compared with all other treatments (F = 3.53; df = 3, 6; P = 0.0172), except for SH (Table 5-1). All other treatments (including SH) gave low yields that were not significantly different than the weedy fallow control (Table 5-1). There was no correlation in 2006 between the key pests (aphids and whiteflies) with the total yield).

In 2007, the SH and the SSG had significantly higher total squash yields than both legume-grass mixtures, SH/PM and SSG/VB ($F = 3.57$; df = 3, 6; $P = 0.0166$) but were not significantly different from the weedy fallow control (Table 5-1).

Also during 2007, there was a strong negative correlation between aphids and total yield ($P = 0.0498$, $R = -0.7550$). However, there was no correlation between whiteflies and total yield.

Marketable Squash Yield

In 2006, marketable squash yields were significantly higher in plots of PM compared with all other treatments except for SH ($F = 3.16$; df = 3, 6; $P = 0.027$) (Table 5-2). All other treatments gave low yield that were not significantly different from the weedy fallow control. During the 2006 field-season, there was no correlation between either aphids or whiteflies and marketable yield.

In 2007, squash yields were significantly higher in SSG plots than in the weedy fallow control plots ($F = 4.74$; df = 3, 6; $P = 0.0046$) (Table 5-2), but this yield was not significantly different from plots with SH, VB or PM. Also during the 2007 field-season, there was no correlation between either aphids or whiteflies and marketable yield.

The SH/PM plots had the lowest squash yields compared with other treatments except the SSG/VB mixture and the control. Marketable yields averaged between 12 and 29 kg/ 36 m^2 per treatment in the 2006 field season compared with 10 to 15 kg / 36 m^2 per treatment in the 2007 field season.

Discussion

Total Squash Yield. High populations of aphids and whiteflies are known to affect squash yields (Frank and Liburd 2004). In the first year of the study, there was a density dependent relationship between insect pest pressures and squash yield. During the 2006 field-season, squash yield was higher in PM plots than the weedy fallow control plot and all other cover crop treatments except the SH, which was not significantly different than the control. High squash yields from the PM treatment may be due to the significantly lower number of aphids that were observed on squash plants from PM plots when *in situ* counts were conducted. A lower population of aphids implies that there was a reduction in feeding damage and virus transmission to these squash plants. Pearl millet is well known for its weed-suppressing allelochemical properties. Once incorporated into the soil, PM may produce allelochemicals that prevent the build up of weeds as hosts for aphids (or whiteflies), thereby reducing pest pressure and influencing yield (Narwal 2006).

Alternatively, squash plants in the PM treatment may have had access to better quality soil due to the incorporation of PM. Healthy soils with adequate nutrients affect the relationship between plant and pests (Häni et al. 1991).Grasses tend to have higher C:N ratios and large amounts of biomass, which improves the soil health when incorporated into the soil (Baldwin and Creamer 2008).

The high population of aphids in both the SH/PM mixture and the weedy fallow control could also account for the low squash yields that were recorded from those treatments. Higher pest pressures imply that the squash plant is exposed to more feeding damage, diseases and virus transmission, which reduces yield. Although there was a high number of whiteflies on squash in the VB plot, since there was no correlation between whiteflies and total yield, other factors such as poor soil quality or inadequate moisture may have contributed to low squash yield.

During the 2007 field-season, SH plots had a significantly higher squash yield than its corresponding SH/PM mixture. Also, SSG had a higher squash yield than its corresponding SSG/VB mixture. For instance, SH plots had significantly lower numbers of aphids and whiteflies than the SH/PM mixture and weedy fallow control. Significantly lower incidences of squash silver leaf (SSL) disorder was recorded in the SH plot, which was associated with lower whitefly pressure (CHAPTER 4) and higher yield. The SSL disorder (due to whitefly immatures) may have influenced the squash total yield in 2007.

Marketable Squash Yield. In 2006 and 2007, both monoculture treatments, SH and PM, had significantly higher marketable squash yields than their corresponding mixture. Although there was no correlation between aphids and whiteflies and marketable yield in 2007, there were noticeable density-dependent trends between the key pests and the yield, which should not be ignored. The SH monoculture had significantly more aphid mummies than the control plots. The parasitoids that emerged from the mummies were identified as various *Aphidius* spp. These parasitoids suppress aphid populations (Powell and Pell 2007), which may account for some of the observed lower pest pressures that could influence yield. The SSG monoculture was also observed to have significantly more parasitoids such as *Lysiphlebus testaceipes* and various *Aphidius* spp. than the SSG/VB mixture. Another possibility is that, varying squash yields may have been due to better soil nutrient and water content based on the incorporation of the different cover crop treatments.

Overall, there was good evidence to show that higher pest populations corresponded with lower squash yields while lower pest populations corresponded with higher squash yields. Future research should involve analyzing PM for chemical compounds that may be responsible for suppressing pests and perhaps be introduced as an organic insecticide for growers. Monocultures that increase the number of beneficial insects should be identified and used in a combination treatment to suppress key pest populations.

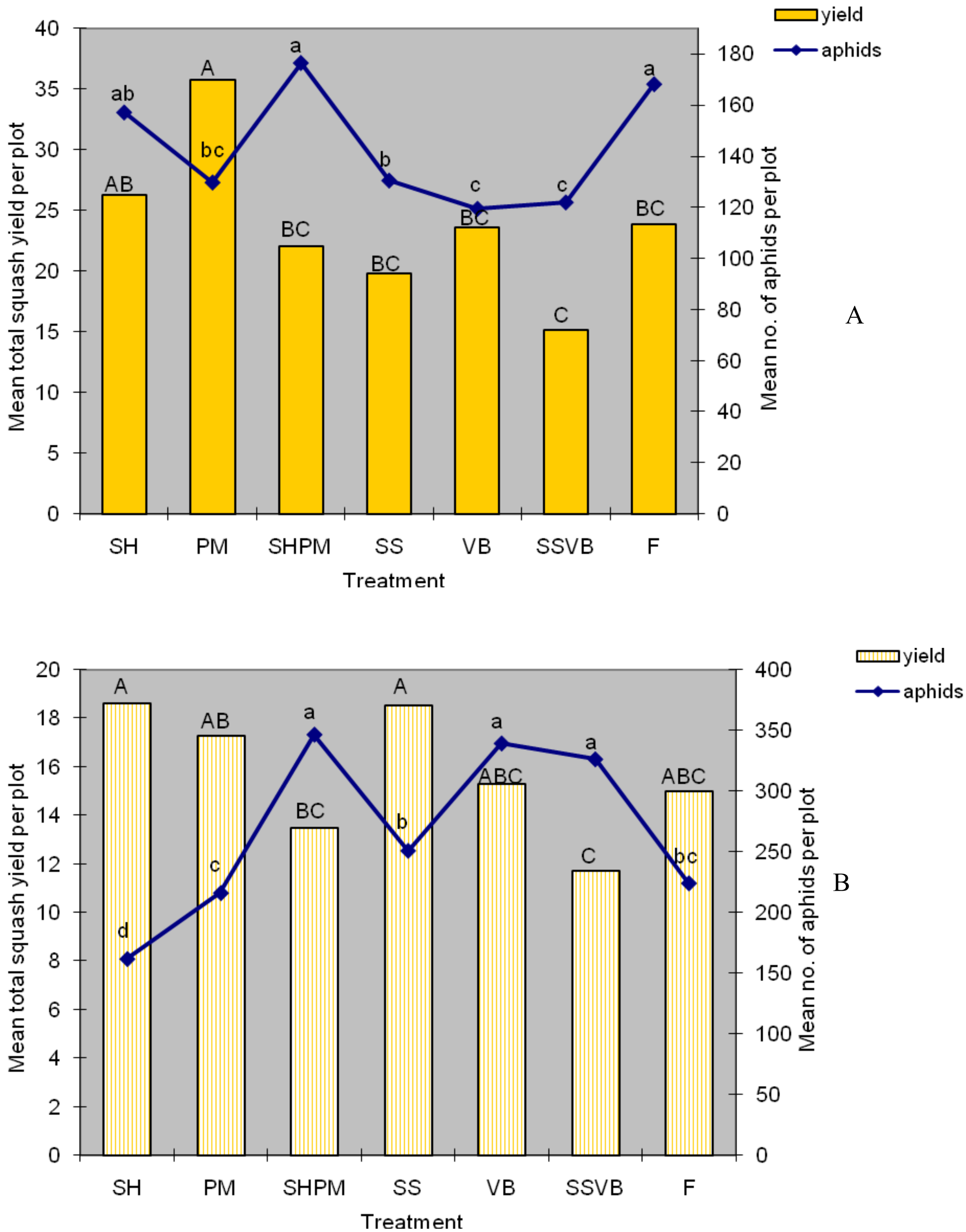


Figure 5-1. Relationship between aphids with total squash yield in A) 2006 and B) 2007.

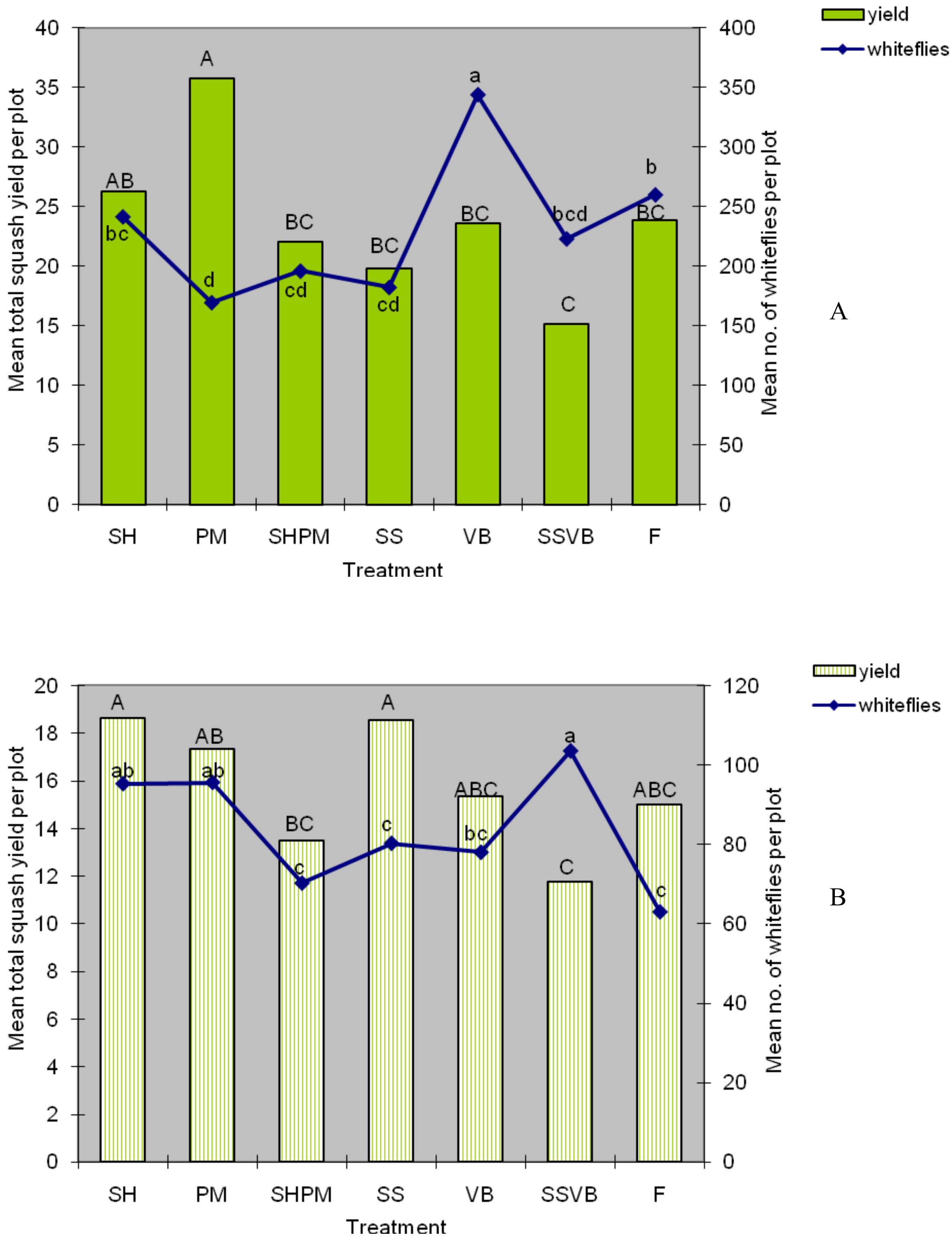


Figure 5-2. Relationship between whiteflies with total squash yield in A) 2006 and B) 2007.

Table 5-1. Mean total squash yield from each cover crop treatment for the 2006 and 2007 field seasons

Treatment	Mean ± SEM total yield (kg/36 m^2) of squash	
	2006	2007
SH	26.31 ± 2.02ab	18.60 ± 1.36a
PM	35.81 ± 3.87a	17.30 ± 1.86ab
SH + PM	22.08 ± 6.38bc	13.48 ± 1.39bc
SSG	19.86 ± 2.74bc	18.53 ± 0.90a
VB	23.58 ± 2.46bc	15.31 ± 1.27abc
SSG + VB	15.17 ± 2.45c	11.73 ± 0.86c
Weedy fallow (control)	23.87 ± 5.24bc	14.99 ± 1.83abc

Total squash yield data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 3.53$; df = 3, 6; $P = 0.0172$
[b] $F = 3.57$; df = 3, 6; $P = 0.0166$

Table 5-2. Mean marketable squash yield from each treatment for the 2006 and 2007 field seasons

Treatment	Mean ± SEM marketable yield (kg/36 m^2) of squash	
	2006	2007
SH	21.36 ± 2.18ab	15.36 ± 1.41ab
PM	29.09 ± 3.55a	13.75 ± 1.23ab
SH + PM	17.71 ± 5.48bc	9.59 ± 1.48d
SSG	16.62 ± 2.39bc	16.44 ± 1.06a
VB	18.99 ± 1.59bc	12.81 ± 0.93abc
SSG + VB	12.21 ± 2.21c	9.87 ± 0.54dc
Weedy fallow (control)	19.57 ± 4.41bc	12.69 ± 1.63bdc

Marketable squash yield data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different P = 0.05 (LSD)

[a] $F = 3.16$; df = 3, 6; $P = 0.027$

[b] $F = 4.74$; df = 3, 6; $P = 0.0046$

CHAPTER 6
TECHNIQUES FOR MONITORING APHIDS (HEMIPTERA: APHIDIDAE) AND WHITEFLIES (HEMIPTERA: ALEYRODIDAE) IN SUMMER SQUASH

Summer and winter squash are major vegetable crops grown commercially in Florida (Roberts and Pernezny 2001). Florida is the second leading state that produces squash for fresh market sales and squash is one of the few crops that are shipped out of Florida every month of the year to grocery stores nationwide (AgMRC 2007, Mossler and Nesheim 2001, Roberts and Pernezny 2001). During the 2007 squash field season, Florida growers harvested 10,900 acres of squash, valued at $53 million USD (NASS 2008).

Summer squash (*Cucurbita pepo* L.) is highly susceptible to several aphid species including, but not limited to, the green peach aphid (*Myzus persicae* Sulzer), the melon aphid (*Aphis gossypii* Glover) and the cowpea aphid (*Aphis craccivora* Koch) (Mossler and Nesheim 2001). These aphids damage and weaken squash by sucking sap from the leaves and excreting honeydew, which promotes the growth of sooty mold, eventually resulting in wilting and leaf distortion (Molinar et al. 1999). Aphids also transmit over 50 viruses in cucurbits (Molinar et al. 1999, Capinera 2007). The melon aphid in particular is regarded as one of the most destructive pests on squash in the United States (Vásquez et al. 2006) because it transmits potyviruses to cucurbits. Other viruses that are transmitted by aphids include: the papaya ring spot virus type W (PRSV-W), the watermelon mosaic virus 2 (WMV2), the zucchini yellow mosaic virus (ZYMV) and four viroids, which causes yellowing, mosaic, leaf blisters and reduced fruit set (Kucharek and Purcifull 1997, Frank and Liburd 2005, Molinar et al. 1999).

Whiteflies are also regarded as a serious pest of cucurbits. The silverleaf whitefly, *Bemisia argentifolii* (Bellows and Perring) destroys squash plants by sucking plant sap which results in defoliation, stunting, poor plant yields and growth of sooty mold from honeydew excretions (Molinar et al. 1999). In Florida, the greenhouse whitefly, *Trialeurodes vaporariorum*

(Westwood) is not an important pest. However the silverleaf whitefly is a key pest of squash that causes annual economic losses for growers (McAuslane 2007, Henneberry and Faust 1999). Feeding of the immature forms of the silverleaf whitefly causes a severe physiological disorder in the squash plant resulting in silvering of the leaf surface and fruit blanching, commonly known as the squash silverleaf (SSL) disorder (Frank and Liburd 2005).

The reproduction of female aphids allow for rapid population increases especially when resources are abundant (Liburd and Nyoike 2008). One aphid female can produce 80 offspring per week and can have several overlapping generations per year (Woiwood and Hanski 1992). A decline in the quality and quantity of resources elicit a switch from apterous (wingless) to alate (winged) morphs and aphids migrate to search for new host plants. Migration increases the efficiency of disease transmission.

In organic agricultural systems, effective and reliable monitoring techniques will aid in the detection and quantification of aphid and whitefly populations in squash plants. Good monitoring techniques will provide information about the pest, its natural enemies and crop biology, which allows growers to use more timely and appropriate management techniques to suppress pest populations. Effective monitoring techniques are dependent on several factors including, labor (time spent to count insects on traps), total trap catches, and first trap catch (Liburd et al. 2000).

Several techniques are used to monitor aphid and whitefly populations. For instance, yellow sticky traps are used to monitor aphids, whiteflies and their associated natural enemies in greenhouse and field conditions (Liburd and Nyoike 2008). Blue pan traps with 5% detergent solution have also been used to assess aphid but not whitefly populations (Frank and Liburd 2005, Liburd and Nyoike 2008). *In situ* counts are another standard technique used to detect the presence of aphids, whiteflies. My objective was to compare several monitoring techniques for aphids and whiteflies so that growers can use this information to improve their timing of management techniques for key pests on organic farms.

Materials and Methods

Field experiments were conducted at the University of Florida, Plant Science Research and Education Unit in Citra, FL on certified organic acreage. The experimental setup was a randomized block design with four replicates of seven cover crop treatments. Each plot measured 12 m x 12 m and was separated by 12 m wide mowed alleys, which intended to reduce the movement of pest and beneficial arthropods between plots. After the cover crop treatments were incorporated into the soil, summer squash was direct seeded into each treatment plot. Each plot had four rows and each row had 27 squash plants. In 2006, summer squash (Cougar F1 cultivar) was direct seeded by hand with 0.46 m spacing between each plant, whereas in 2007 squash was seeded with a monosem planter with 0.23 m spacing between each plant and thinned. Comparisons of the different monitoring techniques were based on total trap catches, early detection and efficiency. Efficiency was measured in terms of labor and time involved in processing traps and was calculated by the product of the average time spent per trap and the cost per hour for counting traps.

Aphids

During 2006 and 2007, aphids (nymphs and adults) were monitored once per week for a 4 week period, from 15 November to 11 December and 8 November to 30 December, respectively. The three different sampling methods evaluated were:

- Unbaited yellow sticky traps
- Blue water pan traps
- *In situ* visual counts

Unbaited yellow sticky traps (treated area 406.45 cm^2, Great Lakes IPM, Vestaburg, MI) were used to primarily monitor for alate aphids. One yellow sticky trap was tied to a 0.6 m stake and placed in the middle of the inner two rows of squash plants in each treatment plot. Traps were left in the field for 48 h and replaced once each week. Traps that were brought back to the laboratory for analysis was covered with a layer of plastic wrap and labeled accordingly. Traps were stored in the refrigerator until ready to be processed. These traps were read by counting a sub-sample of 15 from 63 one inch squares on the traps due to high aphid densities (Finn 2003). This sub-sampling technique eliminates counting errors and reduces the time spent per trap (Finn 2003). Briefly, each yellow sticky trap was overlaid with a gridded transparent paper. The transparency was divided into 63, 1-inch squares. Forty-eight of these squares were colored in, while 15 were left transparent. The area under these 15 transparent 1-inch squares was examined under a 40X dissecting microscope, and aphids and whiteflies were counted and recorded (Finn 2003). Natural enemies in all 63 squares on the sticky traps were counted and recorded for each yellow trap. The time taken to count each trap was recorded.

Blue water pan traps (Packer Ware Bowls, Gainesville, FL) were the second unbaited trap used in this experiment. These traps were supported by tomato wire cages (45 cm x 15 cm x 20 cm), filled with water and detergent (5% detergent solution) (Colgate Palmolive Co. New York, NY) to break the surface tension (Webb et al. 1994). One blue pan trap was placed diagonally across from an unbaited yellow sticky trap on the opposite end on the plot to monitor aphids and their natural enemies. Blue pan traps were constructed by punching three holes into the top edge of a blue plastic bowl 15 cm in diameter. Traps were left in the field for one week. At the end of the week, the contents from each trap were emptied and into a small plastic container, labeled accordingly and taken back to the laboratory to be analyzed. The number of aphids and natural enemies from the blue traps were counted under a 40X dissecting microscope and the contents placed in 15 x 45 mm vials (Fisherbrand®, Fisher Scientific, Pittsburgh, PA) with 70% ethanol for storage. The time taken to go through each blue pan trap was noted and recorded.

Each week visual (*in situ*) counts were conducted in the field for aphids (alate and apterous), whiteflies, as well as other insects. All insects were visually assessed by randomly examining the leaves of eight squash plants from each of the two inner rows for ~1 minute.

Whiteflies

The two different sampling methods used to evaluate whitefly populations were:

- Unbaited yellow sticky traps

- *In situ* visual counts

The techniques used to monitor whitefly populations were the same as those used for aphids.

Statistical Analysis

Aphid data were analyzed using analysis of variance (ANOVA) followed by the least significant difference (LSD) means separation tests (SAS Institute 2003). Whitefly data were analyzed using the Student's t-test (SAS Institute 2003). Means were considered significantly different from each other when P-values ≤ 0.05.

Results

Aphids

Approximately 60% of the aphids recorded in the field from *in situ* counts were alate aphids. Results from the first capture date in 2006 showed that significantly more aphids were observed using the *in situ* monitoring technique ($F = 77.60$; df = 2, 3; $P < 0.001$) compared with unbaited yellow sticky traps or blue pan traps (Figure 6-1). Unbaited yellow sticky traps also had significantly more aphids than the blue pan traps for the first sample date (Figure 6-1). For the

2006 season, *in situ* counts had significantly higher numbers of alate aphids than the yellow sticky traps or blue pan traps in summer squash (F = 1321.29; df = 2, 3; P < 0.001). *In situ* counts had 3 times more aphids compared with the yellow sticky trap and 19 times more aphids than the blue pan trap (Table 6-1). Six times more aphids were captured with yellow sticky traps compared with blue pan traps (Table 6-1).

First capture results in 2007 had similar trends to those in 2006. *In situ* counts recorded significantly higher numbers of aphids than unbaited yellow sticky traps or blue pan traps (F = 118.13; df = 2, 3; P < 0.001) (Figure 6-1). Also, throughout the 2007 field season, *in situ* counts had significantly (~3 times) more aphids than the yellow sticky traps and 23 times more aphids than blue pan traps (F = 898.41; df = 2, 3; P < 0.001) (Table 6-1).

Whiteflies

In 2006, first capture date from both monitoring techniques showed that whiteflies (*B. argentifolli*) were significantly higher in yellow sticky traps compared with *in situ* counts (t = 4.66; df = 6; P = 0.0035) (Figure 6-2). However, throughout the 2006 field season, there were significantly more whiteflies from *in situ* counts compared with yellow sticky traps (t = 20.99; df = 6; P < 0.0001) (Table 6-2).

In 2007 the number of whiteflies were significantly higher with *in situ* counts compared with unbaited yellow sticky traps from first captures (t = 18.12; df = 6; P < 0.0001) (Figure 6-2). Also, throughout the 2007 field season, similar results to 2006 were observed. Whitefly numbers were significantly higher with *in situ* counts compared with yellow sticky traps (t = 20.90; df = 6; P < 0.0001) (Table 6-2).

Time Required Per Monitoring Technique

During 2006 it took significantly longer to count aphids and whiteflies on squash using *in situ* counts compared with unbaited yellow sticky traps or blue pan traps (F = 70.49; df = 2, 9; P

< 0.0001) (Figure 6-3). The time taken to count aphids and whiteflies was approximately 17 minutes per plot while it took approximately 6 to 7 minutes per trap to assess key pests with unbaited yellow sticky traps or blue pan traps. In 2007, the time taken to assess key pests was similar to 2006. *In situ* counts took twice as long to count aphids and whiteflies (F = 435.52; df = 2, 9; P < 0.0001) compared with using unbaited yellow sticky traps or blue pan traps (Figure 6-3).

Discussion

Aphids. These experiments indicated that *in situ* counts can be considered the best method to use when monitoring aphid populations in the field because aphid numbers were significantly higher with this method when compared with the two other monitoring techniques. There were significantly higher numbers of aphids that were observed on the first capture date using *in situ* counts during 2006 and 2007 compared with unbaited yellow sticky traps or blue pan traps. This implies that *in situ* counts are more sensitive than yellow sticky traps or blue pan traps for giving fairly accurate assessments of pest densities in the field. Choosing a suitable and effective sampling technique is a function of the labor and cost involved in using such a technique. *In situ* counts can be quite laborious and many hours can be spent in the field inspecting plants. It took twice as long to visually assess aphid populations in the field using *in situ* counts compared with yellow sticky traps or blue pan traps. Once labor costs are factored in, the efficiency of using *in situ* counts is reduced compared with yellow sticky traps or blue pan traps. Also, knowing the degree of pest densities on the plants at a very early stage will improve the timing to make good pest management decisions (Flint and Gouveia 2001) If there are low aphid densities in the field, then *in situ* counts would not necessarily be the best option for getting a proper estimate.Therefore the use of alternate monitoring techniques could be considered.

Both yellow sticky traps and blue pan traps have been used to monitor aphid populations (Adlerz 1978, Frank and Liburd 2005, Liburd and Nyoike 2008). Kring (1972) reported that aphids are attracted to both long and shortwave light. Yellow light (~ 550 nm) in particular will induce aphids to alight on the yellow surface but shortwave light can be highly attractive as they fly upwards towards the light, even avoiding yellow surfaces in the process. Since the blue color of my water pan traps falls in the shortwave length of light (~ 420 to 480 nm), it would be logical to assume that aphids could potentially be similarly attracted to blue pan traps as they are to yellow sticky traps. However, my results showed that yellow sticky traps proved to be a more effective monitoring technique because they captured significantly more aphids than blue pan traps in both 2006 and 2007. According to Kring (1972) when aphids are in cruising or sniffing flight, they can be repelled by shortwave light (reflected by my blue pan traps) and turn by preference toward longwave light (yellow sticky traps). The time spent to assess aphids on yellow sticky traps was not significantly different from the time taken to assess aphids in blue pan traps. However, it took a significantly shorter period of time to assess aphids on yellow sticky traps compared with *in situ* counts. Therefore, in order to save time and labor, it would be more cost effective to consider using yellow sticky traps as alternative to *in situ* counts or blue pan traps. If sticky traps are chosen, the effectiveness of the yellow sticky trap depends on the stickiness of the adhesive and whether the traps are made of cardboard or plastic. Plastic appears to be more durable. If the trap gets wet due to heavy rainfall or dirty because of dirt blown on to it by strong winds, then the efficiency of these traps will decline (Flint and Gouveia 2001). Also, the adhesive nature of the sticky card can make accurate aphid identification a challenging task.

Whiteflies. For first capture date in 2006, my results showed that unbaited yellow sticky traps had more whiteflies than *in situ* counts. This is not surprising because whiteflies are difficult to see when their numbers are low. In addition, they are highly attracted to yellow. In 2007, *in situ* counts had significantly more whiteflies than unbaited yellow sticky traps. The sensitivity of unbaited yellow sticky traps tend to vary depending on pest densities early in the season.

Similar to aphids, *in situ* counts had higher whitefly numbers than yellow sticky traps throughout the season, which may imply that this is also the best technique for monitoring whitefly populations in the field. It took a significantly longer time to count whiteflies with *in situ* counts compared with unbaited yellow sticky traps. The combination of labor costs and the time involved with *in situ* counts reduces the efficiency of this method. Additionally, whiteflies are very mobile insects and care has to be taken when inspecting leaves so as to prevent whiteflies from flying away once the leaf is disturbed.

Unbaited yellow sticky traps are an alternative to *in situ* counts to monitor and control whitefly populations (Hoelmer 1998, Weinzierl et al. 2005). Like aphids, whiteflies use color cues to find landing sites (Byrne and Bellows 1991). They are also highly attractive to yellow wavelengths (535 to 580 nm) (Chu et al. 2000, Byrne and Bellows 1991). Unbaited yellow sticky traps captured fewer whiteflies in 2007 than in 2006, and this could be due to a number of factors. Climatic conditions vary greatly each year in Florida and so weather conditions may have caused the decline in the whitefly numbers in 2007. Reduced numbers could also have been due to increased parasitism in the field.

Overall, my studies demonstrated that *in situ* counts had the highest numbers of aphids and whiteflies, which appears to be a superior method of monitoring aphids and whiteflies. In addition the number of beneficials could be measured very easily, which would allow growers to make the best decision in deciding the type of pest management strategies to adopt in a timely fashion.

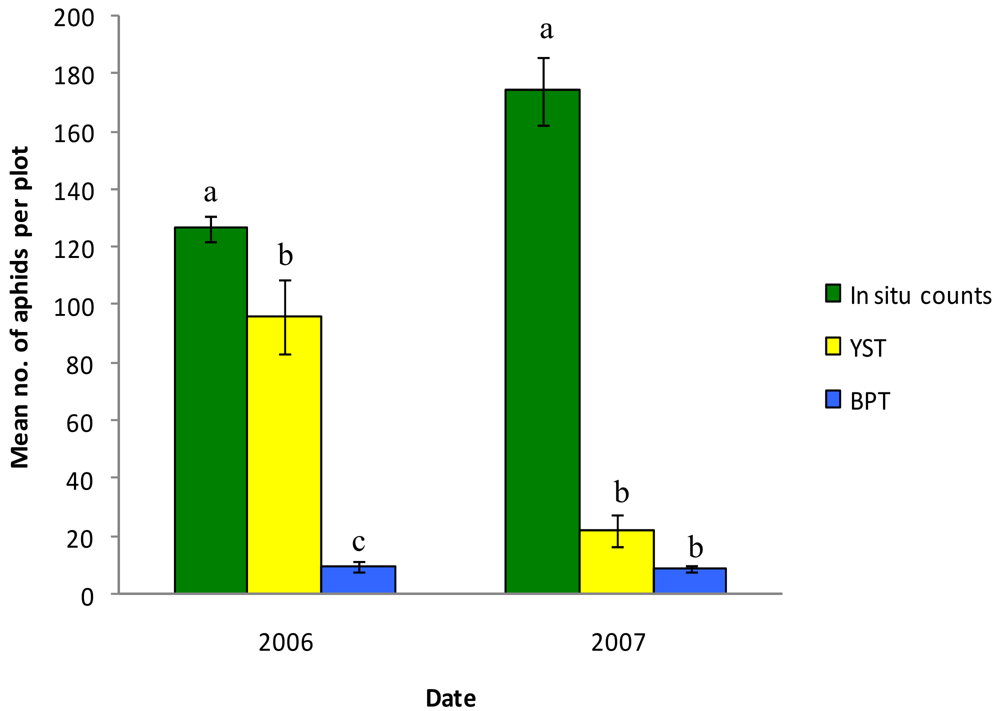


Figure 6-1. First day trap captures *in situ* counts, yellow sticky traps (YST) and blue pan traps (BPT) for aphids in summer squash

Table 6-1. Comparison techniques for monitoring aphids in summer squash throughout the entire season in Citra, FL.

Monitoring techniques used in squash crop	Mean ± SEM aphids from different monitoring techniques	
	2006[a]	2007[b]
Blue pan traps	53.75 ± 6.53c	73.00 ± 4.77c
In situ visual counts	1003.00 ± 10.59a	1708.50 ± 43.66a
Yellow sticky traps	308.75 ± 16.96b	500.50 ± 18.34b

Means followed by the same letters are not significantly different $P = 0.05$ (LSD)
[a] $F = 1321.29$; df = 2, 3; $P < 0.0001$
[b] $F = 898.41$; df = 2, 3; $P < 0.0001$

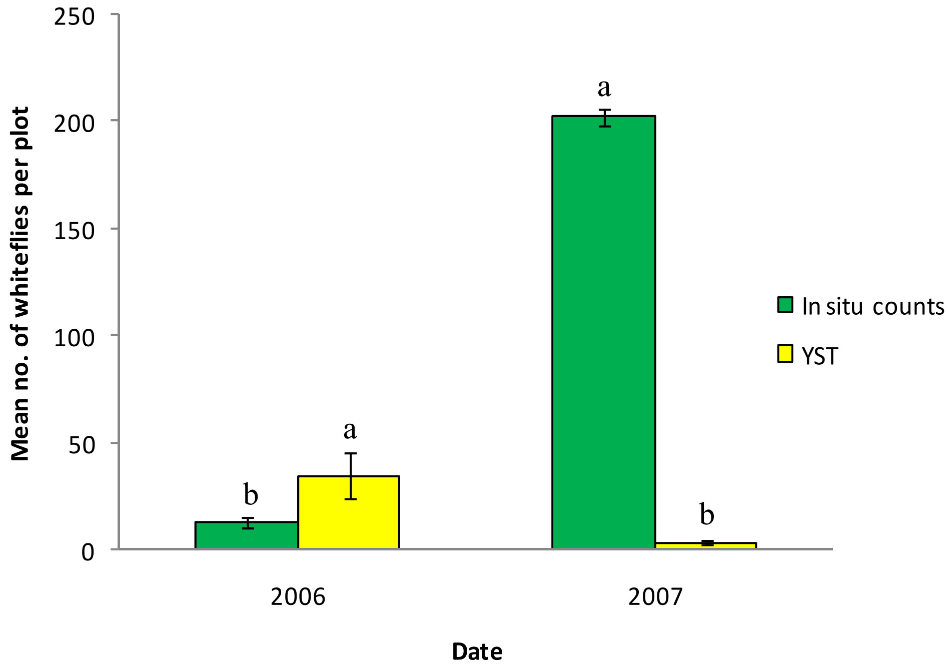


Figure 6-2. First day trap captures in situ counts and yellow sticky traps (YST) for whiteflies in summer squash

Table 6-2. Comparison techniques for monitoring whiteflies in summer squash throughout the entire season in Citra, FL.

Monitoring techniques used in squash crop	Mean ± SEM whitefly adults from different monitoring techniques	
	2006	2007
In situ visual counts	1616.3 ± 71.47a	520.25 ± 23.25a
Yellow sticky traps	112.50 ± 4.73b	20.00 ± 5.70b

Whitefly egg data were square root transformed before analysis but means shown reflect untransformed data. Means followed by the same letters are not significantly different $P = 0.05$ (LSD)

[a] $F = 0.81$; df = 3, 6; $P = 0.5725$

[b] $F = 1.09$; df = 3, 6; $P = 0.4048$

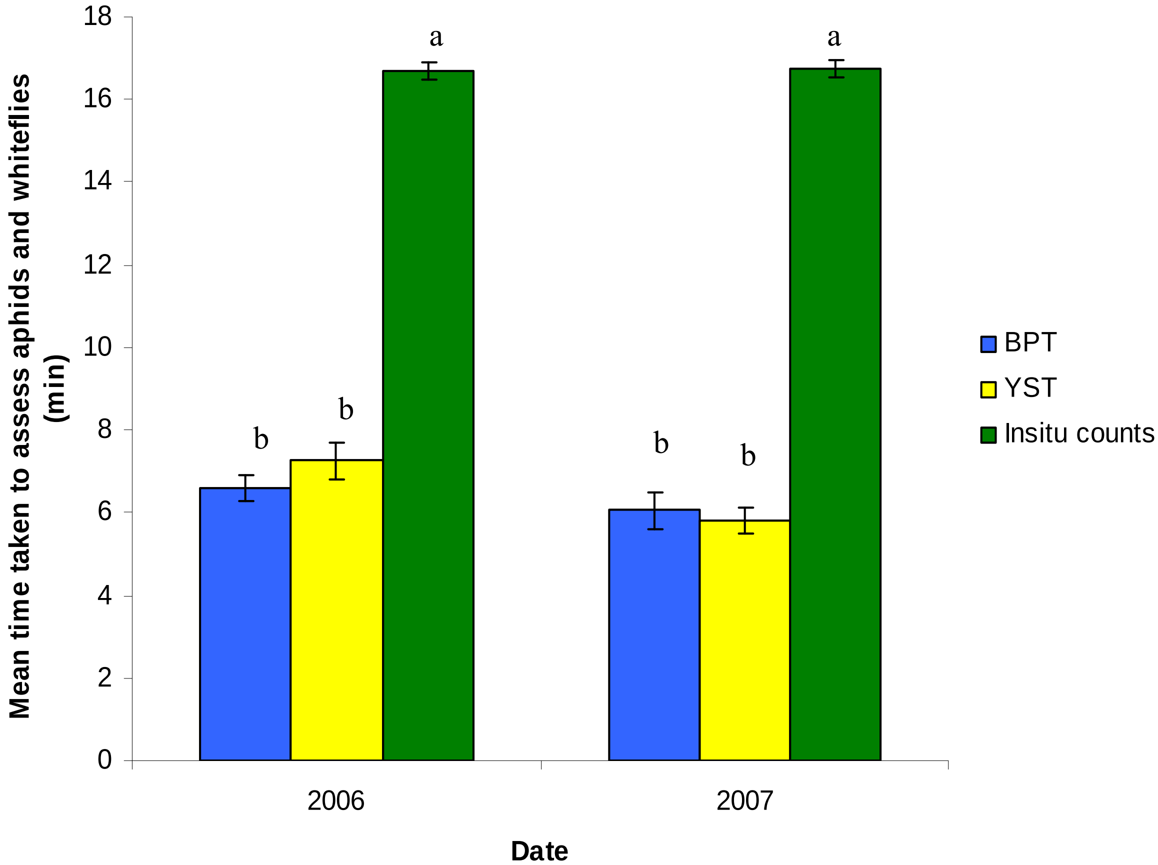


Figure 6-3. Time taken (minutes) to assess aphids and whiteflies for blue pan traps (BPT), yellow sticky traps (YST) and *in situ* counts for aphids and whiteflies in summer squash

LIST OF REFERENCES

Adams, D. B. 2000. Commercial production and management of squash and cucumbers. Bulletin 1178. University of Georgia College of Agricultural and Environmental Sciences Cooperative Extension Service, Athens, GA.

Adlerz, W. C. 1987. Cucurbit potyvirus transmission by alate aphids (Homoptera:Aphididae) trapped alive. J. Econ. Entomol. 80: 87-92.

Gold, M. V. 2007. Organic production/organic food: information access tools; March 2008. U. S. Dep. Agric., Alternative Farming Systems Information Center, Washington, D.C.

Agricultural Marketing and Resource Center (AgMRC). 2007. Squash. Iowa State University, Ames, Iowa (http://www.agmrc.org/agmrc/commodity/vegetables/squash/)

Ambrosino, M. D., P. C. Jepson, and J. M. Luna. 2007. Hoverfly oviposition response to aphids in broccoli fields. Entomol. Exp. Appl. 122: 99 – 107.

Antignus, Y. 2000. Manipulation of wavelength-dependent behavior of insects: an IPM tool to impede insects and restrict epidemics of insect-borne viruses. Virus Res. 71: 213-220.

Antonelli, A. L. 2003. Hover or Syrphid Flies. Washington State University Cooperative Extension WSU Puyallup REC PLS-83, Pullman WA.

Awmack, C. S., and S. R. Leather. 1998. Growth and Development, pp. 135-152. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Bachmann, J. 2002. Farmers' markets marketing and business guides, pp. 1-4 *In*: Appropriate Technology Transfer for Rural Areas (ATTRA) Publication IP146.

Balkcom, K. S. and D. W. Reeves. 2005. Sunn hemp utilized as a legume cover crop for corn production. Agron. J. 97: 26-31.

Beane, K. A. and R. L. Bugg. 1998. Natural and artificial shelter to enhance arthropod biological control agents, pp. 239-254. *In* C. H. Pickett and R. L. Bugg [eds.], Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkley, CA.

Beckage, N. E. and D. B. Gelman. 2004. Wasp parasitoid disruption of host development: implications for new biologically based strategies for insect control. Annu. Rev. Entomol. 49: 299-330.

Bengtsson, J., J. Ahnström, and A. Weibull. 2005. The effects of organic agriculture on biodiversity and abundance: a meta-analysis. J. Appl. Ecol. 42: 261-269.

Blackman, R. L., and V. F. Eastop. 2007. Taxanomic issues, pp. 1-22. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Bonina, J. and D. J. Cantliffe. 2004. Seed production and seed sources of organic vegetables, HS 981. Institute of Food and Agricultural Life Sciences, University of Florida, Gainesville, FL.

Bowman, G., C. Shirley, and C. Cramer, 1998. Benefits of cover crops, pp. 8-11 *In*: A. Clarke [ed.], Managing Cover Crops Profitably, Beltsville, MD.

Brewer, M. J. 1995. Syrphid flies. Extension Publication B-1013.22.University of Wyoming, Laramie, WY.

Brewer, M. J., and N. C. Elliott. 2004. Biological control of cereal aphids in North America and mediating effects of host plant and habitat manipulations. Annu. Rev. Entomol. 49: 219-242.

Browning, H. W., C. W. Melton, and R. R. Saldana. 1985. Release and evaluation of exotic parasites for biological control of the Mexican rice borer on sugar cane in the lower Rio Grande Valley, 1983-1985. Texas Agricultural and Experimental Station Progress Report. 4344.

Bugg, R. L. 1991. Cover crops and control of arthropod pests of agriculture, pp.167-163. *In* Cover crops for clean water: Proceedings of an international conference West Tennessee Experiment Station, 9-11 April 1991, Jackson TN.

Bugg, R. L. and C. H. Pickett, 1998. Introduction: Enhancing biological control - habitat management to promote natural enemies of agricultural pests, pp. 1-24. *In* C. H. Pickett and R. L. Bugg [eds.], Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkley, CA.

Bugg, R. L. and C. Waddington. 1994. Managing cover crops to manage arthropod pests of orchards. Agr. Ecosyst. Environ. 50:11-28.

Bugg, R. L. and R. T. Ellis. 1990. Insects associated with cover crops in Massachusetts. Biol. Agric. Hortic. 7: 47-68.

Bugg, R. L., F. L. Wäckers, K. E. Brunson, J. D. Dutcher, and S. C. Phatak. 1991. Cool-season cover crops relay intercropped with cantaloupe: influence on a general predator, *Geocoris punctipes* (Hemiptera: Lygaeidae). J. Econ. Entomol. 84: 408 – 416.

Bugg, R. L., S. Phatak, and J. D. Dutcher. 1990. Insects associated with cool season cover crops in Southern Georgia: implications for pest control in truck-farm and pecan agroecosystems. Biol. Agric. Hortic. 7: 17-45.

Buitenhuis, R., J. N. McNeil, G. Boivin, and J. Brodeur. 2004. The role of honeydew in host searching of aphid hyperparasitoids. J. Chem. Ecol. 13: 273-285.

Burleigh, J. G., J. H. Young, and R. D. Morrison.1973. Strip-cropping's effect on beneficial insects and spiders associated with cotton in Oklahoma. Environ. Entomol. 2: 281-285.

Byrne, D.N., and T.S. Bellows. 1991. Whitefly Biology. Annu. Rev. Entomol. 36: 431-457.

Caamal-Maldonado, J. A., J. J. Jiménez-Osornio, A. Torred-Barrangán, and A. L. Anaya. 2001. The use of allelopathic legume cover and mulch species for weed control in cropping systems. Agron. J. 93: 27-36.

Capinera, J. L. 2007. Melon aphid or cotton aphid, *Aphis gossypii* Glover (Insecta: Hemiptera: Aphididae), EENY-173. Institute of Food and Agricultural Life Sciences, University of Florida, Gainesville, FL.

Cárcamo, H. A., D. Parkinson, and J. W. A. Volney. 1998. Effects of sulphur contamination on macroinvertebrates in Canadian pine forests. Appl. Soil Ecol. 9: 459-464.

Chambliss, C. G., R. M. Muchovej, and J. J. Mulahey. 2003. Cover Crops, SS- AGR-66. Institute of Food and Agricultural Life Sciences, University of Florida, Gainesville, FL.

Chaney, W. 1998. Biological control of aphids in lettuce using in-field insectaries, pp. 73-84. *In* C. H. Pickett and R. L. Bugg [eds.] Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkley, CA.

Chu, C., T. P. Freeman, J. S. Buckner, T. J. Henneberry, D. R. Nelson, and E. T. Natwick. 2001. Susceptibility of upland cotton cultivars to *Bemisia tabaci* Biotype B (Homoptera: Aleyrodidae) in relation to leaf age and trichome density. Ann. Entomol. Soc. Am. 94: 743 – 749.

Chu, C. C., P. J. Pinter, Jr., T. J. Henneberry, K. Umeda, E. T. Natwick, Y. Wei, V. R. Reddy, and M. Shrepatis. 2000. Use of CC traps with trap base colors for silverleaf whiteflies (Homoptera: Aleyrodidae), thrips (Thysanoptera: Thripidae), and leafhoppers (Homoptera: Cicadellidae). J. Econ. Entomol. 93: 1329-1337.

Corbett, A. 1998. The importance of movement in the response of natural enemies to habitat manipulation, pp. 25-48. *In* C. H. Pickett and R. L. Bugg [eds.], Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkley, CA.

Costello, M. J. and M. A. Altieri. 1995. Abundance, growth rate and parasitism of *Brevicoryne brassicae* and *Myzus persicae* (Homoptera: Aphididae) on broccoli grown in living mulches. Agri. Ecosyst. Environ. 52: 187-196.

Crawford, C. 2007. Organic Opportunity: Diversify your offerings with organic selection. Citrus and Vegetable Magazine, pp. 14 – 16.

Creamer, N. G. 1999. Summer Cover Crops. North Carolina State University Cooperative Extension HIL-37. (http://www.ces.ncsu.edu/depts/hort/hil/hil-37.html)

Creamer, N. G. and K. Baldwin. 2000. An evaluation of summer cover crops for use in vegetable production systems in North Carolina. HortScience 35: 600 – 603.

Dewar, A. M. 2007. Chemical Control, pp. 391-422. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Dixon, A. F. G. 1977. Aphid Ecology: Life cycles, polymorphism and population regulation. Annu. Rev. Ecol. Syst. 8: 329-353.

Dixon, A. F. G. 1985. Structure of aphid populations. Ann. Rev. Entomol. 30: 155-174.

Drees, B. M. 1993. Aphid management. Texas Agricultural Extension Service. (http://entowww.tamu.edu/extension/bulletins/uc/uc-031.html)

Economic Research Service. 1997. USDA, Washington, D.C.

Economic Research Service. 2008. USDA, Washington, D.C.

Ferguson, G. 2005. Factors to consider in using biocontrol agents for aphids. Ontario Ministry of Agriculture Food and Rural Affairs fact sheet. (http://www.omafra.gov.on.ca/english/crops/facts/biocontrol_aphids.htm)

Finn, E. 2003. Developing integrated pest management (IPM) techniques for managing key insect pests of blueberries in the Southeastern United States, pp. 57-59, MS Thesis, Department of Entomology and Nematology, University of Florida, Gainesville, FL.

Frank, L. D. and O. E. Liburd. 2005. Effects of living and synthetic mulch on the population dynamics of whiteflies and aphids, their associated natural enemies, and insect-transmitted plant diseases in zucchini. Environ. Entomol. 34: 857-865.

Frank, H. J. and R. F. Mizell III. 2004. Coccinellidae – Ladybird beetle, Featured Creatures EENY-170. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Godfrey K. E., W. H. Whitcomb, J. L. Stimac. 1989. Arthropod predators of VB caterpillar, *Anticarsia gemmatalis* Hübner (Lepidoptera: Noctuidae), eggs and larvae. Environ. Entomol. 18: 118-123.

Goodell, P., B. Grafton-Cardwell, S. Koike, C. Morgner, M. Parrella, J. Stewart-Leslie, P. Weddle, C. Wilen, F. Zalom. 2001. Monitoring and Decision-Making Guidelines, pp. 149-196. *In* M. L. Flint and P. Gouveia [eds.], IPM in practice: principles and methods of integrated pest management. University of California publication 3418, Oakland, CA.

Goolsby, J. A., M. A. Ciomperlik, B. C. Legaspi, Jr, J. C. Legaspi, and L. E. Wendel. 1998. Laboratory and field evaluation of exotic parasitoids of *Bemisia tabaci* (Gennadius) (Biotype "B") (Homoptera: Aleyrodidae) in the lower Rio Grande Valley of Texas. Biol.Control 12: 127-135.

Greene, C. and A. Kremen. 2000. U.S. organic farming in 2000-2001, adoption of certified systems. (http://www.ers.usda.gov/Publications/aib780/)

Häni, F. J., E. F. Boller, and S. Keller. 1998. Natural regulation at the farm level, pp. 161-210. *In* C. H. Pickett and R. L. Bugg [eds.], Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkley, CA.

Hanna, R., F. G. Zalom, and C. L. Elmore. 1995. Integrating cover crops in grapevine pest and nutrition management: The transition phase. Sustainable Agriculture. (http://www.sarep.ucdavis.edu/newsltr/v7n3/sa-7.htm)

Harrington, R., M. Hullé, and M. Plantegenest. 1998. Monitoring and Forecasting, pp. 515-536. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Henneberry, T. J., and R. M. Faust [eds.]. 1999. Silverleaf whitefly supplement to the 5-year national research, action, and technology transfer plan (1997-2001), 31 January 2 February 1999, Albuquerque, NM. USDA-ARS 1999-01.

Hoddle, M. S., R. G. Van Driesche, and J. P. Sanderson. 1998. Biology and the use of the whitefly parasitoid *Encarsia formosa*. Annu. Rev. Entomol.34: 645 – 669.

Hoelmer, K. A. 1998. Selectivity of whitefly traps in cotton for Eretmocerus eremicus (Hymneoptera: Aphelinidae), a native of *Bemisia argentifolii* (Homoptera: Aleyrodidae). Environ. Entomol. 27: 1039-1044.

Hoelmer, K. A., L. S. Osborne, and R. K. Yokomi. 1993. Reproduction and feeding behavior of *Delpastus pusillus* (Coleoptera: Coccinellidae) a predator of *Bemisia tabaci* (Homoptera: Alyerodidae). J. Econ. Entomol. 86:322-329.

Hoffmann, M. P. and A. C. Frodsham. 1993. Natural enemies of vegetable insect pests, pp. 63. *In*: A Cornell Cooperative Extension, Cornell University, Ithaca, NY.

Hooks, C. R. R., H. R. Valenzuela, J. DeFrank. 1998. Incidence of pests and arthropod natural enemies in zucchini grown with living mulches. Agric. Ecosyst. Environ. 69: 217-231.

Hooks, C. R. R. and M. W. Johnson. 2002. Lepidopteran pest populations and crop yields in row intercropped broccoli. Agric.Forest Entomol. 4: 117 – 125.

International Crops Research Institute for the Semi-Arid Tropics (ICRISAT). June 2007. (http://www.icrisat.org/PearlMillet/PearlMillet.htm.)

Irwin, M. E., G. E. Kampmeier, and W. W. Weisser. 1998. Aphid movement: process and consequences, pp. 153-186. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Kalule, T., and D. J. Wright. 2005. Effect of cultivars with varying levels of resistance to aphids on developmental time, sex ration, size and longevity of the parasitoid, *Aphidius colemani*. Biocontrol 50: 235-246.

Khan, M. A., K. B. Marwat, G. Hassan, and Z. Hussain.2005. Bioherbicidal effects of tree extracts on seed germination and growths of crops and weeds. Pal. J. Weed. Sci. Res. 11: 89-94.

Kemble, J. M., E. J. Sikora, M. G. Patterson, G. W. Zehnder and E. Bauske. 2005. Guide to commercial summer squash production, ANR-1014. Alabama Cooperative Extension System, Alabama A&M University, Normal, AL.

Kring, J.B. 1972. Flight behavior of aphids. Annu. Rev. Entomol.17: 461–492.

Kromp, B. 1999. Carabid beetles in sustainable agriculture: a review on efficacy, cultivation impacts and enhancement. Agric. Ecosyst. Environ. 74: 187 – 228.

Kucharek, T., and D. Purcifull. 2001. Aphid-transmitted viruses of cucurbits in Florida, Circ-1184. Institute of Food and Agricultural Sciences. University of Florida, Gainesville, FL.

Laub, C. A., and Luna, J. M., 1992. Winter cover crop suppression practices and natural enemies of armyworm (Lepidoptera: Noctuidae) in no-till corn. Environ. Entomol. 12: 41–49.

Liburd, O. E., S. Polavarapu, S. R. Alm, and R. A. Casagrande. 2000. Effect of trap size, placement, and age on captures of blueberry maggot flies (Diptera: Tephritidae). J. Econ. Entomol. 93: 1452-1458.

Liburd O. E., and D. L. Frank. 2007. A review of synthetic and living mulches for control of homopteran pests (aphids and silverleaf whiteflies) and diseases in vegetables. *In:* G. Saxena and K.G. Mukerji [eds.], Management of Nematode and Insect Borne Diseases. The Haworth Press, Inc., New York, NY.

Liburd,O. E., and T. W. Nyoike. 2008. Biology and management of aphids in sustainable field production of cucurbits, ENY-847. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Lövei, G. L. and K. Sunderland. 1996. Ecology and behavior of ground beetles (Coleopteran: Carabidae). Annu. Rev. Entomol.41: 231- 256.

Lu, Y., B. Watkins, J. R. Teasdale, and A. A. Abdul-Baki. 2000. Cover crops in sustainable food production. Food Rev.Int.16: 121 – 157.

Lyon, W. F. and F. F. Purrington. Ground Beetles. Extension Fact Sheet. Ohio State University, Columbus, OH.

Mäder, P., A. Fließbach, D. Dubois, L. Gunst, P. Fried, and U. Niggli. 2002. Soil fertility and biodiversity in organic farming. Science 296: 1694-1697.

Martin, H. 2006. Introduction to Organic farming. Ministry of Agriculture Food and Rural Affairs fact sheet 100/10.

McAuslane H. J. 2007. Sweetpotato whitefly B biotype or silverleaf whitefly Featured Creatures, EENY-12. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

McAuslane, H. J., S. E. Webb, and G. W. Elmstrom. 1996. Resistance in germplasm of *Cucurbita pepo* to silverleaf, a disorder associated with *Bemisia argentifolii* (Homoptera: Aleyrodidae). Fla. Entomol. 79: 206 – 221.

McAuslane, H. J. 2005. Sweetpotato Whitefly B Biotype of Silverleaf Whitefly, *Bemisia tabaci* (Gennadius) or *Bemisia argentifolii* Bellows and Perring (Insecta: Homoptera: Aleyrodidae), EENY-129. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Meagher, R. L., and P. J. Landlot. 2008. Attractiveness of binary blends of floral odorant compounds to moths in Florida, USA (Lepidoptera: Noctuidae, Pyralidae). Entomol. Exp. Appl. (in press).

Metcalf, R. L., and R. A. Metcalf. 1993. Insects injurious to vegetable gardens and truck crops, pp.14.16. *In*: F. J. Kotowski (Jr.) [ed.], Destructive and useful insects: their habits and control, McGraw Hill, NY.

Molinar, R., J. Aguiar, M. Gaskell, and K. Mayberry. 1999. Summer squash production in California. Vegetable Research and Production Center. 76-SR.

Moschetti, R. 2003. Aphid parasites, *Aphidius* spp. Technical Bulletin: Greenhouse Management Series. (http://www.ipmofalaska.com/files/aphidius.html).

Mossler, M. A., and O. N. Nesheim. 2001. Florida Crop/Pest Management Profile: Squash, CIR 1265. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Mundy, C.A., L. J. Allen-Williams, N. Underwood, S. Warrington. 2000. Prey selection and foraging behaviour by *Pterostichus cupreus* L. (Col., Carabidae) under laboratory conditions. J Appl. Entomol.124: 349-358.

Myers, R. L. 2002. Pearl millet. National Edition Alternative Crop Guide. (http://www.jeffersoninstitute.org/pdf/pearlmillet_crop_guide.pdf)

Narwal, S. S. 2006. Allelopathy in ecological sustainable agriculture, pp. 537-564. *In* M. J. Reigosa, N. Pedrol, and L. Gonzalez [eds.], Allelopathy: A Physiological process with ecological implications, Springer Netherlands, Dordrecht, The Netherlands.

National Agricultural Statistics Service (NASS). 2008. USDA, Washington, D.C.

Nault, L. R. 1997. Arthropod transmission of plant viruses: a new synthesis. Ann. Entomol. Soc. Am. 90: 521-541.

Nebreda, M., A. Moreno, N. Perez, I. Palacios, V. Seco-Fernandez, A. Fereres. 2004. Activity of aphids associated with lettuce and broccoli in Spain and their efficiency as vectors of lettuce mosaic virus. Virus Res. 100: 83 – 88.

Nebreda, M., G. Nombela, and M. Muniz. 2005. Comparative host suitability of some Brassica cultivars for the whitefly, *Aleyrodes proletella* (Homoptera: Aleyrodidae). Environ. Entomol.34: 205 – 209.

Nentwig, W. 1998. Weedy plant species and their beneficial arthropods: potential for manipulation in field crops, pp. 49-72. *In* C. H. Pickett and R. L. Bugg [eds.], Enhancing biological control: habitat management to promote natural enemies of agricultural pests. University of California Press, Berkeley, CA.

Oberholtzer, L., C. Dimitri, and C. Greene. 2005. Price premiums hold on as U.S. organic produce market expands. U.S. Dep. Agric., Economic Research Service, Washington, D.C.

Obrycki J. J., Kring T. J. 1998. Predaceous coccinellidae in biological control. Annu. Rev. Entomol.43: 295–321.

Palumbo, J. C., N. C. Toscano, M. J. Blua, and H. A. Yoshida 2000. Impact of *Bemisia* whiteflies (Homoptera: Aleyrodidae) on alfalfa growth, forage yield, and quality. J. Econ. Entomol. 93: 1688-1694.

Parajulee, M. N., R. Montadon, and J. E. Slosser. 1997. Relay intercropping to enhance abundance of insect predators of cotton aphid (*Aphis gossypii* Glover) in Texas cotton. Int. J. Pest Manage. 43: 227 – 232.

Páre, P. W., and J. H. Tumlinson. 1999. Plant volatiles as a defense against insect herbivores. Plant Physiol. 121: 325-331.

Pedigo, L. P. 2002. Insect Classification, pp. 118. *In* S. Helba, D. Yarnell, L. Dalberg, and M. Carnis [eds.], Entomology and Pest Management. Prentice-Hall, Englewood Cliffs, NJ.

Pedigo, L. P. 2002. Management with natural enemies and other biological agents, pp. 319-321. *In* S. Helba, D. Yarnell, L. Dalberg, and M. Carnis [eds.], Entomology and Pest Management. Prentice-Hall, Englewood Cliffs, NJ.

Peet, M. 1996. Cover crops and living mulches, pp. 1- 174. *In*: Sustainable vegetable practices in the South. Focus Publishing, Newburyport MA.

Pettersson, J., W. F. Tjallingii, and J. Hardie. 1998. Host-plant selection and feeding, pp. 87-114. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Phatak, S. C. 1998. Managing pests with cover crops, pp. 25-33. *In* G. Bowman, C. Shirley, and C. Cramer [eds.], Managing cover crops profitably. National Agricultural Library, Beltsville, MD.

Pickett, J. A. and R. T. Glinwood. 1998. Chemical Ecology, pp. 235-260. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Powell, G., C. R. Tosh, and J. Hardie. 2006. Host plant selection by aphids: Behavioral, evolutionary, and applied perspectives. Annu. Rev. Entomol.51: 309-330.

Powell, W. and J. K. Pell. 1998. Biological Control, pp. 469-514. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Ragsdale, J. 2006. Influence of cover crops on nutrient availability in a sweet potato cropping system in South Florida, pp.2-4, MS Thesis. Department of Horticultural Sciences, University of Florida, Gainesville, FL.

Rich, J., D. Wright, J. Marois, and D. Sprenkel. 2003. Selected legumes used as summer cover crops, ENY-688. Institute of Food and Agricultural Sciences. University of Florida, Gainesville, FL.

Roberts, P., and K. Pernezy. 2001. Disease control for squash in Florida, Number 63. Institute of Food and Agricultural Sciences. University of Florida, Gainesville, FL.

Robertson, T., V. Benson, J . R. Williams, C. H. Lander, and D. L. Schertz 1991. Cover crops and control of arthropod pests of agriculture, pp.117-120. *In* Cover crops for clean water: Proceedings of an international conference West Tennessee Experiment Station, 9-11 April 1991, Jackson TN.

Robinson, R. R., J. H. Young, and R. D. Morrison. 1972. Stripcropping effects on abundance of predatory and harmful cotton insects in Oklahoma. Environ. Entomol. 1: 145-149.

Rodriguez, J. C. 2008, personal communication.

Rodrigues, S. M. M., and V. H. P. Bueno. 2001. Parasitism rates of *Lysiphlebus testaceipes* (Cresson) (Hym.:Aphidiidae) on *Schizaphis graminum* (Rond.) and *Aphis gossypi* Glover (Hem.: Aphididae). Neo. Entomol. 30: 625-629.

Root, R. B. 1973. Organization of a plant-arthropod association in simple and diverse habitats: the fauna of collards (*Brassica oleracea*). Ecol. Monog. 43: 95-120.

Sarrantonio, M. 1998. Selecting the best cover crops for your farm, pp. 12-15. *In* G. Bowman, C. Shirley, and C. Cramer, Managing Cover Crops Profitably. National Agricultural Library, Beltsville, MD.

SAS Institute. 2003. The SAS/STAT Software: changes and enhancements through release 9.1. SAS Institute, Cary, NC.

Schooley, T. N. 2005. Crop profile for summer squash in Virginia. General Production Facts and Region Sheet. (http://www.ipmcenters.org/cropprofiles/docs/VAsquash.html)

Shannag, H. K., and W. M. Obeidat. 2006. Voracity and conversion efficacy by larvae of *Coccinella septempunctata* L. (Coleoptera: Coccinellidae) on *Aphis fabae* Scop. (Homoptera: Aphididae) reared on two faba bean cultivars with different levels of resistance. Appl. Entomol. Zool. 41: 521-527.

Shaver, T. N and J. R. Raulston. 1971. A soybean-wheat germ diet for rearing the tobacco budworm. Ann. Entomol. Soc. Am. 64: 1077-1079.

Simonne, E.H., C. M. Hutchinson, M. D. Dukes, G. J. Hochmuth and R. C. Hochmuth. 2003. Update and Outlook for 2003 of Florida's BMP program for vegetable crops, HS 916. Institute of Food and Agricultural Sciences. University of Florida, Gainesville, FL.

Simmons, A. M. 2002. Settling of crawlers of *Bemisia tabaci* (Homoptera: Aleyrodidae) on five vegetable hosts. Annu. Entomol. Soc. Am. 95: 464 - 468.

Simmons, A. M., S. Abd-Rabou, and G. S. Mccutcheon. 2002. Incidence of parasitoids and parasitism of *Bemisia tabaci* (Homoptera: Aleyrodidae) in numerous crops. Environ. Entomol. 31: 1030 - 1036.

Sithanantham, S., T. H. Abera, J. Baumgärtner, S. A. Hassan, B. Löhr, J. C. Monje, W. A. Overholt, A. V. N. Paul, F. Hao Wan, and C. P. W. Zebitz. 2001. Egg parasitoids for augmentative biological control of lepidopteran vegetable pests in Africa: Research status and needs. Insect Sci. Appl. 21: 189 – 205.

Smith, C. 2008. Federal Register. Department of Agriculture Animal and Plant Health Inspection Service Docket No. APHIS-2007-0144. (http://edocket.access.gpo.gov/2008/E8-10920.htm)

Smith, H. A. 1999. Intercropping and whitefly (Homoptera: Aleyrodidae) management, PhD Dissertation, pp. 2-8, Department of Entomology and Nematology, University of Florida, Gainesville, FL.

Smith, H. A. and W. E. Chaney. 2007. A survey of syrphid predators of *Nasonovia ribisnigri* in organic lettuce on the central coast of California. J. Econ. Entomol.100: 39 – 48.

Stapleton, J. J., and C. G. Summers. 2002. Reflective mulches for management of aphids and aphid-borne virus diseases in late season cantaloupe (*Cucumis melo* L. var. Cantalupensis). Crop Prot. 21: 891-898.

Sullivan, P. 2003. Overview of cover crops and green manures, pp. 16., Fundamentals of Sustainable Agriculture. Appropriate Technology Transfer for Rural Area (ATTRA) Publicaton IP107.

Symondson, W.O. C., K. D. Sunderland, and M. H. Greenstone. 2002. Can generalist predators be effective biocontrol agents? Annu. Rev. Entomol. 47: 561-594.

Taylor, A. J., C. B. Müller, and H. C. J. Godfray. 1998. Effect of aphid predators on oviposition behavior of aphid parasitoids. J. Insect Behav.11: 297 – 302.

Tillman, G. H. Schomberg, S. Phatak, B. Mullins, S. Lachnicht, P. Timper, D. Olson. 2004. Influence of cover crops on insect pests and predators in conservation tillage cotton. J. Econ. Entomol. 97: 1217-1232.

Torres, A. F., H. P. Bueno, M. V. Sampaio, and B. F. De Conti. 2007. Fertility Life Table of *Aphidius colemani* Viereck (Hymenoptera: Braconidae, Aphidiinae) on *Aphis gossypii* Glover (Hemiptera: Aphididae). Neotrop. Entomol. 36: 532-536.

Treadwell, D. D. 2007. Introduction to organic vegetable production, pp. 135-148 *In*. S. M. Olsen and E. Simmone [eds.], Vegetable production handbook for Florida 2006-2007 IFAS Extension. University of Florida, Gainesville, FL.

Valenzuela, H., and J. Smith. 2002. Sorghum-sudangrass hybrids. Sustainable Agriculture Green Manure Crops, SA-GM-10. University of Hawaii at Manoa, Honolulu, HI.

Vásquez, G. M., D. B. Orr, and J. R. Baker. 2006. Efficacy assessment of *Aphidius colemani* (Hymenoptera: Braconidae) for suppression of *Aphis gossypii* (Homodoptera: Aphididae) in greenhouse-grown chrysanthemum. J. Econ. Entomol. 99: 1104-1111.

Völkl, W., M. Mackauer, J. K. Pell, and J. Brodeur. 1998. Predators, parasitoids, and pathogens, pp. 187-234. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Wang, K. H., an d R. McSorley. 2004. Management of nematodes and soil fertility with SH cover crop, ENY-717. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Wang, Q. Y. Li, W. Klassen, and Z. Handoo. 2006. Influence of cover crops and soil amendments on okra (*Abelmoschus esculentus* L.) production and soil nematodes. Renew. Agr. Food Syst. 22: 41-53.

Webb, S. E. 1998. IPM case studies: cucurbits, pp. 639-650. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Webb, S. 2006. Insect management for Cucurbits (Cucumber, Squash, Cantaloupe and Watermelon), IFAS Extension ENY-460. University of Florida, Gainesville, FL.

Wei, J. N., B. B. Bai, T. S. Yin, Y. Wang, L. H. Zhao, R. P. Kuang, and R. J. Xiang. 2005. Development and use of parasitoids (Hymenoptera: Aphidiidae and Aphelinidae) for biological control of aphids in China. Biocontrol Sci.Techn.15: 533 – 551.

Weinzierl, R., T. Henn., P. G. Koehler, and C. L. Tucker. 2005. Insect Attractants and traps, ENY-27. Institute of Food and Agricultural Sciences, University of Florida, Gainesville, FL.

Williams, I. S., and A. F. G. Dixon. 1998. Life cycles and polymorphism, pp. 69-86. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Woiwood, I. P. and Hanski, I. J. 1992. Patterns of density dependence in moths and aphids. J. Anim. Ecol. 60: 749–766.

Wratten, S. D., G. M. Gurr, J. M. Tylianakis, and K. A. Robinson. 1998. Cultural Control, pp. 423-446. *In* H. F. van Emden and R. Harrington [eds.], Aphids as Crop Pests. CAB International, Cromwell Press, Trowbridge, UK.

Zhang, W. Q. and S. A. Hassan. 2003. Use of the parasitoid *Diaeretiella rapae* (McIntoch) to control the cabbage aphid *Brevicoryne brassicae* (L.). J. Appl. Entomol.127: 522 – 526.

BIOGRAPHICAL SKETCH

Corraine Athol Scott was born in Manchester, Jamaica in 1983. She graduated from Glenmuir High School, May Pen, Clarendon, in July 2001. After graduation, she earned a Presidential scholarship to attend Randolph Macon Woman's College (now Randolph College) in Lynchburg, VA where she earned her BS in biology with minors in chemistry and sociology/anthropology in May 2005. After graduation, Corraine went to Virginia Commonwealth University, where she spent a year as an adjunct biology lecturer for introductory biology labs and was the resident director for a freshman residence hall while taking graduate courses. Corraine transferred to the University of Florida where she pursued her MS degree in entomology in fall 2006. After completing her degree requirements, Corraine will continue her graduate studies as a PhD entomology student studying various semiochemical strategies to manage pepper weevil.