PERENNIAL LEGUME GROUND COVERS AND AUGMENTATIVE RELEASES OF *TRICHOGRAMMA* FOR LOW-INPUT PECAN MANAGEMENT

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PREFACE

The purpose of this study was to evaluate 'Kenland' red clover and 'Lousiana S-1' white clover for a low-input pecan management system in supplying N and enhancing arthropod populations that may control pecan aphids. This study also includes the evaluation of *Trichogramma pretiosum* release to suppress damage caused by pecan nut casebearer by parasitizing eggs.

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CHAPTER I

INTRODUCTION

Pecan (*Carya illinoinensis* [Wangenhaim] C. Koch, Juglandaceae) is one of the most popular commercial horticultural crops of all the nuts native to North America (Briston, 1974). This premium quality nut crop covers about 250,000 ha in the U.S. with an annual production of 135 million kg (Harris, 1990). The production of pecan is centered in the southeastern, south central and southwestern regions of the U.S., stretching from North Carolina to southern California, where the average growing season has at least 200 frost free days. The lead pecan producing states are Georgia (averaging more than 30 percent of the annual production), Louisiana, Texas, New Mexico, Alabama and Oklahoma (Charlet and Henneberry, 1992). Pecan is also commercially grown in other parts of the world including Canada, Mexico, Australia, Brazil, Israel and South Africa (Harris, 1983).

Pecan is classified as a hickory (*Carya*) and is a member of the walnut family (Juglandaceae) (Briston, 1974). In its most favorable environment, the majestic tree can grow to a height of 46 m with a trunk diameter of 1.5 to 2 m. In North America pecan production is divided into native and cultivar orchards. The native orchards consist of wild trees that have been cleared of competing vegetation and thinned periodically to the correct spacing for optimum production (Harris, 1983). These trees generally grow along river banks, creeks and dry stream beds. From 1970 through 1990, native pecans accounted for 38% of production in the U.S. and 92% in Oklahoma (Napper, 1991). Oklahoma and Texas possess the largest number of native trees of all the pecan producing states. About 70% of the native pecan orchards in Oklahoma are combined with cattle grazing (Mitchell and Wright, 1991). The second type of orchard is planted to improved cultivars.

Pecan management practices in both native and cultivar orchards are highly variable. Most pecan orchards receive heavy applications of fertilizer (mainly N), pesticides, irrigation water and mowing (Bugg et al., 1991a). Nitrogen is most frequently applied in February or March, with an annual application rate of more than 100 kg ha^{-1} (Harris, 1983). The native orchards are mostly located on alluvial flood plains where they can compete successfully for light and space amid the other plant growth. In other regions pecans usually occupy deep well-drained soils. Typical ground cover management in pecan orchards consists of closely mowed permanent sods such as bermudagrass (*Cynodon dactylon* [L.] Pers.) which reduces the growth and yield of pecan trees by competing for moisture and nutrients (Gossard and Hammer, 1957; Ware and Johnson, 1958). One of the important operations in pecan management is insect control. The monocultural conditions of pecan orchards promote the development of many induced pecan pests. The most important insect pests associated with pecan include pecan nut casebearer (*Acrobasis nuxvorella* [Neunzig]), pecan weevil (*Curculio caryae* [Horn]), hickory shuckworm (*Cydia caryana* [Fitch]), and aphids (Homoptera: Aphididae). The first three are destructive nut pests of pecan that can destroy a pecan crop. In many pecan producing areas insect and mite complexes present a continuing threat all season long to pecan production (Harris et al., 1992). Growers use intense pest control programs with eight to 10 insecticide applications per year in the southeastern U.S. (Tedders, 1983; Bugg et al., 1991a), and five to seven insecticide applications per year in the south central U.S. In the east, pecan growers use from six to eight applications of aphidicides per year (Wood et al., 1983). These pest control programs are expensive and decrease the profit of pecan growers. Moreover, excessive pesticide use has resulted in the development of pest resistance and outbreak of secondary pests creating the need for more pesticide use (Ball, 1981; Mizell, 1991).

Many insecticides have a negative impact in non-target organisms such as beneficial insects and mites that may be important in the natural control of primary and secondary pests (Dutcher, 1983; Dutcher and Payne, 1983; Tedders, 1983; Mizell, 1990). In response to the adverse effects of excessive pesticide use and the expense of fertilizers, lowering chemical inputs has been emphasized.

Low-input Sustainable Systems Approach

In recent years, concern over environmental and health risks from agrichemicals has increased (Deberkow and Reichelderfer, 1988). Intensive management practices with heavy use of chemicals have created a variety of economic, environmental and ecological problems. Some of the recognized problems resulting from excessive use of pesticides and fertilizers are the increased contamination of soils and water systems (Myers et al., 1985; Hallberg, 1986; Edwards, 1992).

Groundwater accounts for 95% of U.S. freshwater reserves (Anderson et al., 1985). Groundwater supplies about 50% of drinking water and 40% of irrigation needs and is the major drinking water source for 85% of the rural population (Saliba, 1985). Studies monitoring ground water quality reveal disturbing trends regarding the presence of several pesticides and nutrients (Holden, 1986; Hallberg, 1987; Nielsen and Lee, 1987). Nitrate is considered a major contaminant of groundwater and is widespread in the U.S. (Benbrook, 1989). The causes and related environmental effects of NO_3-N pollution have been discussed in a number of articles (Greenwood, 1990; USDA, 1991). Nitrate is very mobile in the underground environment and moves rapidly with deep percolation through the vadose zone to underlying groundwater. In several local areas NO_3 concentration is very high. The basis for much of this concern is the potential effects of NO_3-N on the health of human infants and animals. Infants younger than three months of age that consume water contaminated with NO_3-N are susceptible to methemoglobinemia, also known as "blue baby syndrome." In addition to causing acute toxicity in infants, nitrates form nitrous compounds that are known to be potent animal carcinogens and are suspected of causing cancer in humans (Madden, 1988).

In view of these facts lowering chemical inputs in agricultural production has been emphasized. Low-input sustainable systems avoid the use of synthetically manufactured fertilizers, pesticides and growth regulators (Edwards, 1992; Schaller, 1990). A low input sustainable agricultural system is a combination and sequence of low-input farming methods or technologies integrated into a whole-farm managerial plan. It encompasses a wide array of approaches that reduce the farmers dependance on certain kinds of purchased environmental hazards, and ensures a more sustainable agriculture for generations to come (Lockeretz and Wernick, 1983; Madden, 1988). Low-input systems approaches range from the release of exotic natural enemies to rather commonplace methods such as controlling weeds by mechanical cultivation and crop rotations. Also included in this category are various integrated pest management strategies such as biological control of pests through enhancement of natural enemies, use of legume crops in place of some purchased fertilizers, and application of livestock manures and compost (Edwards, 1989; Madden, 1988). The widespread adoption of this alternative farming method is expected to lead to major changes in the agricultural practices. Much of the current research on low-input agriculture examines possible barriers to this adoption, including yield reduction, higher labor requirements, higher variable costs, and the limited availability of alternative sources of N.

In line with the above considerations, interest in the development of low-input management for pecan orchards has also increased. Lower input systems may offer the best alternative by increasing or maintaining the net income of the pecan grower and also protecting the environment. The native pecan agroecosystem is considered suitable for the low-input management approach (Reid and Eikenbary, 1990). Lower inputs of pesticides and N fertilizer in pecan orchards may be achieved by integrated management systems such as using legume cover crops, releasing exotic natural enemies or enhancing native natural enemies (Blackmon, 1948; Bugg et al., 1991a). Legumes may have several potential benefits for pecans that include decreased dependence on commercial N sources, reduced potential for NO_3 pollution, soil improvement from incorporated organic matter and may also assist in biological control of insect pests.

Legumes as a Nitrogen Source

The use of legumes in agricultural practices appears as a thread of archeological and written record since the emergence of evidence for the management of plants for food (Delwiche, 1978). They have served as a primary source of N for many cropping systems (Power, 1987). Heichel (1987a) reports that on a global basis agriculturally important legumes provide 80 million metric tons of N per year, compared with 50 million tons of N fertilizer manufactured by the Haber-Bosch process. In addition to supplying N, legumes can decrease soil erosion, especially during the winter and early spring (Finch and Sharp, 1981; White et al., 1981; Langdale and Leonard, 1983; Hall et al., 1984; Power, 1987b); may increase water infiltration (Wilson et al., 1982; Frye et al., 1988), soil aeration (White et al., 1981) and may improve soil organic activity, water storage (Smith et al., 1987; Bolton et al., 1985), weed control and overall productivity (Frye et al., 1988).

Legumes are recognized for their capacity to extract N from the air and change it into a useable form for plants through a symbiotic relationship with a bacteria called *Rhizobium* (Allen, 1962; Chapman and Carter, 1976). The total N in a legume plant is contained both in the roots and above ground portion. Less than one third of the total N content of most legumes is contained in the roots (Mitchell and Teel, 1977). The ability of legumes to fix N can be greatly reduced by the addition of N or the presence of readily available N in the soil. If available soil N is present, the legumes use it in preference to atmospheric N (Allison, 1957; Weber, 1966; Erdman, 1967; Atkins, 1986).

Legumes supply a significant amount of biologically fixed N to succeeding crops (Evans and Sturkie, 1974; Michell and Teel, 1977). They influence plant nutrient availability by increasing soil organic C, total N, exchangeable cations, and pH (Hargrove, 1986; Wilson et al., 1982). They also enhance nutrient retention in the soil (Ebelhar et al., 1984). Essential plant nutrients are incorporated into biomass during the winter, then decompose and become available to the summer crop. The return of micronutrients and the addition of synthesized bioregulators (vitamins and plant hormones) to the soil surface may influence the growth and development of subsequent crops. Legume species and amount of residue will influence soil temperature reduction (Finch and Sharp, 1981). Soil aggregate stability is maintained with winter legume rotations, and soil porosity and permeability are increased. The deep penetration of the legume rooting system allows exudates to exist at greater soil depths upon decomposition. Metabolites of these exudates may serve as solubilizing and or chelating agents of plant nutrients fixed in the unavailable form. Legumes may reduce residual soil nitrates (Zachariassen and Power, 1991), and may also accumulate higher concentrations of P, K, Ca and Mg than grass covers in the upper soil layers (Groffman et al., 1987).

Numerous researchers have reported that winter legume cover crops can be significant sources of N for subsequent non-leguminous crops, replacing significant amounts of fertilizer N (Michell and Teel, 1977; Touchton et al., 1982; Ebelhar et al., 1984; Touchton et al., 1984; Hargrove, 1986; Neely et al., 1987). The contribution of legumes to succeeding non-legume crops can be variable and is dependant on many factors, including the N status of the soil, dry matter yield and N concentration of the legume covers (Fribourgh and Johnson, 1955). Research on 'Amclo' arrow leaf clover (*Trifolium vesiculosum* Salli) and 'Cahaba' white vetch (*Vicia sativa* L.x v. Cordata), indicated that these legumes contained 182 and 138 kg.ha^{-1} N respectively. The clover contained higher total N due to greater biomass production (White et al., 1981), and the N concentration of a legume is related more to plant maturity (Fleming et al, 1981). As clover matures N concentration decreases (Akin and Robinson, 1982). When it reaches maturity most of the protein is translocated from the vegetative plant parts into the developing seeds (Allison, 1957; Atkins, 1986). Location and season also affect total dry matter production of the legume crop. Leguminous cover crops left on the soil surface typically contribute on about one-half as much N as those disked under (Zomer and Bugg, 1989). If the legume crop is incorporated into the soil, the N contribution to the succeeding crop is enhanced, but killing it for a surface mulch improves the soil and water conservation value (Frye et al., 1988). A good winter legume cover turned under will increase yield of succeeding crops similar to 57-111 kg ha^{-1} N (Rogers and Giddens, 1957). Studies evaluating the fate of fertilizer N from legume residues decomposing under field conditions concluded that : (1) < 30% of legume N was recovered by a subsequent non-legume crop (2) large amounts of legume were retained in soil, mostly in organic forms : (3) total recovery of legume N in crops and soils after one year averaged 70 to 90%; and (4) < 5% of legume N from the original application was recovered by a second non-legume crop (Ladd et al., 1983; Muller and Sundman, 1988; Harris and Hesterman, 1990; Ta and Farris, 1990). Legume N inputs may contribute more than fertilizer N to long-term soil fertility through buildup of organic N reserves (Ladd et al., 1981; Frye et al., 1988; Jonzen et al., 1990). Harris et al. (1994) compared the fate of applied legume and fertilizer N in a long-term cropping system. He reported that more fertilizer than legume N was recovered by crops (40 vs 17% of input), more legume than fertilizer N was retained in the soil (47 vs 17% of input), and similar amounts of N from both sources were lost from the cropping systems (39% of input) over a two year period. Soil microbial biomass was larger in the legume based system. The greater soil N supplying capacity in the legume based system as compared to fertilizer based system was considered to be the impact of a larger microbial biomass.

The value of legumes in the production of pecans was recognized as early as 1920 (White et al., 1981). They were commonly grown in pecan orchards as cover and green manure crops before widespread access to inexpensive synthetic N fertilizers prior to the 1960's (Tedders, 1983; White et al., 1981). The earliest studies with legumes for pecan orchards evaluated a broad range of summer and winter legume types for N supplying ability (Rouse, 1926; Blackmon, 1948). Winter legumes were superior to summer legumes because they contributed N to the soil in the spring when the pecan tree needed it most (Woodard, 1923). After the 1960's, the use of legume cover crops in pecan orchards declined, but recently because of higher N costs, and environmental and social concerns, interest in using legumes has increased (Mizell, 1991).

Legume species and cultivars vary considerably in their N fixing capacity (Ham, 1978; Heichel et al., 1981). Erdman (1967) showed estimates of N fixed by several legumes ranging from 57 to 207 kg ha^{-1} N. Crimson clover added 57-78 kg ha^{-1} N in pecan trees. The variables affecting the quantity of N fixed include not only legume species, but also soil type and texture, pH, soil NO_3-N concentration, temperature, water regimes and availability of other nutrients (Power, 1987). Legume N production can be affected by planting time (White et al ., 1981) and legume harvest management (Groya and Sheaffer, 1985). Legumes should be planted in early fall in pecan orchards to obtain satisfactory biomass and N production. Legume cover crops frequently increase leaf N concentrations of the pecan trees within one growing season, and the pecan leaf N using arrow leaf clover and 'Cahaba' white vetch were equivalent to that of trees receiving 111 kg ha^{-1} N (White et al., 1981; Bugg et al., 1991a). Additional increases in N may occur during subsequent seasons.

Recently Smith et al. (1994) evaluated several cool season annual and perennial legume cover crops in pecan orchards. Of the perennial legumes evaluated, 'Kenland ' red clover (*Trifolium pratens* L.) was one of the highest in N production, but the stands began declining during the third year. 'Louisiana S-1' white clover *(Trifolium repens* L.) produced a large amount N with the stands improving each year. 'Louisiana S-1' is aggressive and becomes relatively quiescent during the summer, thus minimizing competition for water, and providing an excellent harvest surface during the fall. Nitrogen in the tops of these perennial legumes ranged from 108 to 179 kg ha^{-1} following harvests in June and September. Results from other studies indicate that white clover grown in association with grasses, can fix over 250 kg ha^{-1} N annually (Halliday and Pate, 1976).

White clover also has a rapid and dramatic effect on the development of improved soil structure (Mytton et al., 1993). Red clover is the most widely grown of all the true clovers in the U.S. and is extensively used in the upper midwest for hay, pasture, and soil improvement (Taylor, 1985). It was reported that in intensive harvest management systems the total season fixed N in red clover herbage ranged from 79 to 272 kg ha^{-1} (Taylor, 1985). Butler et al. (1959) found that defoliation and shading of red clover caused a severe reduction in nodule number, while nodules of white clover were not affected. White clover has prostrate and stoloniferous habit of growth which ensure rapid recovery and spread (Jones, 1992).

Some of the drawbacks of using legume cover crops include competition with trees for water and nutrients (Blackmon, 1948; Hardy, 1939) and possible enhancement of a particular pest. Moreover, legumes may contribute to NO_3 leaching into the groundwater. Leaching losses of NO_3 -N were reported to be higher under clover plots than under fertilized nitrogen plots, although the magnitude of the N lost to leaching was small (Groffman et al., 1987). Nitrate leaching is greatest during winter and early spring months (Owens, 1990). Nevertheless, some of these drawbacks can be reduced or eliminated with careful management and control practices.

Legumes as a Biological Control Agent

One of the most serious pests that damages pecans throughout the season is the foliar feeding aphids (Mizell et al., 1990). Aphids may cause considerable damage to the pecan tree and its foliage. Apparent yield reductions are observed after two or three seasons of poor aphid control. Aphids have high reproductive rates and short life cycles that enhance their genetic selection, resulting in tolerance or resistance to frequently used insecticides (Tedders, 1986).

Three aphid species attack pecan: yellow pecan aphids (*Monelliopsis pecanis* [Bissell]), blackmargined aphid (*Monellia carella* [Fitch]), and black pecan aphid (*Melanocallis caryaefoliae* [Davis]) (Tedders, 1978; Tedders et al., 1982). Blackmargined and yellow pecan aphids cause similar leaf injury, and are similar in their biology and control. They are commonly called the yellow aphid complex (Payne et al., 1979). Both are equally damaging and can limit pecan production (Neel et al., 1985). Black pecan aphids are much more destructive than the yellow aphid complex (Moznette, 1934). Feeding by black aphids causes necrosis and eventually leaf loss. Lakin (1972) investigated the damage to pecan foliage by black pecan aphids and the biology, life history and importance.

Records of the seasonal dynamics of the yellow aphid complex in Georgia (Tedders, 1978; Dutcher, 1983) show that two discrete infestation periods occur each season. Early infestations occur in May and June between the pollination and fruit development phenological stages of the tree. Mid-season aphid densities during fruit enlargement and shell hardening are typically very low (Polles and Mellunix, 1977; Leser, 1981; Edelson and Estes, 1983). Vigorous population growth from shell hardening to harvest produces the late season peak in aphid population levels which is usually the most severe infestation. Black aphids typically infest the pecan foliage from July until harvest and a severe infestation can lead to defoliation.

Some researchers suggest that these aphid population peaks coincide with an abundance of highly nutritious and young foliage in early and late season, while the mid-season aphid population crash coincides with the presence of less nutritious mature leaves (Smith and Severson, 1992). Pecan aphids feed on the vascular system of the leaves and damage the leaf veins at the site of feeding by clogging phloem and inducing chlorosis and necrosis of the surrounding mesophyll cells (Tedders and Thompson, 1981; Wood et al., 1985). Large populations can reduce leaf chlorophyll and leaf area (Tedders et al., 1982; Wood and Tedders, 1982; Wood et al., 1985; Tedders and Wood, 1985; Wood et al., 1987), cause premature leaflet abscission (Lakin, 1972; Tedders et al., 1982), irreversibly suppress leaf photosynthesis by up to 75% (Wood and Tedders, 1982; Wood et al., 1988), and deplete leaf carbohydrate and proteins (Wood and Tedders, 1982). All three species reduce growth of roots and stems (Tedders et al., 1982), decrease tree vigor (Dutcher, 1985), reduce yield (Dutcher et al., 1984; Tedders and Wood, 1985), and nut quality (Wood et al., 1987; Tedders and Wood, 1987).

The most noticeable damage of pecan aphids is their deposits of honey dew on the pecan foliage (Tedders and Smith, 1976; Smith and Tedders, 1980). Honey dew is a sticky fluid excreted by aphids that supports the growth of sooty mold fungi. These fungi reduce the amount of sunlight reaching the photosynthetic cells of the leaflets. Sooty mold growth can reduce light transmission to the leaf by the 25% - 50% (Tedders and Smith, 1976). The honey dew itself indicates the amount of carbohydrates and water the aphids extract from the pecan leaves as they feed. Heavy sooty mold growth can block up to 98% of the light, suppressing leaf photosynthesis up to 70% and decreasing tree carbohydrate reserves (Tedders and Wood, 1985; Wood et al., 1988).

The traditional control of pecan aphids has been the use of insecticides. However, the application of these insecticides has been reevaluated because of the loss of effective products, resistance (Dutcher and Htay, 1985), outbreak of secondary pests (Ball, 1981), resurgence of aphids and mites after the use of pesticides to control other pests (Dutcher, 1983), and concern about the environment. Repetitive insecticide application can also produce a sustained reduction in net photosynthesis and could be another factor reducing tree energy reserves (Wood et al., 1983). Results in apple and citrus studies have shown that some pesticides cause large reductions in photosynthesis (Wedding et al., 1952; Ayers and Barden, 1975; Sharma et al., 1977; Ferree and Hall, 1978). Current pest management principles include cultural control, reliance on natural enemies of insect pests and resistant cultivars. These alternate tactics can be useful in reducing pesticide use, retaining the efficacy of available pesticides, and enhancing the effects of natural enemies.

Biological control of aphids in pecan has been considered promising as an alternative approach in pecan management (Tedders, 1983; Liao et al., 1984, Edelson and Estes, 1987; Mizell and Schiffhauer, 1987; Bugg and Dutcher, 1989). Predators and parasites attack the pecan aphid complex causing a considerable reduction in their densities (Tedders 1978; Leser, 1981; Edelson, 1982; Watterson and Stone, 1982). These natural enemies that prey upon pecan aphids and other pests may be adversely affected by pesticide use (Dutcher, 1983; Dutcher and Payne, 1983). The biological control of aphids can be maximized by providing favorable habitat for predatory species to complete their life cycles. This means diversity in landscape, availability of hibernation sites and prey or alternative food sources such as pollen are important components to assure the practicality of biological control (Sotherton, 1985; Stechman, 1986). A monoculture promotes development of species with high reproductive rates because of lack of competition by other species and the absence of natural biological control. Vegetative diversity is a popular scheme to lower pest pressure either by increasing natural enemies or by lowering host plant availability in diversified systems (Russell, 1989).

From the economic point of view it may be wise to exploit naturally occurring antagonists like predators or parasitoids rather than rely upon more costly inputs such as pesticides. Thus, it would be both useful and necessary to preserve efficient antagonists, as well as other non-target arthropod species by careful management (Metcalfe, 1986; Poehling, 1989).

Predators are usually larger than parasites and are much more easily detected. Predators actively seek out, seize, overpower or immobilize their prey, and then consume it entirely or suck it dry of body fluids. Larval stages and often the adults feed on prey, consuming many prey (Gorsuch, 1984).

Several studies have addressed natural enemies that attack pecan aphids (Flores, 1981; Watterson and Stone, 1982; Liao et al., 1984; Liao et al., 1985; Edelson and Estes, 1987). Coccinellids are one of the most recognized predators. Both adult and larvae are voracious predators of pecan aphids and can eat as many as 62 aphids/day (Goff et al., 1989). Some of the Coccinellids known to attack pecan aphids are the seven spotted lady beetles (*Coccinella septempuntata* [L.]), convergent lady beetle, (*Hippodamia convergens*

[Guerin-Meneville]), and ash grey lady beetle (*Olla v-nigrum* [Mulsant]) (Gordon, 1985). The Chrysopids (Neuroptera : Chrysopidae) and spiders (Arachnida) also play a significant role in maintaining aphid densities at low levels (Liao et al., 1985). Chrysopid larva can consume 25 to 30 aphids/day. A number of hover or syrphid (Diptera: Syrphidae) fly species have predacious larvae. The larvae are aphid predators and are often found crawling on a pecan foliage when aphids are present.

Assassin bugs (Hemiptera: Reduviidae) and damsel bugs (Hemiptera : Nabidae) are predators that feed upon a large variety of plant feeding insects (Tedders, 1976). A few stinkbugs (Hemiptera : Pentatomidae) are also predators. The beneficial stinkbugs feed mainly on caterpillars and other large soft-bodied insects. The beneficial stinkbugs are separated from harmful stinkbugs by their mouth part structure (Tedders, 1976).

A number of natural enemies have been associated with the blackmargined aphid on pecan. About 13 species of spider and 27 species of predacious insects including larvae of *Chrysopa* spp. and several members of Coccinellids were reported as the most abundant predator groups (Shepard, 1973; Tedders, 1978; Flores, 1981; Edelson, 1982). Laboratory feeding studies of selected Chrysopid and Coccinellid predators showed average feeding rates of between 25 and 60 aphids/day. Results indicate that natural enemies, particularly predators, play an important role in maintaining *M. caryella* population at low levels in the field (Liao et al., 1985).

In intensively managed pecan orchards the dominant understory are mowed grasses which rarely harbor aphids and seldom sustain aphidophagous species (Bugg et al., 1991a). Under minimal or commercial pecan management, cool-season understory legume cover crops sustained nearly six times more aphids than unmown resident vegetation and approximately 87 times more than mowed grasses and weeds (Bugg et al., 1991a). Many aphidophaga require aphids in their diets to reproduce, and will not colonize crops having low aphid densities. Hence when aphids become scarce the aphidophaga may disperse (Sluss, 1967). Lack of aphids may induce reproductive dormancy in some lady beetles such as convergent lady beetles which may form aggregations and remain inactive throughout the summer (Haggen, 1974). Such disengagements of predator from prey can prompt outbreaks of aphids, because recolonization by predators may occur too late.

These problems have increased interest in enhancing biological control through habitat manipulation (Tedders, 1983). Pecan is considered an ideal crop to demonstrate successful pest control through habitat manipulation. Pecan grows well under diversified conditions, and is inhabited by numerous arthropods. The presence of legume ground covers in pecan orchards has increased the number of numerous beneficial species (Tedders, 1986). Among these are the southern two-spotted lady beetle (*O. v-nigrum*), an arboreal species that feeds on pecan aphids and mites. This species is probably one of the most important predators that normally inhabits pecan trees. However, it becomes active before pecan trees foliate and vacates the monoculture orchard in search of food. The presence of aphids and mites on legumes provides food for lady beetles in the orchard and thus may arrest their migration until aphids appear on the trees (Tedders, 1983; Bugg et al., 1991a).

Legume understory cover crops provide and promote successful overwintering by predators (Mizell and Schiffhauer, 1987) and also provide aphidophaga with alternate prey, nectar and pollen (Altieri and Letourneau, 1982). Nectar or pollen is fed upon by some adults of the following aphidophagous predators : lady beetles (Coleoptera: Coccinellidae) (Haggen, 1962; Bugg, 1987), green lacewings (Neuroptera: Chrysopidae) (Sunby, 1967; Canard et al., 1984; Bugg , 1987), brown lacewings (Neuroptera: Hemerobiidae) (Bugg, 1987), and hover flies (Diptera: Syrphidae) (Ozols, 1964; Schneider, 1969). In addition, Pereyra and Villanueva (1987) noted that three hymenopterous parasites of pecan pests fed on nectar-bearing weeds in Mexican pecan groves. A mixture of cool season plant species might provide a seasonal sequence of foods for lady beetles that might otherwise disperse from pecan orchards (Bugg et al., 1990).

Certain legumes harbor large populations of alternate prey aphids which attract aphid predators and parasitoids (Smith et al., 1994). The alternate prey aphids common on legumes in the southern U.S. are blue alfalfa aphid (*Acyrthosiphon kondoi* [Shinji]), pea aphid (*Acyrthosiphon pisum* [Harris]), cowpea aphid (*Aphis crassivora* [Koch]), and yellow clover aphid (*Therioaphis trifolii* [Monell]) (Bugg et al., 1990). Particularly the pea aphids heavily infest cool season legumes and may attract lady beetles, predacious soldier bugs (*Stiretrus* spp.), damsel bugs (*Nabid* spp.), hover flies (*Syrphid* spp.) and spiders (Smith et al.,1992). When aphid densities on the legumes crash or are eaten, the beneficial arthropods may seek alternative food sources in the pecan trees. Economically important outbreaks of pecan aphids may be controlled in the spring by movement of these beneficial arthropods from the legumes into the trees where they feed on aphids and other pecan pests. Earlier work showed that two generations of *H. convergens*, produced on pea aphids feeding on hairy vetch and clover, reached an estimate of 353,000 individuals per ha under normal field conditions by mid-May. When the legumes mature, the food source of pea aphid disappears causing migration of lady beetles (Tedders, 1986). Bugg et al., (1990) reported that during April and early May, tarnish plant bug adults and nymphs (Hemiptera : Miridae) were particularly abundant as alternative food sources for beneficials on hybrid vetches and intermediate on crimson clover.

The predatory fauna associated with aphids includes generalists and those that are aphid specific. The generalist predators can reproduce or at least subsist on nectar, pollen, thrips (Thrysanoptera), aphids (Homoptera: Aphididae), or spider mites (Acari: Tetranychidae), and thus be "in place" before the arrival of key pests (Ehler and Miller, 1978; Tamaki, 1981; Gonzalez et al., 1982). Edelson and Estes (1981) listed several predatory and one parasitic species of insects plus numerous species of spiders that feed on pecan aphids. The preservation of parasites and predators in the spring should reduce the problems encountered with other pecan pests later in the season. Chrysopids were the major predators encountered in the early season and spiders were the major predators encountered in the late season.

Bugg et al., (1991b) proposed a cover crop management system for pecan orchards that promoted biological control by increasing beneficial arthropods in the orchards. A number of cool-season and warm-season cover crops were evaluated in Georgia, Oklahoma, and Massachusetts to increase aphidophagous insects and other entomophaga to enhance biological control in vegetable and pecan agroecosystems

(Wood et al., 1983; Bugg and Dutcher, 1989; Bugg and Ellis, 1990; Bugg et al 1990, 1991a; Bugg and Dutcher, 1993; Smith et al., 1994). Cool season legumes such as hairy vetch (*Vicia vellosa* [Roth]) and crimson clover (*Trifolium incarnatum* [L.]), together with rye (*Secale cereale* [L.]), sustain aphids that do not attack pecans, along with the Coccinellids (eg: *H. convergenens*, *C. septempunctata*, *O. v-nigrum*) that can disperse to pecan trees and attack pecan aphids. Warm-season legumes such as Sesbania (*Sesbania exaltata* [Rafinesque-schmaltz] Colt) understories harbored substantial populations of cowpea aphids (*A. craccivora* [Koch]) and banded winged whitefly (*Trialeurodes abutilonea* [Haldeman]), and can sustain lady beetles when pecan aphids are scarce. Among several cool season perennial legumes evaluated, 'Kenland' red clover was one of the highest in attracting certain beneficial arthropods but few beneficial arthropods were attracted by 'Louisiana S-1' white clover (Smith et al., 1994). The highest legume aphid density occurred during early spring, and was positively correlated with lady beetle densities.

In a study conducted by Rice (1994), large aphid densities were found on a mixture of 'Dixie' crimson clover and hairy vetch which attracted beneficials. The most abundant beneficial arthropods sampled were spiders, lady beetles, green lacewings and nabids, respectively. The legume ground cover did not affect the densities of beneficial species in the pecan canopies. However, the most abundant beneficial arthropods in the pecan canopies were spiders, green lacewings and lady beetles, respectively .

Augmentation of *Trichogramma* to Control Lepidopterous Pecan Pests

One of the three primary nut feeding insect pests attacking pecan is pecan nut casebearer *(Acrobasis nuxvorella* Neunzig (Lepidoptera: Pyralidae) (McWhorter et al., 1976). Pecan nut casebearer is a native lepidopterous insect and is widely distributed wherever the pecan is indigenous (Bilsing, 1926). This monophagous, multivoltine insect caused damage almost every year from the early 1900s' until the advent of new chemicals and application equipment (Harris, 1990). A gradient of damage by the pecan nut casebearer exits from east to west with occasional losses in Georgia to heavy losses in Texas. This gradient appears to be connected with unmanaged native pecans (Boethel et al., 1979).

The pecan nut casebearer has been reported to destroy up to 90% of the pecan crop (Bilsing, 1926). In southeast Kansas pecan nut casebearer damaged from 6.0 to 20.5% of fruit clusters from 1979 to 1983 (Reid et al., 1984). In 1972, Louisiana pecan growers treated all the commercial pecan orchards with insecticide to control the first generation (Boethel et al., 1979). The abundance of this insect depends upon the extent of the crop the previous year, the number and kinds of parasites present, and climatic conditions during March, April, and May. Some pecan growing regions currently use a degree day model to predict the date that treatments for the pecan nut casebearer should be applied (Ring and Harris, 1983)

Pecan nut casebearer overwinters as a small larva in a cocoon called a hibernaculum attached to a dormant pecan bud. The larva becomes active coincident with budbreak of the pecan in the spring and grows and develops by first feeding on the buds and then tunnels into the rapidly growing shoot causing it to wilt, turn dark, and die. The tunnel entrance is typically identified by webbing and frass that the larva pushes out (Harris, et al., 1988). Larval growth and development is typically completed in a few weeks and larva are ready to pupate. Bilsing (1926) estimated that about 50 percent of the larvae pupate in shoots they have excavated, while the remainder move down the limb to rough bark where crevices provide concealment and protection from predators during this vulnerable period of transformation from larva to adult. The adult moth emerges from the pupa within a couple of weeks, mates, and females begin to lay eggs on the flower stigma, typically laying one egg per flower cluster. The most severe damage from this fruit feeder is caused by the larva of the first generation. One larva hatches from each egg about four days after deposition and feeds for one to two days on tender buds at the base of the fruit cluster. Then the larva moves to the fruit and tunnels into the fruit. Larval growth and development requires about three weeks. Each larva will consume two to three fruit during this period, often leaving the hollow shriveled brown remnants of damaged fruit dangling from the cluster. This cycle is repeated for three to five generations depending on fruit availability and length of the growing season (Bilsing, 1926; Gill, 1924; Harris et al., 1988). In Oklahoma, the first generation of pecan nut casebearer normally occurs about 1-10 June and the second generation emerges in July.

Most pecan growers normally apply insecticides, usually organophosphates, to prevent economic damage of pecan nut casebearer in most years. However, experience has shown that the routine applications of insecticides can lead to a myriad problems such as : (1) elimination of natural enemies (predators and parasites) of all pests including those of the target pest; this leads to (2) pest resurgence and outbreak of aphids that were previously kept in check by natural enemies; (3) resistance to insecticide develops due to the elimination of the susceptible individuals in the pest population leaving only the resistant ones to breed and form the next generation; and (4) the addition of chemicals to the environment increases the probability that pollution of air, soil and water will occur (Harris, 1983). Therefore, an alternative control method for pecan nut casebearer which would not destroy beneficial insect populations could eliminate the need for certain pesticide applications in pecan orchards. One possible strategy could be the augmentation of existing natural enemies. Augmentation is releasing beneficial insects purchased from commercial sources for the purpose of pest control. *Trichogramma* spp. are the most widely used commercially available entomophagous insects in the world (King et al., 1984). It has received the most attention because of its importance in biological control. *Trichogramma* wasps are used more than any other entomophagous species for biological control of insect pests in more than 30 countries (Stinner, 1977; King et al., 1985). According to the data collected by Hassan (1988) and others (Filippove, 1990; Li, 1994) over 32 million ha of agriculture and forestry in the world has been treated annually with *Trichogramma* for controlling insect pests. *Trichogramma* has been introduced to countries where the government and public opinion support biological control for ecological and economical reasons, and where the mechanization for mass-rearing *Trichogramma* has been developed for a long time, or where the labor cost is low.

The field augmentation of naturally occurring introduced *Trichogramma* species is most commonly achieved via inundative releases of *Trichogramma*, wherein the crop is flooded with insectory - reared parasitoids to raise the field parasitism rate sufficiently to prevent economic injury to the crop (Olkowski and Zhang, 1980). The *Trichogramma* wasp develops inside the pest egg and emerges as an adult in eight to ten days, ready to attack more pest eggs. The short life cycle allows for rapid increase in *Trichogramma* when pests appear, resulting in as many as 30 generations per season. The parasitized moth eggs can be easily recognized because the moth egg color darkens to brown or black when the parasite pupates inside. Periodic release of *Trichogramma* assures the presence of overlapping generations of mated females ready to attack the eggs of many moths that fly into the orchard.

Trichogramma spp. have been considered generalists in their choice of hosts (Thompson and Stinner, 1989). It has been suggested that plant-generalist natural enemies use more general chemical cues than plant specialists (Sheehan, 1986). Hence host generalist parasitoids may use a wider range of chemical cues in their search for hosts than host specialists. Kairomones play an important role in stimulating host searching behavior of these parasitoids (Jones et al., 1973). Kairomones not only stimulate host seeking behavior but also may help retain released parasitoids within a targeted area. Adult stages of *Trichogramma* spp. are generally highly susceptible to most broad-spectrum chemical insecticides, especially those used in the management of the larval stages of phytophagous lepidoptera. These parasites may be affected immediately by direct exposure to spray applications or drift of pesticides as well as to post treatment contact with residues of pesticides on foliar surfaces, some of which retain significant levels of toxicity for prolonged periods (Bull and Coleman, 1985). In contrast, immature stages of the parasites developing within host eggs apparently are well protected from even the most toxic compounds.

Control with *Trichogramma* is mostly attempted through mass releases against at least 28 different phytophagous pest species on some 20 different crops (King et al., 1985; Smith et al., 1987; Hassan, 1988; Smith, 1988; Voegele et al, 1988). Extensive utilization of this parasitoid was developed on corn, rice, sugar-cane, cotton, vegetables, fruit trees and forest trees. Development of *Trichogramma* for use in fields of vegetables (cabbage, tomato, beans, beet, etc.) vineyards, and fruit orchards (apple, plum, citrus, etc.) is taking place in more and more countries (Li, 1994). The advantage of *Trichogramma* is not only the direct control of the pests but it may also have a preventative effect. It provides benefits to the successive years of crop cultivation, even if the direct control effect is sometimes not as high as chemical treatment.

Although there have been large releases of *Trichogramma* in inoculative or augmentative programs for the control of lepidopterous pests, the success of such programs is variable (Stinner, 1977; Hassan, 1981). Some of the main factors affecting the success of these programs using *Trichogramma* include the choice of an appropriate wasp species or strain (Pak, 1988), climate and crop (Dijken et al., 1986), reasonable release rate per hectare, release methods, methods of conducting release (Li, 1994), superparasitization of ages during mass-rearing (Salt, 1937), ages of eggs used for mass-rearing (Reznik and Umarova, 1990), kairomones, syromones and other host related cues (Lewis et al., 1972,1975; Altieri et al., 1981; Nordlund et al., 1985; Thompson and

Stinner, 1990) and the conditioning of the wasps to cues related to the factitious or natural hosts (Nordlund et al., 1985; Dijken et al., 1986).

In North America, *T. pretiosum* Rilley (Heimenoptera :Trichogrammatidae) has been used principally as an agent for the control of *Heliothis zea* (Boddie) and *H.virescens* (F.) in cotton in the southeastern United States (King et al., 1985, 1986) and of *H. zea*, *Trichoplusia ni* (Hubber) and *Manduca sexta* (Johanson) in summer processing tomatoes in southern California (Oatman and Planter, 1971, 1978). Nevertheless, the success of this species in some of the projects has varied. For example, the control of *H. zea* by releases of *T. Pretiosum* in cotton was judged a failure, where as releases against *H. zea*, *T. ni* and *M. sexta* in experimental plots of processing tomatoes were judged successful (Bai et al., 1992). The percentage of parasitism by *T. pretiosum* can be enhanced by releasing a higher density. This species usually prefers singly laid eggs to eggs which are laid in overlapping layers and protected by scales (De Sa and Porra, 1994). The reproductive success of *T. pretiosum* in the field may depend on the availability of a carbohydrate source. In the absence of honey or another suitable food source during rearing, the species is short-lived (Stinner et al., 1974).

Dispersal of *Trichogramma* spp. in an apple orchard and the rate of parasitism of the eggs of the codling moth, *Cydia pomonella* (L.) was studied by Yu et al.(1984). It was reported that vertical and horizontal dispersal within the tree by *T. pretiosum* was even in all directions and was not affected by wind direction. The vertical and horizontal distribution of *T. minutum* was also even in all directions; however, vertical distribution of *T. minutum* within the trees adjacent to the release site was skewed toward the lower part of the tree canopy.

Trichogramma spp. vary greatly in their searching behavior, host preference and response to environmental conditions. Consequently, they also vary in their suitability for use in biological control. Failure of egg parasites to control agricultural pests could be due to the use of less suitable *Trichogramma* strains (Hassan, 1989). Climatic factors such as temperature, humidity and dew, wind and possibly duration of photophase significantly affect the movement and efficacy of the parasites. In addition, other authors have also indicated particular attributes that would increase the biological abilities of the parasite and its effectiveness in the field (Lenteren, 1986; Pak, 1988; Bigler, 1989; Pak et al., 1991; Pavlik, 1993). A survey of the natural abundance of egg parasites and their potential for use in biological control was reported by Hassan, (1992). For the parasite to be successful in the field it must locate food, host eggs and shelter. The abundance and distribution of the host is equally important. Adult parasites that do not rapidly find food and shelter have a much shorter lifespan and less chance of locating hosts (Hassan, 1994). Recent research on *Trichogramma* spp. has focused on host preference and host suitability (Pak, 1988). Attempts have also been made to quantify behavioral traits associated with host acceptance and oviposition as quality control and strain selection criteria.

Early studies in central Texas indicate that *Trichogramma* spp. is the most important parasite of the first generation of the pecan nut casebearer (Nickles, 1931). An average of 14.3% of the eggs collected were parasitized. The eggs of the leaf casebearer, shuckworm, walnut caterpillar, and possibly several species that feed on pecan are also attacked by *Trichogramma*. Previous records show that not less than 24 species of primary parasites attack the larva of the pecan nut casebearer. However, the majority of the species larval parasites that attack the nut casebearer do not usually destroy their host until after it has fed on pecan for a period of approximately seven to 30 days. The larval parasites of the nut casebearer decrease the rate of multiplication of their host and also reduce to a limited extent the number of fruit that are destroyed by the larvae which they attack. In contrast, *Trichogramma* destroys the eggs of the nut casebearer and thereby prevents the development of the larval stage, which is the one injurious to pecan. *T. pretiosum* was known to attack the eggs of hickory shuckworm and pecan bud moth, and probably other lepidopterous species. This species was tested against hickory shuckworm in the laboratory and in the orchard. The result indicated that 90% of the eggs in the laboratory were parasitized; however, control was not achieved in the field (Calcote and Tedders, 1990).

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CHAPTER II

PERENNIAL LEGUME GROUND COVERS INCREASE BENEFICIAL ARTHROPODS AND SUPPLY NITROGEN IN PECAN ORCHARDS

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Abstract: Perennial legume ground covers were evaluated in pecan (*Carya illinoinensis* [Wangenheim] C. Koch) to supply N and increase beneficial arthropods. Treatments were ground covers as pure stands or a mixture of 'Kenland' red clover (*Trifolium pratense* L.) and 'Louisiana S-1' white clover (*Trifolium repense* L.), plus a grass sod (primarily burmudagrass, *Cynodon dactylon* L.). Nitrogen was applied at 0-168 kg/ha in 56 kg intervals to the trees in the grass plots but no N was applied to the legume plots. Aphids and beneficial arthropods were monitored. The most abundant beneficial arthropods sampled in the legumes were spiders, lady beetles, predacious stinkbugs and nabids, respectively. Legume ground covers did not affect the densities of beneficial arthropods in pecan canopies. In pecan canopies, spiders, lady beetles and green lacewings were the most abundant. In legumes, predominant lady beetle species were *Colliomegilla maculata lengi* (Mulsant) and *Coccinella septempunctata* (L.) (Coleoptera: Coccinellidae). *Olla v-nigrum* (Mulsant) and *Harmonia axyridis* (Pallas) were lady beetles found only in pecan. The combination of red plus white clover appeared to yield sufficient N to meet the pecan requirements.

Introduction

Legumes were widely used in pecan orchards prior to the 1950s, but with the availability of inexpensive commercial N fertilizers they disappeared (Tedders, 1983). Interest in using legumes in orchards has recently increased due to higher N fertilizer costs, and legume potential to attract beneficial arthropods. Frequent pesticide applications have led resistance in certain insects and mites (Boethel, 1981; Dutcher and Htay, 1985) and outbreaks of secondary pests by elimination of natural enemies due to pesticide application(Mizell, 1991).

Current pecan management practices rely on heavy application of fertilizers, pesticides and mowing (Bugg et al., 1991a). Pecans produced from "native" trees represent more than one third of the total U.S. production. In Kansas, Oklahoma, and Missouri, native pecans account for over 90% of the pecan acreage (Thompson, 1984). Trees in native pecan orchards are thinned to optimize production (Harris, 1983). They are mostly located on alluvial soils near streams. The common sod in both native and cultivar orchards is bermudagrass (*Cynodon dactylon* [L.] Pers.), which reduces growth and yield of pecan trees by competing for moisture and nutrients (Gossard and Hammar, 1957; Ware and Johnson, 1958). Nitrogen fertilizer is applied annually at an application rate of more than 100 kg.ha^{-1} N (Harris, 1983). This high rate of application combined with unpredictable rainfall and a shallow water table have caused concern about the potential for ground and surface water contamination by NO_3. Nitrate is considered one of the major contaminants of ground water and is widespread in the U. S. (Benbrook, 1989).

The monocultural conditions of pecan orchards coupled with a more aggressive management style has promoted the development of many induced pecan pests, mainly aphids. Aphid species (Homoptera: Aphididae) that feed on pecan include blackmargined aphid, *Monellia caryella* (Fitch), yellow pecan aphid, *Menolliopsis pecanis* (Bissell), and black pecan aphid, *Melanocallis cayaefoliae* (Davis) (Tedders, 1978; Tedders et al., 1982). Yellow pecan aphids and blackmargined aphid cause damage to the foliage at the feeding site by clogging phloem, inducing chlorosis and necrosis of the surrounding mesophyll cells (Tedders and Thompson, 1981; Wood et al., 1985), suppressing photosynthesis in pecan leaves, and reducing nut size, weight and yield (Tedders and Wood, 1985; Wood et al., 1988). These aphids have been controlled with insecticides. Growers follow intense pest control programs with eight to 10 insecticide applications annually in the southeastern U.S. (Tedders, 1983; Bugg et al., 1991a), and five to seven insecticide applications in the south central U.S. These intesive pest control programs are expensive and reduce profits of pecan growers. Moreover, excessive use of certain insecticides has resulted in the development of pesticide resistance in aphids (Dutcher and Htay, 1985), and outbreaks of secondary pests (Mizell, 1991) creating the need for more pesticide use. Many pesticides have a negative impact on non-target organisms such as beneficial insects and mites that may be important in the natural control of both primary and secondary pests (Mizell, 1990).

Legumes and mixtures of legumes grown as ground covers in pecan orchards may offer certain advantages over current cultural practices. Nitrogen is fixed by symbiotic bacteria associated with legume roots then is used by the legume. When the legume roots and top decompose, stored N in the legume is released and a portion of the N may be used by the tree. This is especially important in native pecan orchards where inputs are restricted because yield and profit potential are lower than in cultivar orchards. Estimates of N fixed by legumes ranges from 57 to 207 kg. ha^{-1} N (Allison, 1957; Erdman, 1967). Crimson clover (*Trifolium incarnatum* L.) has been shown to add 57 to 78 kg. ha^{-1} in pecan orchards (Hunter, 1960). Rice (1994) found that a mixture of crimson clover and hairy vetch (*Vicia villisa* Roth) supplied the equivalent of over 100 kg.ha^{-1} N to the pecan trees. Smith et al. (1994) reported that red clover (*Trifolium pratens* [L.]) and white clover (*Trifolium rapens* [L.] are well adapted perennial clovers for pecan orchards in the southern U.S., fixing up to 180 kg. ha^{-1} N. Results from other studies indicated that white clover can fix up to 250 kg. ha^{-1} N annually (Halliday and Pate, 1976), and may rapidly improve soil structure (Taylor, 1985).

An additional benefit of legume ground covers is increased beneficial arthropods in the pecan orchard. Certain legumes harbor large aphid populations that attract beneficial arthropods that feed on aphids. When the aphid populations on the legumes decline or the legumes senesce, beneficial arthropods associated with the legumes may migrate to the pecan canopy, controlling pecan aphids and other pests (Smith et al., 1994). Legumes may be used to provide or promote successful overwintering by predators

(Mizell and Schiffhauer, 1987) and provide alternate prey, nectar and pollen (Altieri and Letourneau, 1982) that ensure retention of beneficial arthropods until pecan aphids become available. The alternate prey aphids common on legumes in the southern U.S. are blue alfalfa aphid, *Acyrthosiphon kondoi* (Shimji), pea aphid, *Acyrthosiphon pisum* (Harris), cowpea aphid, *Aphis craccivora* (Koch), and yellow clover aphid, *Therioaphis trifolii* (Monell) (Bugg et al., 1990). These aphids are not pests of pecan.

A cover crop management system that promotes biological control by increasing beneficial arthropods in pecan orchards has been proposed by Bugg et al. (1991a). Several cool-season and warm-season cover crops were evaluated in Georgia, Oklahoma, and Massachusetts to increase aphidophagous insects and other entomophaga to enhance biological control in vegetable and pecan agroecosystems (Bugg and Dutcher, 1989; Bugg and Ellis, 1990; Bugg et al., 1990, 1991a; Bugg and Dutcher, 1993; Smith et al.,1994). The purpose of this study was to evaluate two perennial legume ground covers, 'Kenland' red clover (*Trifolium pratense* L.) and 'Louisiana S-1' white clover (*Trifolium rapens* L.) as stands and in combination in pecan orchards for their N supplying capacity and potential to enhance beneficial arthropod densities that may control pecan aphids.

Materials and Methods

<u>The site</u>. This study was conducted in two commercial orchards in Oklahoma; one near Sapulpa and the second near Beggs. The soil at Sapulpa is a port loam (fine-silty, mixed, thermic; Cumulic Haplustoll; Mollisols) that is well drained. At Beggs, the soil is a Mason

silt loam (fine-silty, mixed, thermic; Cumulic Haplustoll; Mollisols) and relatively poorly drained with a perched water table. These soils occur on river bottom land, and are typical pecan soils in this region. Tree density at the Sapulpa orchard was 36 trees/ha with a cross-sectional trunk area of 2.7 m^2/ha, and at Beggs there were 51 trees/ha with a cross-sectional trunk area of 2.7 m^2 /ha. Native pecan orchards were on both sites.

Soil preparation and planting. Existing vegetation was killed by applying glyphosate in late August 1991. The soil was lightly disced in September and 'Kenland' red clover and 'Louisiana S-1' white clover, inoculated with the appropriate *Rhizobium,* were planted in 1991 at Beggs and in 1992 at Sapulpa. At Beggs, legumes were red clover, white clover, and a mixture of red and white clover. At Sapulpa, legumes evaluated were a mixture of red clover and white clover. Seeding rates at each site were 8 kg/ha for red clover, and 4 kg/ha for white clover. The seeds were distributed with a broadcast planter, then the soil was packed with a roller. A grass ground cover consisting primarily of bermudagrass was left intact as a control plot at each site. Trees at Sapulpa site were irrigated by sprinklers, but those at Beggs were not irrigated. Each ground cover treatment was about 5 ha.

In 1992 at Sapulpa, *Bacillus thuringiensis* var. kurstaki was applied during June for control of the first generation of pecan nut casebearer (*Acrobasis nuxvorella* Neunzig, Lepidoptera: Pyralidae). In 1993, chloropyrifos was applied during April and May against *Phylloxera notabilis* Pergande, *P. russelae Stotzel*, and *P. devastatrix* Pergande (Homoptera: Phylloxeridae), and June 17 against pecan nut casebearer. Carbaryl was applied to control pecan weevil *(Curculio caryae* [Horn], Coleoptera: Curculionidae) on September 15 and October 1, 1992 and August 28 and September 15, 1993 at Sapulpa.

The red and white clover plots were mowed in late June, and again during mid-October. The residue remained on the soil surface.

Arthropod Sampling. Arthropod populations were sampled on the legume ground cover during 1992 through 1994 at seven to 14 day intervals, using a 38-cm diameter sweep net. Five replicate samples consisting of 10 sweeps per sample were collected between 10 a.m. and 2 p.m. and placed in plastic bags with pesticide impregnated strips (S.C. Johnson, Inc., Racine, Wisconsin), transported to the laboratory, and frozen until the arthropods of interest could be identified and counted. The arthropods monitored on the legumes were aphids (Homoptera: Aphididae), lady beetles (*Hippodamia convergens* Guerin-Meneville, *Coccinella septempunctata* L., *Coleomegilla maculata lengi* Timberlake, *Cycloneda munda* [Say], *Olla v-nigrum* Mulsant, *Anatis labiculata* [Say] and *Harmonia axyridis* Pallas, Coleoptera: Coccinellidae), green lacewings (*Chrysoperla* and *Chrysopa* spp.); brown lacewings (Neurotera: Hemerobiidae); nabids (*Nabis* spp., Hemiptera: Nabidae), hover flies (Diptera: Syrphidae), predaceous stinkbugs (Hemiptera: Pentatomidae), phytophagous stinkbug (Hemiptera: Pentatomidae), and spiders (Araneida). Aphids were divided into alatae (winged) and apterae (wingless) plus nymphs. Lady beetle adults were separated then counted by species, and larvae pooled and counted. Larvae and adults of both green lacewings and brown lacewing, as well as nabid and hover fly adults, were counted. Counts of predaceous stinkbugs and phytophagous stinkbugs did not distinguish between adults and nymphs.

Arthropods in the tree canopy were sampled at seven to 10 day intervals from May through September using a vacuum sampling device (D-vac CO., Ventura, Ca.). Care was taken to position the collection device below the limb before the limb was disturbed to avoid arthropods, particularly lady beetles, escaping collection. Five 0.1 m^2 areas per tree on five trees each in the legume and the grass plots were sampled on each date between 10 a.m. and 2 p.m. The collected samples were transferred into plastic bags containing pesticide strips, transported to the laboratory, frozen and later identified and counted. Arthropods monitored in the tree canopy by vacuum sampling were the same as in legume arthropod samples. Aphids (*Monelliopsis pecanis, Monellia caryella* and *Melanocallis caryaefoliae*) and unhatched lacewing eggs were counted on 10 compound leaves per tree and five trees per treatment at seven to 10 day intervals. Tree canopy arthropod samples, aphid and green lacewing egg counts were 3 to 4 m.

Nitrogen Analysis. At Beggs, five 1 m^2 area legume biomass samples (tops only) per treatment were harvested at the canopy periphery during June, 1992 and in 1993 during June and September. At Sapulpa, legume samples were harvested during June only. The samples were sorted into red clover, white clover and non-legumes, oven dried at 70°C, weighed, and analyzed for N using the macro-Kjeldahl procedure (Horowitz, 1980). Total N in the legume top was calculated from the dry weight of the legumes in each sample and the concentration in the legume tops.

Ammonium nitrate was applied at 0, 56, 112, and 168 kg/ha N to bermudagrass plots during March of each year. No N was applied to the legume plots. Pecan leaf samples (50 leaflet pairs from the middle leaf on current seasons' growth of each tree) were collected in July from 10 trees in legume plots and 50 trees in grass plots with known application rates (10 trees/N rate with each tree representing one replication). They were dried, ground to pass 20- mesh screen and analyzed for N by the macro-Kjeldahl method. Regression analysis was used to determine the relationship between applied N and leaf N. Leaf N supplied by the legumes was calculated based on the leaf N concentration from trees in the legume plots.

Soil samples were collected from different N rates applied to grass plots and from legume plots during October. Three subsamples were collected per replication from different locations at the canopy periphery at 0 to 15 cm and 15 to 30 cm depths. Each replication was replicated ten times. These samples were bagged then transported to the laboratory and frozen for later analysis. Before analysis samples were thawed for about eight hours, ground, sieved and 10 gm of soil was weighed for extraction and analysis. An additional 10 gm sample was oven dried to determine moisture content. Results were adjusted based on the moisture content and expressed on a dry weight basis. Nitrate-N was extracted using 2M KCL and was analyzed by the cadmium reduction method (Page et al., 1982) and non-NO_3- N utilizing macro-Kjeldahl method (Page et al., 1982).

Phytophagous stinkbug damage was determined by harvesting one 40 - nut sample per tree from 10 trees each from legume and grass plots. These pecans were then analyzed for stinkbug damage by examining kernels for black spots caused by feeding.

Crop load was compared among treatments by counting the number of fruiting shoots at mid-canopy height in two 6 m^2 areas per tree in September. In both legume and N treatments, 10 trees each were used, with the same trees used for crop load comparisons each year. Covariant analysis, with the previous years' yield as the covariant, was used to determine the effect of treatment on fruiting shoots.

Results

Aphids. During 1992 aphid densities at Beggs peaked in mid-April in the legumes, then declined (Table 1). Alatae were a maximum of 7% of the total aphids present in 1992. Aphid densities appeared to be associated with the rainfall patterns. Periods of frequent rainfall appeared to cause a rapid decline in aphid density.

In 1993, aphid densities at Beggs peaked in late April and early June, then declined in the legumes (Table 2). At Sapulpa the highest aphid density was observed on the initial sampling date (April 8), then declined. The next peak was on May 20, then aphid densities sharply declined. The alatae density was 7% of the total aphid density at both sites. Aphids species identified on the legume ground cover during 1993 were blue alfalfa aphid (0.5%), pea aphid (99%) and cowpea aphid (0.5%).

Aphid densities at Beggs in 1994 peaked May 26, with the highest aphid density observed in the combination of red and white clover (Table 3). At Sapulpa, aphid densities peaked on June 2 then declined rapidly.

Lady beetles. The density of lady beetle larvae in 1992 at Beggs peaked April 21, then declined and remained low through October (Table 4). Adult lady beetle densities peaked in mid-May following closely in the peak in lady beetle larvae. There were few differences in densities of lady beetle larvae among the legumes. The adult lady beetles in order of abundance in legumes were *H. convergens* (27%), *C. maculata lengi* (59%), *C. septempunctata* (8%), and *C. munda* (6%). Lady beetle densities in the tree canopies were small, and no significant differences were found among the lady beetle species in the tree canopy (data not shown). In 1993 at Beggs, lady beetle larvae collected on the legumes from June 8 through July 22 were stable with a slight population peak on June 8 and June 24 (Table 5). Lady beetle adult densities peaked June 24. The lady beetle species distribution was *H. convergens* (3%), *C. maculata lengi* (76%), *C. semptempunctata* (14%) and *C. munda* (7%). No *O. v-nigrum*, *A. labuculata* or *H. axyridis* were found (Table 5). Lady beetle larvae found in the tree canopies at Beggs were not significantly different between grass and legume plots (Table 6). Lady beetle density in pecan was low in 1993 coinciding with a low density of pecan aphids (Table 2). In pecan canopies *C. maculata lengi* (40%) was the most abundant lady beetle species followed by *O. v-nigrum* (20%), *C. munda* (15%), *H. convergens* (10%), *C. septempunctata* (10%) and *A. labuculata* (5%).

At Sapulpa, few lady beetle larvae were observed in 1993 on the legumes. Lady beetle adult densities were erratic with a slight peak May 13 (Table 5). Species distribution was *H. convergens* (10%), *C. septempunctata* (4%), *C. maculata lengi* (80%) and *C. munda* (5%). No *A. labuculata*, *O. v-nigrum* or *H. axyridis* were found in the legumes. Adult *H. convergens*, *C. maculata lengi*, *O. v-nigrum* and *C. munda* in pecan canopies were not significantly different between grass and legume plots (data not shown). No *C. maculata lengi*, *C. septempunctata*, *O. v-nigrum*, *H. axyridis* and *A. labuculata* were found in the trees. The most abundant lady beetle species were *C. munda* (67%) followed by the *H. convergens* (33%). The highest densities of lady beetle adult occurred in early July.

Lady beetle larval densities in 1994 at Beggs peaked in June 21 on the legumes followed by a rapid decline. This rapid decline in larval density coincided with the decline in aphid availability. Adult lady beetle densities in the legumes were greatest June 21, with the highest density in the combination of red and white clover (Table 7). Species distribution was similar to 1993, with *C. maculata lengi* (55%) followed by *C. septempunctata* (19%), *C. munda* (15%) and *H. convergens* (10%). Adult lady beetle density in pecan canopy peaked June 21 coinciding with the peak on the legumes (Table 8). Lady beetle adults in pecan were not significantly different between treatments. The most abundant lady beetle species were *H. axyridis* (39%) followed by *C. munda* (22%), *O. v-nigrum* (17%), *C. maculata lengi* (5%). No *H. convergens* or *A. labiculata* were collected from pecan.

In 1994 at Sapulpa, lady beetle larval density on the legumes was erratic, with the highest density occurring June 21. Peak aphid densities occurred prior to peak lady beetle larval densities. Adult lady beetle density was greatest on June 21(Table 7). Three percent of the adult lady beetles in the legumes were *H. convergens*, 3% *C. munda*, 11% *C. septempunctata*, 76% *H. axyridis*, 8% *C. maculata lengi* and 0.2% *A. labiculata*. No *O. v-nigrum* was found on the legumes.

Peak lady beetle adult density in pecan canopy occurred in April 26 (Table 8). The most abundant lady beetle species were *H. axyridis* (37%), followed by *H. convergens* (16%) and *O. v-nigrum* (16%), and finally *C. munda* (10%). No *C. septempunctata*, *A. labiculata* and *C. maculata lengi* were found.

Species distribution of lady beetles was different between legumes and pecan canopies at both sites. At Beggs in 1993, *C. maculata lengi* and *C. septempunctata* were frequently collected in both legumes and trees. In tree canopies *C. munda* occurred in much larger density than in legumes, and *O. v-nigrum* was the second most abundant species in canopies, but was not collected in legumes. At Sapulpa in 1993, 67% of the lady beetles sampled in the trees were *C. munda* but only 5% were *C. munda* from the legumes. *H. convergens* was abundant in the pecan canopies (33%), but scarce in legumes (10%).

In the legumes, *C. maculata lengi* was the most abundant species in both 1993 and 1994. In pecan, *C. maculata lengi* was the most abundant in 1993 and lowest in 1994. *Olla v-nigrum* and *H. axyridis* were major lady beetle species collected only in pecan. Habitat preference varies among lady beetles. Some are arboreal, and others are terrestrial in nature. For instance, it was reported that *C. septempunctata* preferred plants with lower strata (Ipert, 1965). *Olla v-nigrum* and *C. munda* have more of an arboreal nature, preferring the habitat of the tree canopy to that of the legumes. *Hippodamia convergens* had a positive photo response, and neutral response to humidity, which favors both the legumes and pecan canopies. Bugg and Dutcher (1993) reported that large densities of lady beetles in the understory did not lead to significantly different lady beetle or aphid densities in the associated pecan trees. Liao et al. (1984) reported that lady beetle density was not closely associated with aphid density in pecan. These results support our findings.

<u>Green lacewing</u>. In 1992 at Beggs, green lacewing adult densities in the legumes peaked in mid-June and mid-September (Table 9). Green lacewing larvae in the legumes peaked slightly in late April. In pecan trees, green lacewing density peaked twice, first in mid-August then in early October. Green lacewing larvae populations in the trees were

highest during mid-August (Table 10). Neither green lacewing adult nor larval densities in the trees were affected by the ground cover.

In 1993, green lacewing adult and larval densities at Beggs peaked simultaneously in the legumes July 22 (Table 11). Green lacewing adults in the canopies peaked July 8, then increased again August 19 and 26 (Table 6). Peak green lacewing larval density occurred July 22. Green lacewing adult and larval densities in the canopies were not significantly different between grass and legumes (Table 6). At Sapulpa in 1993, green lacewing adult densities in the legumes were highest during early May. Green lacewing adults in the canopies were erratic, but most abundant during early June. Green lacewing larvae were present in the legumes on only one sampling date (May) (Table 11).

In 1994 at Beggs, there were few green lacewing adults, and consistent trends or differences were not observed among the treatments (Table 12). Peak green lacewing adults in the canopies occurred during early June. Green lacewing larvae were not found in the legumes, and in the pecan canopies larvae were present on only one sampling date. At Sapulpa in 1994, green lacewing adult densities in the legumes were erratic (Table 12). Green lacewing adult densities in the trees peaked in late May (Table 8). Green lacewing larvae in the legumes were found on only one sampling date (Table 12) and none was found in the canopies (Table 8).

Green lacewing adults in pecan, were usually not affected by treatment. Edelson and Estes (1987) reported that Neuroptera were the most abundant aphidophagous insect found in pecans. Liao et al. (1984) reported aphid density was correlated with green lacewing egg density.

Spiders. In 1992, the density of spiders in the legumes at Beggs peaked from July 8 to August 28 (Table 9). Spider densities were at their highest in the trees of both grass and legume plots in mid-August (Table 10), but there were no significant differences between the clover and grass plots. Similarly, spider density in the legumes in 1993 at Beggs was relatively low during spring and early summer then peaked in late July (Table 11). Peak spider density in the tree canopy occurred in late July (Table 6). Canopy samples showed no significant differences between legume and grass plots.

At Sapulpa in 1993, spider densities in the legumes were abundant in mid-August (Table 11). Spider densities in the canopies peaked in early August. Spiders were present throughout the sample period in both grass and legume plots (Table 6). In 1994 at Beggs, the density of spiders in the legumes was highest in early July (Table 12). Spider densities in the canopies were not significantly different between grass and legume plots (Table 8), but they were most abundant in late May. Spider densities in the legumes at Sapulpa in 1994 peaked in early July, but were present throughout the sample period in all the treatments. In the canopies, spider densities at Sapulpa were most abundant in late May.

Spider densities were large in both legumes and tree canopies at both sites. However, there were few differences in spider densities in the canopies associated with the ground cover treatment. Spiders in canopies are primarily arboreal species, whereas those in legumes are terrestrial species, and unlikely to migrate to tree canopies. Spiders are general predators, and their feeding activity was associated with arthropods other than aphids.

Other species. Damsel bugs at Beggs in 1992 were erratic in the legumes, peaking in mid-

June, and there were no significant differences among the treatments (Table 9). In 1993, damsel bug density in the legumes were also erratic but relatively abundant in late June. No significant differences were observed among the clovers (Table 11). Damsil bugs in 1994 peaked in mid-June and there was no significant difference among the treatments (Table 12). Negligible damsil bugs were found in the trees from 1992 through 1994.

At Sapulpa in 1993, damsel bug density was low, peaking in mid-June (Table 11). In 1994, damsel bugs were most abundant in early June in the legumes. No damsel bugs were found in the pecan canopies in either 1993 or 1994.

Predaceous stinkbugs sampled in the legumes at Beggs in 1992 were most abundant in mid-August, but there was no significant difference among the clovers. The phytophagous stinkbugs were found in most of the sample period with the major peak August 14. Negligible densities of predaceous and phytophagous stinkbugs were found in the pecan canopies (data not shown). Predaceous stinkbug densities in both 1993 and 1994 at Beggs were low and erratic with a slight peak in mid-June (Table 11, 12). The phytophagous stinkbug densities in legumes were most abundant during late July in 1993 and early-July in 1994. There were no significant differences in either year among the legumes (Table 11, 12). Predaceous and phytophagous stinkbug densities in pecan canopies were negligible in both 1993 and 1994 (data not shown). Nut damage by phytophagous stinkbugs was significantly different among the treatments in both 1992 and 1994, but no significant difference was observed in 1993. In 1992, nut damage in the legume plots was higher than in the grass plot (Table 13). Stinkbug damage over the three year study ranged from 0 to 5 %.

At Sapulpa in 1993, both predaceous and phytophagous stinkbug densities peaked in late April in the legumes and there were no significant differences among legumes (Table 11). In 1994, predaceous and phytophagous stinkbug densities were low and erratic, but the predaceous stinkbugs were more abundant in early June (Table 12). Predaceous stinkbugs were not abundant in pecan canopies and did not show consistent trends or differences among the treatments in either year (data not shown). Nut damage in 1993 was negligible in both legume and grass plots (Table 13). In 1994, nut damage was also low but significantly higher in the grass plot than in the legume plot. During both years nut damage ranged from 0.2% to 4%.

Hover fly densities in 1992, in the pecan canopies were not significantly different between treatments (Table 10). No hover flies were found in the legumes. Brown lacewing adults and larvae in the legumes and pecan canopies during 1992-1994 at both sites had negligible densities (data not shown).

No assassin bugs were found in the legume samples at Beggs in 1993. In pecan canopies, assassin bugs were not significant between control and legume plots and were only found on one date on the combination of red and white clover. At Sapulpa, assassin bugs in pecan canopies were erratic, with the peak density in early August (Table 6). In 1994, assassin bugs at Beggs in the legumes were negligible while those in pecan canopies were sampled from June 21 to September 6, with a peak in early July. There was no significant difference between grass and legume plots (Table 8). Assassin bugs at Sapulpa were sampled from mid-July to the end of September in the canopies, but were not significantly different between treatments (Table 8).

Pecan aphid and lacewing egg densities. In 1992, pecan aphid densities at Beggs were higher on September 15 on the trees of the red clover plus white clover plots (Table 14). Aphid densities were usually low in all the treatments, and no significant difference was observed. Unhatched lacewing eggs were also very low and not affected by the treatments (Table 15). Pecan aphids in 1993 at Beggs peaked on July 1on the red clover then remained low in all the treatments for the remaining sampling dates (Table 14). Aphids were low throughout the growing season (maximum density 2.0 aphids/leaf). No significant difference was observed in aphid densities among the treatments in all the sampling dates. Unhatched lacewing eggs in the canopies were negligible for all the treatments throughout the sampling dates, except on August 10 and 18 on white clover (Table 15). Aphid densities in 1994 were most abundant from late May to mid-June then decreased to zero by end of June. There were more aphids on May 26 and June 9 than on the other sampling dates (Table 14). In 1994, aphid densities peaked from May 24 to June 2 and the second peak was on June 20.

At Sapulpa, pecan aphid densities in the trees in 1993 peaked on June 30 on the grass plot (Table 14). However, aphid densities were very low and no significant difference was observed between the grass and the legumes. Unhatched lacewing eggs showed no significant difference between grass and clover plots. In 1994 pecan aphid densities were low, but significantly higher densities were found in the grass plots on some of the sampling dates (Table 14).

Legume biomass and nitrogen. Legume biomass from a single harvest at Beggs in 1992 was higher for red clover than white clover (Table 16). The red and white clover mixture contained 42% more white clover than red clover. Legumes accounted for 27-58% of the biomass in the plots. In 1993 at Beggs, legumes accounted for 42-90% of the biomass. The red clover and white clover mixture was composed of 35% red clover, 45% white clover, and 20% non-legumes. Thus white clover was the dominant legume when planted as a mixture. At Sapulpa in 1993, legumes accounted for 58% of the biomass and non-legume 42%. The dominant species was red clover.

Legume N concentration in the tops during 1992 at Beggs for white clover ranges from 2.77 to 2.95% and for red clover from 2.34 to 2.71%. In 1993 at Beggs, the legume N concentration for white clover ranged from 3.86% to 3.87% and red clover from 2.25 to 3.31%. Hence, during both years legume N concentration was higher for white clover ranging from 2.77 to 3.87%. At Sapulpa in 1993, the legume N concentration for red clover was 2.94% (Table 16).

Total N in 1992 at Beggs in red clover ranged from 8 kg/ha to 56 kg/ha and in white clover from 19 kg/ha to 45 kg/ha. In 1993 at Beggs, the total N in red clover ranged from 11 kg/ha to 23 kg/ha and in white clover from 17 kg/ha to 35 kg/ha. At Sapulpa in 1993, the total N in red clover was 31 kg/ha. Thus total N in the legume tops at Beggs was not consistent between the clovers. At Sapulpa legumes were only harvested one time in 1993.

Soil nitrogen. At Beggs, Kjeldahl-N concentration was variable among the treatments (Table 17). There were significant cubic and quadratic trends in Kjeldahl-N related to N application rate at 0-15 cm and 15-30 cm depths in 1992 and 1993. In some instances the Kjeldahl-N concentration were higher using a legume ground cover and lower in other instances when compared to the various fertilizer rates with a grass sod. In 1994, there was more Kjeldahl-N in the red plus white clover plot than the fertilized grass plots at 0-15 cm and 15-30 cm depth (Table 17). At Sapulpa in 1993 and 1994 at 0-15 cm and in 1993 at 15-30 cm, there were no significant differences in Kjeldahl-N between the legumes and fertilized grass plots. In 1994 at 15-30 cm depth, there was significantly more Kjeldahl-N in the soil with red clover plus white clover than in the N applied soils. Kjeldahl-N concentrations were higher at Sapulpa than at Beggs. The poorly drained soil with a perched water table at Beggs probably accounts for the variable results obtained at this site and the lower Kjeldahl-N concentrations.

At Beggs in 1992, there were no significant differences in soil NO_3 in the upper 15 cm between fertilized grass plots and the legumes, and at the 15-30 cm level there were few differences between the legumes and grass plots. In 1993, soil NO_3 concentration in the red clover plus white clover was greater than any of the fertilized grass plot treatments at both 0-15 cm and 15-30 cm depth. The white clover plot had more soil NO_3 at the 15-30 cm level than the fertilized grass plots, but treatment did not affect the upper 15 cm, except at the 112 kg/ha N rate. Soil NO_3 was similar using red clover or fertilized grass plots. In 1994, soil NO_3 concentration was higher in the 15-30 cm level in the 168 kg/ha N plot than the legume plot. However, in both 1993 and 1994, the NO_3 concentrations in the red clover plus white clover plots were greater than in red or white clover plots at both depths (Table 17).

At Sapulpa in 1993, there were no significant differences between soil NO_3 concentration of the legumes and fertilizer grass plots at either soil depth. However, in

1994 soil NO_3 concentrations at both depths were higher using a red clover plus white clover than in the fertilized grass plots (Table 17).

Yield. The percentage of fruiting shoots/tree in 1992 and 1993 at Beggs and in 1993 at Sapulpa were not significantly different among the treatments (Table 18). In 1994, there was a greater percentage of the fruiting shoots in the fertilized grass plots than the legumes at Sapulpa, but not at Beggs. Fruiting has been found to influence the subsequent crop (Spark, 1983).

Nitrogen supplied by legumes. Pecan N concentrations at Beggs were below the minimum recommended level (2.25%; Smith et al., 1992) during all three years of the study using either white clover or red clover. The red plus white clover ground covers yielded pecan leaf N concentrations barely above the minimum sufficiency level. Estimates of nitrogen supplied to the trees by the legumes, based on regression equations derived from known N application rates to trees with grass sod, ranged from none to 132 kg/ha N (Table 19). Correlations of applied N with leaf N concentration were significant, but weak. At this site both red and white clover grew well; however, poor soil aeration resulted in little response to N derived from the legumes or applied N. Legumes with low aeration may have few nodules formed (Sprent and Sprent, 1990), and therefore there will be little N fixed by the legume. Earlier research indicated that flooding or water saturated soils severely reduces leaf N concentration in pecan (Wazir et al., 1988).

At Sapulpa, leaf N concentrations were maintained above the minimum sufficiency level by red clover plus white clover during 1993 and 1994 (Table 19). Estimates of N supplied by the legumes ranged from 93 to 105 kg/ha N for the red clover plus white clover.

Conclusions

Aphid densities on the clovers were generally low at both sites during the study periods. However, relatively higher densities were observed on the mixture of red plus white clover. The most abundant beneficial arthropods sampled on the legume ground covers were spiders, lady beetles, predacious stink bugs and nabids, respectively. Legume ground covers did not affect the densities of beneficial arthropods in pecan canopies. This may be attributed to a low pecan aphid density in the trees or differences in habitat preference of certain beneficial species.

There were relatively low and erratic densities of aphids in pecan canopies during the three years. In 1994 at Sapulpa, significantly more aphids were found on some of the sampling dates in the canopies with the grass plot than in the clovers. At other times aphid densities were not affected by treatments. The low densities of aphids in the canopies would necessitate the beneficial species to migrate to areas in which a food source existed.

The most abundant beneficial arthropods in pecan canopies were spiders, lady beetles and green lacewings, respectively. Spiders are general predators and their density may not be associated with pecan aphid densities. Lady beetle species distribution was different between pecan canopies and legumes. In legumes, *C. maculata legi* was the most abundant species each year, but in pecans it was the most abundant species in 1993 and lowest in 1994. *Olla v-nigrum* and *H. axyridis* were major lady beetles species found in pecan, but they were not collected from the legume ground covers. Green lacewings were collected from both the pecan canopies and legume ground covers. However, their densities were low and inconsistent.

Total biomass from a single harvest of the legumes ranged from 817 kg/ha (dry weight) to 2475 kg/ha. The legume N concentration for red clover ranged from 2.34 to 3.31% and white clover 2.77 to 3.87%. A mixture of red plus white clover at both Sapulpa and Beggs appeared to yield sufficient N ranging from 2.26 to 2.51% to meet the N requirement of pecans. Either red or white clover resulted in low pecan leaf N. At Beggs, leaf N concentrations were substantially below those at Sapulpa. This was because of poor soil aeration at the site affecting the N fixing capacity of the legumes which ultimately limits the accumulation of N in the pecan leaves.

Soil nitrate concentrations were frequently higher during October using a legume ground cover than most of the applied N rates on grass sod. During winter pecan roots are not active to readily absorb the accumulated NO_3 in the soil. Therefore, the chance of NO_3 leaching below the root zone might be higher by using legumes than well timed N application. However, the leaching problem may depend on the intensity and quantity of rainfall, soil type, as well as the amount of N accumulated in the soil.

Overall these data suggest that use of certain legume ground covers to increase beneficial arthropods for biological control of pecan pests is not likely to be successful. Their impact on the beneficials in the trees was small. Others evaluating ground covers that increased beneficial arthropods to control pecan aphids were not successful (Bugg et al., 1990; Rice, 1994). Hence, the primary benefit of these legume ground covers appeared to be the N contribution to pecans.

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Table 1. Legume aphid densities in 1992 at Beggs.

		Aphids/sweep		
Date	Clover	Apterae	Alatae	Total aphids
April 14	Red	9.68(7.51)[z]	0.46(.50)	10.14(7.52)
	White	12.40(6.45)	0.54(.39)	12.94(6.79)
	Red+white	17.84(5.79)	0.24(.20)	18.02(5.86)
April 21	Red	3.28(2.85)	0.44(.21)	3.72(2.92)
	White	1.22(1.06)	0.08(.13)	1.30(1.05)
	Red+white	3.32(4.82)	0.64(.75)	3.96(5.52)
May 5	Red	2.55(.52)	0.55(.10)	3.10(.44)
	White	1.50(.39)	0.60(.33)	2.10(.52)
	Red+white	0.50(.48)	0.16(.05)	0.66(.52)
May 22	Red	2.18(1.24)	0.22(.22)	2.40(1.32)
	White	1.34(.92)	0.02(.04)	1.36(.94)
	Red+white	1.02(.92)	0.06(.05)	1.08(.97)
June 17	Red	0.16(.26)	0(0)	0.16(.26)
	White	0.18(.18)	0(0)	0.08(.18)
	Red+white	0.04(.05)	0(0)	0.04(.05)
July 8	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 24	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Aug 14	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0.04(.09)	0(0)	0.04(.09)
Aug 28	Red	0.48(1.03)	0(0)	0.46(1.03)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Sept 15	Red	5.78(4.14)	0.88(.64)	6.66(4.44)
	White	1.30(1.63)	0.04(.09)	1.34(1.72)
	Red+white	2.56(1.27)	0.40(.25)	2.96(1.46)
Oct 1	Red	3.38(3.19)	0.16(.16)	3.54(3.35)
	White	0(0)	0(0)	0(0)
	Red+white	0.10(.22)	0(0)	0.10(.22)
Oct 12	Red	2.74(3.11)	0.40(.48)	3.14(3.56)
	White	0(0)	0(0)	0(0)
	Red+white	0.02(.04)	0(0)	0.02(.04)

[z] Mean (standard deviation).

Table 2. Legume aphid densities in 1993.

		Aphids/sweep		
Date	Clover	Apterae	Alatae	Total aphids
		Beggs,OK		
April 15	Red	0.94(.77)[Z]	0(0)	0.94(.77)
	White	1.12(.85)	0(0)	1.12(.85)
	Red+white	0.96(.74)	0.02(.04)	0.98(.78)
April 22	Red	3.50(1.30)	0(0)	3.50(1.30)
	White	1.28(1.17)	0.02(.04)	1.30(1.16)
	Red+white	2.12(1.72)	0.02(.04)	2.14(1.71)
June 1	Red	1.82(1.50)	0.04(.09)	1.86(1.48)
	White	3.32(1.66)	0.58(.21)	3.90(1.67)
	Red+white	4.06(1.92)	0(0)	4.06(1.92)
June 8	Red	0.52(.35)	0.06(.05)	0.58(.31)
	White	0.98(.81)	0.02(.04)	1.00(.84)
	Red+white	2.58(2.10)	0.06(.09)	2.64(2.13)
June 16	Red	0.60(.28)	0(0)	0.68(.28)
	White	0.72(.26)	0.46(.37)	1.18(.26)
	Red+white	5.46(3.69)	0.82(.36)	6.28(3.94)
June 24	Red	0.02(.04)	0(0)	0.02(.04)
	White	0(0)	0(0)	0(0)
	Red+white	0.12(.08)	0.92(.28)	1.04(.29)
June 30	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 8	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 15	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 22	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 29	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Aug 10	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)

Table 2. (Continued)

		Aphids/sweep		
Date	Clover	Apterae	Alatae	Total aphids
	Red+white	0(0)	0(0)	0(0)
Aug 19	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Aug 26	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
		Sapulpa, OK		
April 8	Red+white	5.34(3.78)	0.14(.13)	5.48(3.79)
April 15	Red+white	2.06(1.14)	0.02(.04)	2.08(1.10)
April 22	Red+white	3.68(2.89)	0.04(.09)	3.72(2.91)
April 29	Red+white	1.26(2.43)	0.08(.13)	1.34(2.55)
May 5	Red+white	0.74(.89)	0.30(.22)	1.04(.97)
May 13	Red+white	1.10(.58)	0(0)	1.10(.58)
May 20	Red+white	4.42(3.36)	0.02(.04)	4.44(3.33)
June 1	Red+white	1.58(2.18)	0.18(.13)	2.36(1.60)
June 16	Red+white	0.16(.09)	0(0)	0.16(.09)
June 24	Red+white	0.04(.09)	0(0)	0.04(.09)
June 30	Red+white	0.02(.04)	0(0)	0.02(.04)
July 8	Red+white	0(0)	0(0)	0(0)
July 15	Red+white	0(0)	0(0)	0(0)
Aug 3	Red+white	0.02(.04)	0(0)	0.02(.04)
Aug 10	Red+white	0(0)	0(0)	0(0)
Aug 19	Red+white	0(0)	0(0)	0(0)

[z] Mean (standard deviation).

Table 3. Legume aphid densities in 1994.

		Aphids/sweep		
Date	Clover	Apterae	Alatae	Total aphid
		Beggs, OK		
April 8	Red	0(0)[z]	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
April 19	Red	0.1(.10)	0.04(.05)	0.14(.13)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
April 27	Red	0.82(.58)	0.06(.13)	0.88(.55)
	White	0.52(.17)	0.06(.09)	0.58(.14)
	Red+white	0.46(.21)	0.02(.04)	0.48(.20)
May 13	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
May 19	Red	0.06(.09)	0(0)	0.06(.09)
	White	0(0)	0(0)	0(0)
	Red+white	0.60(.31)	0(0)	0.60(.31)
May 26	Red	0.30(.18)	0(0)	0.30(.17)
	White	0.84(.98)	0.02(.04)	0.86(.97)
	Red+white	6.66(6.40)	0.10(.14)	6.76(6.32)
June 3	Red	0.30(.07)	0(0)	0.30(0.07)
	White	0.66(.45)	0.04(.09)	0.70(.50)
	Red+white	5.46(4.61)	0(0)	5.46(4.61)
June 9	Red	0.20(.12)	0(0)	0.20(.12)
	White	0.58(.39)	0(0)	0.70(.28)
	Red+white	0.54(.76)	0.12(.13)	0.54(.76)
June 21	Red	0(0)	0(0)	0(0)
	White	0.04(.09)	0(0)	0.10(.14)
	Red+white	0.22(.08)	0.06(.13)	0.22(.08)
July 8	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
July 28	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Aug 8	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)

Table 3. (Continued)

Date	Clover	Aphids/sweep		
		Apterae	Alatae	Total aphid
	Red+white	0(0)	0(0)	0(0)
Aug 19	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Sept 6	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
Sept 20	Red	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)
		<u>Sapulpa, OK</u>		
April 8	Red+white	0.62(.67)	0.18(.11)	0.80(.57)
April 19	Red+white	0.26(.19)	0.04(.09)	0.30(.26)
April 26	Red+white	1.68(.87)	0(0)	1.68(.87)
May 5	Red+white	0.47(.28)	0(0)	0.47(.28)
May 12	Red+white	0.12(.18)	0(0)	0.12(.18)
May 17	Red+white	0.12(.16)	0.10(.10)	0.22(.24)
May 24	Red+white	0.06(.13)	0(0)	0.06(.13)
June 2	Red+white	3.70(4.32)	0.16(.16)	3.86(4.31)
June 7	Red+white	2.52(1.49)	0.22(.15)	2.74(1.40)
June20	Red+white	0.02(.04)	0(0)	0.02(.04)
July 7	Red+white	0(0)	0(0)	0(0)
July 21	Red+white	0(0)	0(0)	0(0)
Aug 4	Red+white	0(0)	0(0)	0(0)
Aug 15	Red+white	0(0)	0(0)	0(0)
Aug 30	Red+white	0(0)	0(0)	0(0)
Sept 13	Red+white	0(0)	0(0)	0(0)
Sept 27	Red+white	0(0)	0(0)	0(0)

[z] Mean (standard deviation).

Table 4. Lady beetle densities on legumes in 1992 at Beggs.

Date	Clover	Lady beetle adults/sweep					
		Hippodamia convergens	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
April 8	Red	0.08(.08)[z]	0.02(.04)	0.2(.23)	0(0)	0.30(.25)	0.42(.46)
	White	0.02(.04)	0(0)	0.46(.39)	0(0)	0.48(.36)	1.08(1.74)
	Red+white	0.06(.09)	0(0)	0.26(.22)	0(0)	0.32(.21)	0.76(1.09)
April 21	Red	0.22(.28)	0.08(.08)	0(0)	0(0)	0.30(.34)	0.18(.21)
	White	0.08(.04)	0.02(.04)	0.18(.16)	0(0)	0.28(.22)	1.70(1.91)
	Red+white	0.06(.09)	0(0)	0.08(.11)	0(0)	0.14(.13)	0.28(.57)
May 5	Red	0.10(.08)	0.10(.08)	0.02(.05)	0(0)	0.22(.12)	0(0)
	White	0.10(.10)	0.06(.09)	0.06(.09)	0(0)	0.22(.18)	0.22(.21)
	Red+white	0.06(.05)	0.04(.05)	0.04(.05)	0(0)	0.14(.09)	0.38(.25)
May 22	Red	0.30(.30)	0.06(.05)	0.26(.36)	0.10(.14)	0.72(.47)	0.46(.45)
	White	0.44(.45)	0.12(.08)	0.54(.39)	0.06(.05)	1.16(.83)	0.56(.84)
	Red+white	0.04(.05)	0.02(.04)	0.36(.23)	0.12(.13)	0.54(.19)	0.76(.61)
June 17	Red	0.12(.17)	0(0)	0.08(.13)	0.06(.05)	0.26(.15)	0.26(.16)
	White	0.30(.41)	0.06(.09)	0.12(.11)	0.02(.04)	0.50(.42)	0.34(.28)
	Red+white	0.16(.19)	0(0)	0.14(.05)	0(0)	0.30(.32)	0.44(.35)
July 8	Red	0(0)	0.02(.05)	0.25(.23)	0(0)	0.27(.29)	0.07(.09)
	White	0(0)	0(0)	0.36(.38)	0(0)	0.36(.38)	0.24(.18)
	Red+white	0(0)	0.02(.04)	0.54(.32)	0(0)	0.56(.32)	0.06(.13)
July 24	Red	0(0)	0(0)	0.06(.09)	0(0)	0.06(.09)	0(0)
	White	0(0)	0(0)	0.20(.21)	0(0)	0.20(.21)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)

Table 4. (Continued)

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
Aug 14	Red	0(0)	0(0)	0.04(.05)	0.02(.04)	0.06(.05)	0.12(.13)
	White	0(0)	0(0)	0.14(.11)	0(0)	0.14(.11)	0.24(.25)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0.06(.09)
Aug 28	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Sept 15	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Oct 1	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0.02(.04)	0(0)	0(0)	0(0)	0.02(.04)	0(0)
	Red+white	0.02(.04)	0(0)	0(0)	0(0)	0.02(.04)	0(0)
Oct 12	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)

[z] Mean (standard deviation).

Table 5. Lady beetle densities on legumes in 1993.

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
Beggs, OK							
April 15	Red	0(0)[z]	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0.08(.11)	0.02(.04)	0.10(.10)	0(0)
April 22	Red	0.02(.04)	0(0)	0.02(.04)	0(0)	0.04(.05)	0(0)
	White	0(0)	0(0)	0.58(.54)	0.02(.04)	0.60(.53)	0(0)
	Red+white	0(0)	0(0)	0.04(.05)	0(0)	0.04(.05)	0(0)
June 1	Red	0.10(.17)	0.02(.04)	0.10(.10)	0.02(.04)	0.24(.15)	0(0)
	White	0(0)	0(0)	0.18(.29)	0.26(.15)	0.18(.29)	0(0)
	Red+white	0.04(.05)	0(0)	0.10(.07)	0.06(.09)	0.20(.10)	0(0)
June 8	Red	0.02(.04)	0.02(.04)	0.16(.21)	0.04(.05)	0.24(.20)	1.06(.48)
	White	0(0)	0(0)	0.38(.19)	0.02(.04)	0.40(.20)	0.12(.13)
	Red+white	0(0)	0.02(.04)	0.22(.13)	0.02(.04)	0(0)	0.72(.30)
June 16	Red	0(0)	0.02(.04)	0.24(.15)	0.04(.05)	0.30(.20)	0.10(.14)
	White	0(0)	0(0)	0.26(.27)	0(0)	0.26(.27)	0.24(.53)
	Red+white	0.08(.08)	0.02(.04)	0.40(.43)	0.08(.08)	0.58(.42)	0.52(.55)
June 24	Red	0.06(.13)	0(0)	1.22(1.24)	0.26(.42)	1.54(1.22)	0.18(.29)
	White	0.02(.04)	0(0)	0.42(.39)	0.08(.13)	0.52(.50)	0.54(1.09)
	Red+white	0(0)	0(0)	0.72(.22)	0.18(.15)	0.90(.23)	1.00(.65)
June 30	Red	0.02(.04)	0.02(.04)	0.74(.58)	0.06(.05)	0.84(.59)	0.12(.17)
	White	0(0)	0(0)	0.34(.19)	0(0)	0.34(.19)	0.60(.20)

Table 5. (Continued)

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
	Red+white	0(0)	0(0)	0.44(.31)	0(0)	0.44(.31)	0.70(.25)
July 8	Red	0.02(.04)	0.26(.41)	0.42(.60)	0(0)	0.70(.61)	0.12(.16)
	White	0(0)	0.50(.33)	0.22(.38)	0(0)	0.72(.22)	0.40(.25)
	Red+white	0(0)	0.88(.52)	0.08(.13)	0(0)	0.96(.41)	0.32(.41)
July 15	Red	0(0)	0.10(.12)	0.24(.23)	0.04(.05)	0.38(.25)	0.08(.13)
	White	0(0)	0.02(.04)	0.18(.23)	0(0)	0.20(.22)	0.10(.14)
	Red+white	0(0)	0(0)	0.18(.19)	0(0)	0.18(.19)	0.04(.05)
July 22	Red	0.02(.04)	0.10(.12)	0.84(.79)	0.02(.04)	0.98(.87)	0.32(.13)
	White	0(0)	0(0)	1.12(.98)	0.02(.04)	1.14(1.02)	0.30(.14)
	Red+white	0(0)	0(0)	0.76(.71)	0(0)	0.76(.71)	0.06(.05)
July 29	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Aug 10	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0(0)
Aug 19	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0.02(.04)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0(0)
Aug 26	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)

Table 5. (Continued)

Date	Clover	Lady beetle adults/sweep *Hippodamia convergens*	 *Coccinella semptempunctata*	 *Coleomegilla maculata lengi*	 *Cycloneda munda*	 Total	Lady beetle larvae
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
				<u>Sapulpa, OK</u>			
April 8	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
April 15	Red+white	0(0)	0(0)	0.04(.05)	0(0)	0.04(.05)	0(0)
April 22	Red+white	0(0)	0.02(.04)	0.12(.13)	0(0)	0.14(.11)	0(0)
April 29	Red+white	0.08(.13)	0(0)	0.06(.13)	0(0)	0.14(.26)	0(0)
May 5	Red+white	0.04(.05)	0(0)	0.08(.08)	0.02(.04)	0.14(.16)	0(0)
May 13	Red+white	0.02(.04)	0(0)	0.32(.22)	0.04(.05)	0.38(.26)	0(0)
May 20	Red+white	0.04(.05)	0.02(.04)	0.30(.29)	0(0)	0.36(.32)	0(0)
June 1	Red+white	0.02(.04)	0(0)	0.04(.05)	0(0)	0.06(.09)	0(0)
June 16	Red+white	0(0)	0(0)	0.34(.15)	0(0)	0.34(.15)	0(0)
June 24	Red+white	0(0)	0.02(.04)	0.20(.23)	0(0)	0.22(.22)	0(0)
June 30	Red+white	0.04(.05)	0(0)	0.04(.05)	0(0)	0.08(.08)	0(0)
July 8	Red+white	0(0)	0(0)	0.02(.04)	0.04(.05)	0.06(.05)	0.06(.09)
July 15	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Aug 3	Red+white	0(0)	0.02(.04)	0.14(.15)	0.02(.04)	0.18(.16)	0(0)
Aug 10	Red+white	0(0)	0(0)	0.12(.11)	0(0)	0.12(.11)	0.02(.04)
Aug 19	Red+white	0(0)	0.02(.04)	0.06(.09)	0(0)	0.08(.13)	0(0)

[z] Mean (standard deviation).

Table 6. Arthropod densities in pecan canopies in 1993.

		Arthropods/m^2					
Date	Treatment	Green lacewing adult	Green lacewing larvae	Spider	Assassin bugs	Lady beetle larvae	Lady beetle adults
			Beggs, OK				
June 8	Red clover	0[z]	0	0.8	0	0	0.8
	White clover	0	0	2.0	0	0	0.4
	Red+white clover	0	0	1.2	0	0.4	0
	Grass	0.8	0	2.0	0	0	0
June 16	Red clover	0.4	0	0	0	0	0
	White clover	0.4	0	0.4	0	0	0.8
	Red+white clover	0.4	0	1.6	0	0.4	1.2
	Grass	0.4	0	0	0	0	1.6
June 24	Red clover	0	0	1.2	0	0	0.8
	White clover	0	0.4	0.4	0	0	1.2
	Red+white clover	0.4	0	2.0	0	0	0
	Grass	0	0	0.4	0	0	1.6
June 30	Red clover	0	0	0	0	0	0
	White clover	0	0	0	0	0	0.8
	Red+white clover	0	0	1.2	0	0	0.4
	Grass	0.4	0	0.8	0	0	0.8
July 8	Red clover	0	0	2.0	0	0	0.4
	White clover	0.4	0.4	2.4	0	0	2.4
	Red+white clover	0	0.8	1.6	0	0	0
	Grass	1.6	0	2.0	0	0	0.8
July 15	Red clover	0	0	2.0	0	0	0.4
	White clover	0	0	2.8	0	0	0.8
	Red+white clover	0	0	3.2	0	0	0
	Grass	0	0	1.6	0	0	0
July 22	Red clover	0	0	0	0	0	0.4
	White clover	0	2.0	0	0	0	0.4
	Red+white clover	0	0	5.6	0	0	0
	Grass	0.4	0	2.8	0.8	0	0.4
July 29	Red clover	0	0	0.4	0	0	0
	White clover	0	0	0.8	0	0	0
	Red+white clover	0	0	1.2	0	0	0
	Grass	0	0	3.2	0	0	0.4
Aug 10	Red clover	0	0	0.8	0	0	0
	White clover	0	0	1.6	0	0	0
	Red+white clover	0	0	2.8	0	0	0
	Grass	0	0	3.6	0	0	0.4
Aug 19	Red clover	0	0	0	0	0	0
	White clover	0	0	0	0	0	0

Table 6. (Continued).

		Arthropods/m^2					
Date	Treatment	Green lacewing adult	Green lacewing larvae	Spider	Assassin bugs	Lady beetle larvae	Lady beetle adults
Aug 19	Red+white clover	0.4	0	0.4	0	0	0
	Grass	0.8	0	0.4	0	0	0
Aug 26	Red clover	0.4	0	1.6	0	0	0
	White clover	0	0	0.4	0	0	0
	Red+white clover	0	0	0.8	0	0	0.4
	Grass	0.8	0	1.6	0	0	0
			Sapulpa. OK				
May 20	Red+white clover	0	0	0	0	0	0
	Grass	0	0	0.8	0	0	0
May 25	Red+white clover	0	0	1.2	0	0	0
	Grass	0	0	2.8	0	0.4	0
June1	Red+white clover	0.4	0	2.4	0	0	0
	Grass	0.4	0	0.8	0	0	0
June 8	Red+white clover	1.6	0	3.6	0	0	0.4
	Grass	0	0	1.8	0.4	0	0.4
June 24	Red+white clover	0	0	3.6	0	0	0
	Grass	0	0	1.2	0.4	0	0.4
June 30	Red+white clover	0	0	1.6	0	0	0
	Grass	0.4	0	1.6	0	0	0
July 8	Red+white clover	1.2	0	2.4	0	0	0.4
	Grass	0	0.4	0.4	0	0	0.8
July 13	Red+white clover	0.4	0	0	0	0	0
	Grass	0	0	1.2	0.4	0	0
July 18	Red+white clover	0	0	0.8	0	0	0
	Grass	0	0.4	2.4	0	0	0
Aug 2	Red+white clover	0	0	6.0	0.4	0	0
	Grass	0.4	0	1.6	1.2	0	0
Aug 18	Red+white clover	0	0	0.8	0	0	0
	Grass	1.2	0	0.8	0	0	0

[z] Treatments were not significantly different at the 5% level by Fishers F-test.

Table 7. Lady beetle densities on legumes in 1994.

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
				Beggs, OK			
April 8	Red	0(0)^z	0(0)	0(0)	0.02(.04)	0.02(.04)	0.02(.04)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0.04(.09)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0.42(.34)
April 19	Red	0(0)	0.02(.04)	0(0)	0(0)	0.02(.04)	0(0)
	White	0(0)	0.06(.05)	0(0)	0(0)	0.06(.05)	0.06(.05)
	Red+white	0(0)	0.02(.04)	0(0)	0(0)	0.02(.04)	0(0)
April 27	Red	0.22(.18)	0.48(.47)	0(0)	0.04(.09)	0.74(.66)	0.14(.11)
	White	0.06(.09)	0.10(.12)	0(0)	0(0)	0.16(.19)	0(0)
	Red+white	0.02(.04)	0.16(18)	0(0)	0(0)	0.18(.21)	0.02(.04)
May 13	Red	0.08(.11)	0.14(.11)	0.02(.04)	0.02(.04)	0.26(.05)	0.08(.08)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0.14(.15)
	Red+white	0.02(.04)	0.02(.04)	0.02(.04)	0(0)	0.06(.09)	0.18(.16)
May 19	Red	0.04(.05)	0(0)	0(0)	0(0)	0.04(.05)	0.14(13)
	White	0(0)	0.02(.04)	0.06(.09)	0(0)	0.08(.13)	0.06(.13)
	Red+white	0(0)	0.02(.04)	0.02(.04)	0(0)	0.04(.05)	0.04(.09)
May 26	Red	0.12(.16)	0.06(.09)	0.04(.05)	0(0)	0.22(.20)	0.08(.11)
	White	0(0)	0.04(.05)	0.28(.38)	0(0)	0.32(.41)	0.04(.09)
	Red+white	0.02(.04)	0.04(.05)	0.24(.16)	0.02(.04)	0.34(.21)	0.08(.11)
June 3	Red	0.02(.04)	0(0)	0.02(.04)	0(0)	0.04(.09)	0.14(.16)
	White	0(0)	0(0)	0.04(.05)	0(0)	0.04(.05)	0.04(.09)
	Red+white	0(0)	0(0)	0.04(.05)	0.04(.05)	0.08(.11)	0.12(.21)

Table 7. (Continued)

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
June 9	Red	0.04(.05)	0.02(.04)	0.14(.11)	0.08(.08)	0.28(.16)	0.06(.09)
	White	0.04(.05)	0.08(.13)	0.20(.16)	0.04(.05)	0.36(27)	0.28(.26)
	Red+white	0.02(.04)	0.02(.04)	0.18(.21)	0.04(.09)	0.26(.22)	0.14(.19)
June 21	Red	0(0)	0.04(.05)	0.78(.82)	0.16(.18)	0.98(.95)	0.16(.30)
	White	0.02(.04)	0.06(.09)	0.36(.47)	0.12(.16)	0.56(.41)	0.20(.21)
	Red+white	0.08(.11)	0.02(.04)	0.98(.46)	0.32(.56)	1.42(1.13)	0.74(.41)
July 8	Red	0.02(.04)	0.04(.09)	0.16(.18)	0.12(.13)	0.34(.33)	0.06(.13)
	White	0(0)	0.02(.04)	0.16(.15)	0(0)	0.18(.16)	0(0)
	Red+white	0(0)	0(0)	0.34(29)	0.06(.09)	0.40(.36)	0(0)
July 28	Red	0(0)	0.02(.04)	0.06(.09)	0(0)	0.08(.08)	0(0)
	White	0(0)	0(0)	0(0)	0.02(.04)	0.02(.04)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.02(.04)	0(0)
Aug 8	Red	0(0)	0.02(.04)	0.02(.04)	0.02(.04)	0.06(.09)	0(0)
	White	0(0)	0(0)	0.14(.09)	0(0)	0.14(.09)	0(0)
	Red+white	0(0)	0(0)	0.06(.09)	0(0)	0.06(.09)	0(0)
Aug 19	Red	0(0)	0.02(.04)	0.04(.05)	0.02(.04)	0.08(.04)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Sept 6	Red	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Sept 20	Red	0(0)	0(0)	0(0)	0.04(.05)	0.04(.05)	0(0)
	White	0(0)	0(0)	0(0)	0.02(.04)	0.02(.04)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)

Table 7. (Continued)

		Lady beetle adults/sweep					
Date	Clover	*Hippodamia convergens*	*Coccinella semptempunctata*	*Coleomegilla maculata lengi*	*Cycloneda munda*	Total	Lady beetle larvae
				<u>Sapulpa, OK</u>			
April 8	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.02(.04)	0.18(.20)
April 19	Red+white	0(0)	0.12(.21)	0(0)	0(0)	0.12(.21)	0.14(.05)
April 26	Red+white	0.08(.11)	0.96(.51)	0.02(.04)	0(0)	11.16(22.89)	0.16(.20)
May 5	Red+white	0.05(.10)	0.20(.20)	0.02(.05)	0(0)	0.27(.17)	0.12(.09)
May 12	Red+white	0.02(.04)	0.04(.09)	0.08(.13)	0(0)	0.14(.11)	0(0)
May 17	Red+white	0.02(.04)	0.06(.05)	0(0)	0(0)	0.08(.08)	0.06(.13)
May 24	Red+white	0.02(.04)	0(0)	0.02(.04)	0.08(.04)	0.12(.08)	0(0)
June 2	Red+white	0.04(.05)	0(0)	0.16(.11)	0(0)	0.20(.16)	0.04(.05)
June 7	Red+white	0.10(.22)	0.04(.05)	0.16(.11)	0.02(.04)	0.32(.29)	0.02(.04)
June 20	Red+white	0.04(.09)	0.02(.04)	0.06(.13)	0.02(.04)	0.14(.11)	0.26(.26)
July 7	Red+white	0(0)	0(0)	0.26(.19)	0.04(.05)	0.34(.27)	0(0)
July 21	Red+white	0(0)	0(0)	0.10(.07)	0.06(.09)	0.16(.05)	0.02(.04)
Aug 4	Red+white	0(0)	0(0)	0.04(.05)	0.02(.04)	0.06(.05)	0(0)
Aug 15	Red+white	0(0)	0(0)	0(0)	0.06(.05)	0.06(.05)	0(0)
Aug 30	Red+white	0(0)	0(0)	0.12(.11)	0.04(.09)	0.16(.09)	0(0)
Sept 13	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
Sept 27	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)

[z] Mean (standard deviation).

Table 8. Arthropod densities in pecan canopies in 1994.

		Arthropods/m^2				
Date	Treatment	Green lacewing adult	Green lacewing larvae	Spider	Assassin bugs	Lady beetle adults
			Beggs, OK			
April 27	Red clover	0[z]	0	0.4	0	0
	White clover	0	0	1.6	0	0.4
	Red+white clover	0	0	0.8	0	0
	Grass	0	0	0	0	0
May 19	Red clover	0	0	2.4	0	0.8
	White clover	0	0.8	1.2	0	0.4
	Red+white clover	0.8	0	2.4	0	0.4
	Grass	0.4	0	2.8	0	1.2
May 26	Red clover	2.8	0	1.6	0	1.2
	White clover	0	0	0.8	0	0.4
	Red+white clover	0.4	0	3.2	0	2.0
	Grass	0.4	0	2.0	0	0.8
June 3	Red clover	0	0	1.2	0	0.8
	White clover	0.4	0	2.0	0	2.4
	Red+white clover	0	0	2.4	0	0.4
	Grass	0	0	2.0	0	0.8
June 9	Red clover	0	0	0.8	0	1.6
	White clover	3.2	0	0.4	0	2.8
	Red+white clover	4.0	0	0.8	0	1.2
	Grass	2.8	0	0	0	0.8
June 21	Red clover	0	0	2.4	0	2.0
	White clover	0	0	0.8	0	4.8
	Red+white clover	0	0	2.4	0	2.0
	Grass	0.4	0	2.8	0	0.4
July 8	Red clover	0.4	0	1.6	0.4	2.4
	White clover	0	0	1.6	0	0.8
	Red+white clover	0	0	0	0	0
	Grass	0	0	1.6	1.2	0
July 28	Red clover	0	0	0.4	0	0.4
	White clover	0	0	2.0	0	0.4
	Red+white clover	0	0	2.8	0.4	0.8
	Grass	0	0	1.6	0	0.4
Aug 8	Red clover	0	0	2.4	0	1.6
	White clover	0	0	1.2	0.8	3.2
	Red+white clover	0	0	1.6	0.8	2.0
	Grass	0	0	1.2	0.4	0.4
Aug 19	Red clover	0.4	0	1.6	0	2.4
	White clover	0	0	2.0	0	0.8
	Red+white clover	0	0	5.2	0	2.8
	Grass	0	0	1.6	0.8	0

Table 8. (Continued)

		Arthropods/m^2				
Date	Treatment	Green lacewing adult	Green lacewing larvae	Spider	Assassin bugs	Lady beetle adults
Sept 6	Red clover	0.4	0	1.2	0.4	0.4
	White clover	0.4	0	1.6	0.4	0.4
	Red+white clover	0	0	1.6	0	2.4
	Grass	0.4	0	3.2	0	0
Sept 20	Red clover	0	0	3.2	0	0
	White clover	0	0	1.6	0	0
	Red+white clover	0.4	0	2.0	0	0
	Grass	0	0	0.8	0	0
			Sapulpa, OK			
April 26	Red+white clover	0.4	0	0.4	0	2.4
	Grass	0.4	0	0.8	0	0.4
May 12	Red+white clover	0	0	2.3	0	0
	Grass	0	0	1.2	0	0
May 17	Red+white clover	0	0	2.0	0	0
	Grass	0	0	2.8	0	0.8
May 24	Red+white clover	2.4	0	1.6	0	0.8
	Grass	0	0	5.2	0	0.8
June 2	Red+white clover	0	0	3.2	0	0
	Grass	0.4	0	1.2	0	0.8
June 7	Red+white clover	0.4	0	1.2	0	0.8
	Grass	0.4	0	0.8	0	0.8
June 20	Red+white clover	0.4	0	1.6	0	0.8
	Grass	0	0	0.8	0	1.6
July 7	Red+white clover	0.8	0	1.6	0.8	0.4
	Grass	1.2	0	0	0.4	0.4
July 21	Red+white clover	0	0	2.0	0	1.2
	Grass	0.4	0	0.8	0	1.2
Aug 4	Red+white clover	0	0	1.6	0.4	0.4
	Grass	0	0	0.8	0	1.6
Aug 15	Red+white clover	0.4	0	2.8	0	0.8
	Grass	0.4	0	1.2	0.4	0.8
Aug 30	Red+white clover	0.4	0	1.6	0.4	0.8
	Grass	0.4	0	1.2	0.4	0.8
Sept 13	Red+white clover	0	0	0.8	0.4	0
	Grass	0	0	0.8	0.8	0
Sept 27	Red+white clover	0.4	0	2.8	0	0
	Grass	0	0	1.6	0.4	0

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[z] Treatments were not significantly different at the 5% level by Fishers F-test.

Table 9. Arthropod densities on legumes in 1992 at Beggs.

		Arthropods/sweep					
Date	Clover	Green lacewing adult	Green lacewing larvae	Predacious stink bugs	Phytophagous stink bugs	Spider	Nabid sp.
April 14	Red	0(0)[z]	0(0)	0(0)	0.08(.08)	0.16(.11)	0.20(.18)
	White	0(0)	0(0)	0.02(.04)	0.08(.04)	0.28(.24)	0.14(.11)
	Red+white	0.02(.04)	0(0)	0.02(.04)	0.06(.13)	0.50(.25)	0.06(.13)
April 21	Red	0(0)	0(0)	0(0)	0(0)	0.10(.10)	0(0)
	White	0(0)	0.04(.09)	0.02(.04)	0.04(.08)	0.20(.10)	0.10(.12)
	Red+white	0(0)	0.04(.09)	0.02(.04)	0.02(.04)	0.30(.14)	0.08(.08)
May 5	Red	0(0)	0(0)	0(0)	0(0)	0.45(.17)	0.10(.08)
	White	0.06(.09)	0.02(.04)	0(0)	0(0)	0.50(.34)	0.1(.07)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.50(.18)	0.04(.09)
May 22	Red	0(0)	0.02(.04)	0.04(.09)	0.02(.04)	0.82(.30)	0.68(.39)
	White	0.04(.05)	0.02(.04)	0(0)	0.18(.04)	0.52(.18)	0.88(.45)
	Red+white	0(0)	0.02(.04)	0.02(.04)	0.14(.08)	0.64(.27)	0.22(.08)
June 17	Red	0.06(.09)	0.02(.04)	0(0)	0.08(.11)	0.54(.52)	0.24(.18)
	White	0.12(.08)	0.02(.04)	0(0)	0.06(.13)	0.28(.24)	1.28(1.75)
	Red+white	0.04(.05)	0(0)	0.02(.04)	0.02(.04)	0.24(.11)	0.40(.40)
July 8	Red	0(0)	0(0)	0.07(.05)	0.10(.14)	0.90(.14)	0(0)
	White	0(0)	0(0)	0.04(.09)	0.04(.05)	0.76(.30)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0.04(.05)	1.10(.36)	0(0)
July 24	Red	0(0)	0(0)	0(0)	0.04(.05)	0.86(.41)	0(0)
	White	0(0)	0(0)	0(0)	0.04(.05)	1.38(1.29)	0.02(.04)
	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.32(.13)	0.20(.04)
Aug14	Red	0.04(.09)	0(0)	0.06(.13)	0.20(.14)	1.02(.18)	0.02(.04)
	White	0(0)	0(0)	0.12(.13)	0.24(.18)	0.80(.48)	0(0)
	Red+white	0(0)	0(0)	0.04(.09)	0.30(.10)	0.76(.25)	0(0)
Aug 28	Red	0(0)	0(0)	0.02(.04)	0.26(.23)	1.02(.30)	0(0)
	White	0(0)	0(0)	0(0)	0.04(.05)	0.72(.48)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.14(.11)	0.60(.30)	0(0)
Sept 15	Red	0.06(.09)	0.02(.04)	0.02(.04)	0.10(.07)	0.58(.39)	0.08(.08)
	White	0(0)	0(0)	0(0)	0.02(.04)	0.08(.08)	0.06(.05)
	Red+white	0.12(.11)	0(0)	0.02(.04)	0.04(.05)	0.66(.27)	0.06(.05)
Oct 1	Red	0(0)	0.02(.04)	0.02(.04)	0.08(.08)	0.40(.24)	0(0)
	White	0(0)	0(0)	0(0)	0.04(.05)	0.28(.22)	0(0)
	Red+white	0(0)	0.02(.04)	0.02(.04)	0.12(.08)	0.46(.18)	0.08(.04)
Oct 12	Red	0(0)	0(0)	0(0)	0(0)	0.40(.27)	0(0)
	White	0(0)	0(0)	0.02(.04)	0.02(.04)	0.20(.12)	0(0)
	Red+white	0(0)	0(0)	0.04(.09)	0.04(.05)	0.50(.36)	0(0)

[z] Mean (standard deviation).

Table 10. Arthropod densities in pecan canopies in 1992 at Beggs.

		Arthropods/m^2					
Date	Treatment	Green lacewing adult	Green lacewing larvae	Spider	Assassin bugs	Hover flies	Lady beetle adults
May 5	Red clover	0[z]	0	2.0	1.2	0.4	0
	White clover	0	0	1.2	0	0	0
	Red+white clover	0	0	1.6	0	0	0
	Grass	0	0	2.8	0	0	0
May 22	Red clover	0	0	0.5	0	0	0
	White clover	0	0	0.5	0	0	0
	Red+white clover	0	0	0.8	0	0	0
	Grass	0.4	0	0.4	0	0	0
June 17	Red clover	0.4	0	0.4	0	0.8	0
	White clover	0.4	0	2.4	0	0.4	0
	Red+white clover	0	0	1.2	0	0	0
	Grass	0	0	2.4	0	0	0
July 8	Red clover	0	0	0	0	0	0
	White clover	0	0	0.8	1.2	0.4	0
	Red+white clover	0	0	1.6	0.4	0	0
	Grass	0	0	1.6	0	0	0
July 24	Red clover	0	0	1.2	0	0	0
	White clover	0	0	1.2	0.4	0.8	0
	Red+white clover	0	0	2.0	0	0	0
	Grass	0	0	0	0.4	0	0
Aug 14	Red clover	1.6	0.4	3.6	0	0	0
	White clover	3.6	1.2	1.6	0.4	0	0
	Red+white clover	1.2	0.4	4.4	0	0	0
	Grass	2.4	0.8	5.2	0	0	0
Aug 28	Red clover	0	0	0.4	0	0	0
	White clover	0	0	2.4	0.4	0	0
	Red+white clover	0	0	0.4	0	0	0
	Grass	0	0	1.2	0	0	0
Sept 15	Red clover	1.2	0.4	0	0	0	0
	White clover	1.2	0	0	0	0	0
	Red+white clover	0.8	0	0	0	0	0
	Grass	1.2	0.4	0	0	0	0.8
Oct 1	Red clover	0	0	0	0	0	0
	White clover	0	0	0	0	0	0
	Red+white clover	3.6	0	0	0	0	0
	Grass	0.4	0	0.4	0	0	0
Oct 12	Red clover	0	0	0	0	0	0.4
	White clover	2.0	0	0	0	0	0
	Red+white clover	1.6	0	0	0	0	0
	Grass	0	0	0	0	0	0

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[z] Treatments were not significantly different at the 5% level by Fishers F-test.

Table 11. Arthropod densities on legumes in 1993.

		Arthropods/sweep					
Date	Clover	Green lacewing adult	Green lacewing larvae	Predacious stink bugs	Phytophagous stink bugs	Spider	Nabis sp.
				Beggs, OK			
April 15	Red	0(0)[z]	0(0)	0(0)	0(0)	0.14(.11)	0(0)
	White	0.02(.04)	0(0)	0(0)	0(0)	0.02(.04)	0(0)
	Red+white	0.04(.05)	0(0)	0(0)	0(0)	0.10(.14)	0(0)
April 22	Red	0.02(.04)	0(0)	0(0)	0(0)	0(0)	0(0)
	White	0.02(.04)	0(0)	0(0)	0(0)	0.06(.09)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
June 1	Red	0(0)	0(0)	0.02(.04)	0(0)	0.46(.18)	0.02(.04)
	White	0(0)	0(0)	0.02(.04)	0.02(.04)	0.30(.24)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0.02(.04)	0.54(.15)	0.02(.04)
June 8	Red	0(0)	0.04(.05)	0.08(.04)	0.10(.17)	0.44(.23)	0.04(.09)
	White	0(0)	0(0)	0.02(.04)	0(0)	0.78(.59)	0.22(.24)
	Red+white	0(0)	0.02(.04)	0.04(.09)	0(0)	0.68(.44)	0.04(.09)
June 16	Red	0(0)	0(0)	0.02(.04)	0.04(.05)	0.08(.13)	0(0)
	White	0(0)	0(0)	0.10(.17)	0.06(.13)	0.48(.18)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.38(.25)	0.06(.13)
June 24	Red	0.02(.04)	0(0)	0(0)	0.20(.12)	0.60(.31)	0.02(.04)
	White	0(0)	0(0)	0(0)	0.06(.13)	0.28(.21)	0.02(.04)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.46(.38)	0(0)
June 30	Red	0(0)	0(0)	0.02(.04)	0.16(.21)	0.58(.52)	0.04(.09)
	White	0.02(.04)	0(0)	0(0)	0.02(.04)	0.76(.51)	0(0)
	Red+white	0(0)	0(0)	0.04(.05)	0.02(.04)	0.90(.43)	0.24(.26)
July 8	Red	0(0)	0(0)	0.04(.09)	0.12(.11)	0.56(.23)	0.06(.09)
	White	0.10(.17)	0(0)	0.06(.09)	0.18(.13)	0.70(.71)	0.14(.15)
	Red+white	0.06(.05)	0(0)	0.02(.04)	0.20(.07)	1.12(.42)	0.18(.18)
July 15	Red	0.04(.05)	0(0)	0.04(.09)	0.20(.21)	0.70(.21)	0(0)
	White	0(0)	0(0)	0.06(.05)	0.06(.13)	0.74(.25)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.16(.09)	0.72(.41)	0(0)
July 22	Red	0.22(.33)	0.20(.29)	0.02(.04)	0.48(.27)	0.38(.74)	0.08(.13)
	White	0(0)	0.18(.34)	0(0)	0.40(.28)	0.80(.89)	0.12(.21)
	Red+white	0(0)	0.04(.09)	0(0)	0.26(.21)	1.94(.55)	0(0)
July 29	Red	0(0)	0(0)	0.06(.05)	0.24(.11)	0.04(.09)	0(0)
	White	0.02(.04)	0(0)	0(0)	0.38(.32)	0.36(.25)	0(0)
	Red+white	0(0)	0.04(.09)	0(0)	0.26(.21)	1.94(.55)	0(0)

Table 11. (Continued)

		Arthropods/sweep					
Date	Clover	Green lacewing adult	Green lacewing larvae	Predacious stink bugs	Phytophagous stink bugs	Spider	Nabis sp.
Aug 10	Red	0(0)	0(0)	0(0)	0.10(.14)	0.14(.09)	0(0)
	White	0(0)	0(0)	0(0)	0.06(.05)	0.30(.18)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.06(.09)	0.30(.24)	0(0)
Aug 19	Red	0(0)	0(0)	0.08(.08)	0.20(.20)	0.04(.05)	0(0)
	White	0(0)	0(0)	0.04(.09)	0.02(.04)	0.20(.17)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0.04(.05)	0.18(.08)	0(0)
Aug 26	Red	0(0)	0(0)	0.04(.09)	0.04(.05)	0.14(.05)	0(0)
	White	0(0)	0(0)	0(0)	0.04(.05)	0.06(.05)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.44(.45)	0(0)
Sapulpa,OK							
April 8	Red+white	0(0)	0(0)	0(0)	0(0)	0.24(.09)	0(0)
April 15	Red+white	0.02(.04)	0(0)	0(0)	0.02(.04)	0.12(.13)	0(0)
April 22	Red+white	0.02(.04)	0(0)	0(0)	0(0)	0.02(.04)	0.06(.13)
April 29	Red+white	0.06(.09)	0(0)	0.14(.26)	0.16(.21)	0.02(.04)	0(0)
May 5	Red+white	0.10(.07)	0(0)	0(0)	0.02(.04)	0.08(.08)	0.06(.09)
May 13	Red+white	0(0)	0(0)	0.12(.16)	0.02(.04)	0.06(.09)	0(0)
May 20	Red+white	0.06(.09)	0.02(.04)	0(0)	0(0)	0.20(.23)	0(0)
June 1	Red+white	0.02(.04)	0(0)	0(0)	0(0)	0.16(.05)	0(0)
June 16	Red+white	0(0)	0(0)	0.02(.04)	0.02(.04)	0.18(.14)	0.12(.08)
June 24	Red+white	0.02(.04)	0(0)	0.02(.04)	0.08(.08)	0.34(.18)	0.04(.05)
June 30	Red+white	0.02(.04)	0(0)	0(0)	0(0)	0.22(.14)	0.04(.05)
July 8	Red+white	0(0)	0(0)	0.04(.05)	0.02(.04)	0.24(.15)	0(0)
July 15	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.26(.31)	0(0)
Aug 3	Red+white	0.02(.04)	0(0)	0(0)	0.02(.04)	0.30(.17)	0(0)
Aug 10	Red+white	0(0)	0(0)	0(0)	0(0)	0.24(.18)	0(0)
Aug 19	Red+white	0(0)	0(0)	0.02(.04)	0.14(.05)	0.42(.38)	0(0)

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[z] Mean (standard deviation).

Table 12. Arthropod densities on legumes in 1994.

		Arthropods/sweep					
Date	Clover	Green lacewing adult	Green lacewing larvae	Predacious stink bugs	Phytophagous stink bugs	Spider	Nabis sp.
				Beggs, OK			
April 8	Red	0(0)[z]	0(0)	0(0)	0(0)	0.04(.05)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0(0)	0(0)
	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0(0)	0(0)
April 19	Red	0(0)	0(0)	0(0)	0(0)	0.20(.14)	0(0)
	White	0(0)	0(0)	0(0)	0.02(.04)	0.18(.13)	0.02(.04)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.84(.61)	0(0)
April 27	Red	0.04(.05)	0(0)	0(0)	0(0)	0.44(.19)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0.10(.10)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.02(.04)	0(0)
May 13	Red	0(0)	0(0)	0(0)	0(0)	0.14(.09)	0.12(.04)
	White	0(0)	0(0)	0(0)	0(0)	0.20(.07)	0.12(.13)
	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.26(.13)	0.08(.13)
May 19	Red	0.02(.04)	0(0)	0(0)	0.02(.04)	0.20(.10)	0(0)
	White	0.02(.04)	0(0)	0(0)	0(0)	0.42(.27)	0.26(.15)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.24(.28)	0(0)
May 26	Red	0(0)	0(0)	0.04(.05)	0.02(.04)	0.16(.22)	0.10(.14)
	White	0(0)	0(0)	0(0)	0(0)	0.38(.08)	0.20(.16)
	Red+white	0(0)	0(0)	0.04(.09)	0.04(.05)	0.20(.07)	0.28(.24)
June 3	Red	0.02(.04)	0(0)	0(0)	0(0)	0.20(.12)	0.12(.08)
	White	0(0)	0(0)	0.08(.11)	0.02(.04)	0.18(.18)	0.36(.27)
	Red+white	0.02(.04)	0(0)	0.02(.04)	0.02(.04)	0.24(.16)	0.28(.26)
June 9	Red	0(0)	0(0)	0(0)	0(0)	0.24(.11)	0(0)
	White	0(0)	0(0)	0.06(.13)	0(0)	0.26(.35)	0.24(.29)
	Red+white	0(0)	0(0)	0.10(.10)	0.04(.05)	0.14(.05)	0.74(.49)
June 21	Red	0(0)	0(0)	0.04(.05)	0.02(.04)	0.42(.27)	0.12(.13)
	White	0(0)	0(0)	0.14(.19)	0.10(.17)	0.22(.13)	0.04(.05)
	Red+white	0.04(.05)	0(0)	0.04(.05)	0(0)	0.52(.19)	0.60(.39)
July 8	Red	0(0)	0(0)	0.02(.04)	0.14(.11)	1.02(.32)	0(0)
	White	0(0)	0(0)	0(0)	0.08(.04)	1.40(.34)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.10(.10)	0.98(.26)	0(0)
July 28	Red	0(0)	0(0)	0(0)	0.12(.16)	0.80(.42)	0(0)
	White	0(0)	0(0)	0(0)	0.02(.04)	0.54(.39)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.92(.71)	0(0)

Table 12. (Continued)

		Arthropods/sweep					
Date	Clover	Green lacewing adult	Green lacewing larvae	Predacious stink bugs	Phytophagous stink bugs	Spider	Nabis sp.
Aug 8	Red	0(0)	0(0)	0(0)	0.04(.05)	0.34(.05)	0.02(.04)
	White	0(0)	0(0)	0(0)	0(0)	0.46(.27)	0(0)
	Red+white	0(0)	0(0)	0.04(.05)	0(0)	1.02(..45)	0(0)
Aug 19	Red	0(0)	0(0)	0.02(.04)	0.02(.04)	0.50(.07)	0(0)
	White	0(0)	0(0)	0(0)	0.04(.05)	0.50(.31)	0(0)
	Red+white	0(0)	0(0)	0(0)	0.04(.09)	1.04(.30)	0(0)
Sept 6	Red	0(0)	0(0)	0(0)	0(0)	0.20(.10)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0.08(.08)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.12(.04)	0(0)
Sept 20	Red	0(0)	0(0)	0.02(.04)	0(0)	0.10(.07)	0(0)
	White	0(0)	0(0)	0(0)	0(0)	0.08(.04)	0(0)
	Red+white	0(0)	0(0)	0(0)	0(0)	0.24(.11)	0(0)
Sapulpa,OK							
April 8	Red+white	0.04(.05)	0(0)	0(0)	0(0)	0.12(.21)	0.02(.04)
April 19	Red+white	0(0)	0(0)	0(0)	0(0)	0.22(.21)	0.08(.08)
April 26	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.16(.09)	0.18(.21)
May 5	Red+white	0(0)	0(0)	0.02(.05)	0(0)	0.20(.18)	0.08(.11)
May 12	Red+white	0(0)	0(0)	0(0)	0(0)	0.48(.18)	0.08(.08)
May 17	Red+white	0(0)	0(0)	0.04(.09)	0(0)	0.18(.11)	0(0)
May 24	Red+white	0(0)	0(0)	0(0)	0(0)	0.22(.16)	0(0)
June 2	Red+white	0(0)	0(0)	0.10(.10)	0(0)	0.26(.22)	0.34(.25)
June 7	Red+white	0.02(.04)	0(0)	0.04(.09)	0(0)	0.04(.05)	0.36(.27)
June 20	Red+white	0.02(.04)	0(0)	0(0)	0(0)	0.32(.19)	0.22(.23)
July 7	Red+white	0(0)	0(0)	0(0)	0.02(.04)	0.94(38)	0.18(.21)
July 21	Red+white	0(0)	0(0)	0(0)	0(0)	0.76(.26)	0.02(.04)
Aug 4	Red+white	0(0)	0(0)	0.02(.04)	0(0)	0.44(.23)	0(0)
Aug 15	Red+white	0(0)	0(0)	0.02(.04)	0.02(.04)	0.30(.07)	0(0)
Aug 30	Red+white	0.02(.04)	0.02(.04)	0(0)	0.04(.05)	0.10(.07)	0(0)
Sept 13	Red+white	0(0)	0(0)	0(0)	0(0)	0.14(.05)	0(0)
Sept 27	Red+white	0(0)	0(0)	0(0)	0(0)	0.18(.16)	0(0)

^{z} Mean (standard deviation).

Table 13. Phytophagous stinkbug damage to pecan kernels.

		Kernels damaged (%)		
Location	Treatmeant[z]	1992	1993	1994
Beggs	Red clover	-	0	3.7^{a}
	White clover	-	0.2	4.0^{a}
	Red+white clover	12^{a}	0	1.0^{b}
	Grass	3.5^{b}	0	0.5^{b}
			NS	
Sapulpa	Red+white clover		0.2	0.2^{b}
	Grass		0.2	4.0^{a}
			NS	

[z] Means within columns followed by the same letter are not significantly different at 5 % level by LSD.

Table 14. Aphids per compound pecan leaf.

1992 Date	Treatment	Aphids/leaf	1993 Date	Treatment	Aphids/leaf	1994 Date	Treatment	Aphids/leaf
				<u>Beggs, OK</u>				
May 5	Red clover	0[z]	June 8	Red clover	0.8	April 27	Red clover	0.3^{a}
	White clover	0		White clover	1.3		White clover	0.4^{a}
	Red+white clover	0		Red+white clover	1.1		Red+white clover	0.5^{a}
	Grass	0		Grass	0.8		Grass	0.5^{a}
		NS			NS			
May 22	Red clover	0.1	June 16	Red clover	1.18	May 19	Red clover	7.7^{a}
	White clover	1.0		White clover	0.7		White clover	4.8^{ab}
	Red+white clover	0.3		Red+white clover	1.0		Red+white clover	5.5^{ab}
	Grass	0.3		Grass	0.4		Grass	3.1^{b}
		NS			NS			
June 17	Red clover	0.1	June 24	Red clover	0.4	May 26	Red clover	15.5^{a}
	White clover	0		White clover	0.2		White clover	17.5^{a}
	Red+white clover	0		Red+white clover	1.7		Red+white clover	7.7^{b}
	Grass	0.1		Grass	0.1		Grass	3.0^{c}
		NS			NS			
July 8	Red clover	0	July 1	Red clover	2.1	June 3	Red clover	5.2^{a}
	White clover	0		White clover	1.1		White clover	9.8^{a}
	Red+white clover	0		Red+white clover	1.8		Red+white clover	6.7^{a}
	Grass	0		Grass	1.1		Grass	6.4^{a}
		NS			NS			
July 24	Red clover	0	July 8	Red clover	0.1	June 9	Red clover	9.1^{c}
	White clover	0		White clover	0		White clover	16.4^{a}
	Red+white clover	0		Red+white clover	0.1		Red+white clover	14.0^{b}
	Grass	0		Grass	0		Grass	8.3^{c}
		NS			NS			

Table 14. (Continued)

1992 Date	Treatment	Aphids/leaf	1993 Date	Treatment	Aphids/leaf	1994 Date	Treatment	Aphids/leaf
Aug 14	Red clover	0.1	July 15	Red clover	0	June 21	Red clover	5.8^{a}
	White clover	0		White clover	0		White clover	4.2^{ab}
	Red+white clover	0		Red+white clover	0		Red+white clover	2.6^{b}
	Grass	0		Grass	0		Grass	2.7^{ab}
		NS			NS			
Aug 28	Red clover	1.6	July 22	Red clover	0	July 8	Red clover	0^{a}
	White clover	1.1		White clover	0		White clover	0^{a}
	Red+white clover	5.2		Red+white clover	0		Red+white clover	0^{a}
	Grass	1.0		Grass	0		Grass	0^{a}
		NS			NS			
Sept 15	Red clover	9.1	July 29	Red clover	0	July 28	Red clover	0.1^{a}
	White clover	5.0		White clover	0		White clover	0^{a}
	Red+white clover	12.2		Red+white clover	0		Red+white clover	0^{a}
	Grass	7.4		Grass	0		Grass	0^{a}
		NS			NS			
Oct 1	Red clover	1.8	Aug 10	Red clover	0	Aug 8	Red clover	0^{a}
	White clover	0.3		White clover	0		White clover	0^{a}
	Red+white clover	0		Red+white clover	0		Red+white clover	0^{a}
	Grass	0.7		Grass	0		Grass	0^{a}
		NS			NS			
Oct 12	Red clover	0	Aug 18	Red clover	0	Aug 19	Red clover	0^{a}
	White clover	0.3		White clover	0		White clover	0^{a}
	Red+white clover	0.1		Red+white clover	0		Red+white clover	0^{a}
	Grass	0.1		Grass	0		Grass	0^{a}
		NS			NS			

Table 14. (Continued)

1992 Date	Treatment	Aphids/leaf	1993 Date	Treatment	Aphids/leaf	1994 Date	Treatment	Aphids/leaf
			Aug 26	Red clover	0.1	Sept 6	Red clover	0^a
				White clover	0		White clover	0^a
				Red+white clover	0		Red+white clover	0^a
				Grass	0		Grass	0^a
					NS			
						Sept 20	Red clover	0.1^a
							White clover	0^a
							Red+white clover	0^a
							Grass	0^a
					<u>Sapulpa, OK</u>			
			May 20	Red+white clover	0.5	April 26	Red+white clover	0.6^b
				Grass	1.2		Grass	2.6^a
					NS			
			June 1	Red+white clover	0.4	May 12	Red+white clover	0.8^a
				Grass	0.3		Grass	0.6^a
					NS			
			June 8	Red+white clover	1.8	May 17	Red+white clover	1.0^a
				Grass	0.3		Grass	1.1^a
					NS			
			June 24	Red+white clover	1.1	May 24	Red+white clover	4.8^a
				Grass	1.8		Grass	7.9^a
					NS			
			June 30	Red+white clover	1.4	June 2	Red+white clover	6.6^a
				Grass	3.9		Grass	11.6^a
					NS			
			July 8	Red+white clover	0.3	June 7	Red+white clover	0.2^a
				Grass	0.4		Grass	0^a
					NS			

Table 14. (Continued)

1992 Date	Treatment	Aphids/leaf	1993 Date	Treatment	Aphids/leaf	1994 Date	Treatment	Aphids/leaf
			July 15	Red+white clover	0.1	June 20	Red+white clover	2.36^{b}
				Grass	0.1		Grass	4.52^{a}
					NS			
			July 18	Red+white clover	0.1	July 7	Red+white clover	0.16^{a}
				Grass	0		Grass	0.04^{a}
					NS			
			Aug 2	Red+white clover	0	July 21	Red+white clover	0.1^{a}
				Grass	0		Grass	0.1^{a}
					NS			
			Aug 10	Red+white clover	0	Aug 4	Red+white clover	0^{a}
				Grass	0		Grass	0^{a}
					NS			
			Aug 18	Red+white clover	0	Aug 15	Red+white clover	0^{a}
				Grass	0		Grass	0^{a}
						Aug 30	Red+white clover	0^{a}
							Grass	0^{a}
						Sept 13	Red+white clover	0.1^{a}
							Grass	0.1^{a}
						Sept 27	Red+white clover	0.2^{a}
							Grass	0.1^{a}

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[z] Means within dates and columns followed by the same letter are not significantly different at the 5% level by LSD.
NS, Non-significant at 5% level.

Table 15. Green lacewing eggs per compound pecan leaf.

1992 Date	Treatment	Green lacewing eggs/leaf	1993 Date	Treatment	Green lacewing eggs/leaf	1994 Date	Treatment	Green lacewing eggs/leaf
			Beggs, OK					
May 5	Red clover	0^z	June 8	Red clover	0	April 27	Red clover	0
	White clover	0		White clover	0		White clover	0
	Red+white clover	0		Red+white clover	0		Red+white clover	0
	Grass	0		Grass	0		Grass	0
		NS			NS			NS
May 22	Red clover	0	June 16	Red clover	0	May 19	Red clover	0.1
	White clover	0		White clover	0		White clover	0
	Red+white clover	0		Red+white clover	0		Red+white clover	0
	Grass	0		Grass	0		Grass	0
		NS			NS			NS
June 17	Red clover	0.1	June 24	Red clover	0	May 26	Red clover	0
	White clover	0.1		White clover	0		White clover	0.1
	Red+white clover	0		Red+white clover	0		Red+white clover	0
	Grass	0.1		Grass	0		Grass	0.1
		NS			NS			NS
July 8	Red clover	0	July 1	Red clover	0	June 3	Red clover	0
	White clover	0.1		White clover	0		White clover	0
	Red+white clover	0		Red+white clover	0		Red+white clover	0
	Grass	0		Grass	0		Grass	0
		NS			NS			NS
July 24	Red clover	0	July 8	Red clover	0	June 9	Red clover	0
	White clover	0		White clover	0		White clover	15.2
	Red+white clover	0		Red+white clover	0		Red+white clover	0
	Grass	0		Grass	0		Grass	0
		NS			NS			NS

Table 15. (Continued).

Date	1992 Treatment	Green lacewing eggs/leaf	1993 Date	Treatment	Green lacewing eggs/leaf	1994 Date	Treatment	Green lacewing eggs/leaf
Aug 14	Red clover	0.4	July 15	Red clover	0	June 21	Red clover	0
	White clover	0.3		White clover	0		White clover	0.1
	Red+white clover	0.4		Red+white clover	0		Red+white clover	0
	Grass	0.3		Grass	0		Grass	0
		NS			NS			NS
Aug 28	Red clover	0.1	July 22	Red clover	0	July 8	Red clover	0
	White clover	0.1		White clover	0		White clover	0.1
	Red+white clover	0.3		Red+white clover	0		Red+white clover	0
	Grass	0.1		Grass	0		Grass	0
		NS			NS			NS
Sept 15	Red clover	0.1	July 29	Red clover	0	July 28	Red clover	0
	White clover	0.1		White clover	0		White clover	0
	Red+white clover	0.1		Red+white clover	0		Red+white clover	0
	Grass	0.1		Grass	0		Grass	0
		NS			NS			NS
Oct 1	Red clover	0	Aug 10	Red clover	0	Aug 8	Red clover	0
	White clover	0		White clover	0.1		White clover	0
	Red+white clover	0.3		Red+white clover	0		Red+white clover	0
	Grass	0		Grass	0		Grass	0
		NS			NS			NS
Oct 12	Red clover	0	Aug 18	Red clover	0	Aug 19	Red clover	0
	White clover	0		White clover	0.1		White clover	0
	Red+white clover	0.2		Red+white clover	0		Red+white clover	0
	Grass	0.2		Grass	0		Grass	0
		NS			NS			NS

Table 15. (Continued)

1992 Date	Treatment	Green lacewing eggs/leaf	1993 Date	Treatment	Green lacewing eggs/leaf	1994 Date	Treatment	Green lacewing eggs/leaf
			Aug 26	Red clover	0	Sept 6	Red clover	0
				White clover	0		White clover	0
				Red+white clover	0		Red+white clover	0
				Grass	0		Grass	0
					NS			NS
						Sept 20	Red clover	0
							White	0
							Red+white clover	0
							Grass	0
								NS
						<u>Sapulpa, OK</u>		
			May 20	Red+white clover	0	April 26	Red+white clover	0
				Grass	0		Grass	0
					NS			NS
			June 1	Red+white clover	0	May 12	Red+white clover	0
			Grass	0		Grass	0	
					NS			NS
			June 8	Red+white clover	0	May 17	Red+white clover	0
				Grass	0		Grass	0
					NS			NS
			June 24	Red+white clover	0.1	May 24	Red+white clover	0
				Grass	0		Grass	0
					NS			NS
			July 1	Red+white clover	0.1	June 2	Red+white clover	0.1
				Grass	0.1		Grass	0
					NS			NS
			July 8	Red+white clover	0.2	June 7	Red+white clover	0
				Grass	0		Grass	15.2
					NS			NS

Table 15. (Continued)

Date	1992 Treatment	Green lacewing eggs/leaf	1993 Date	Treatment	Green lacewing eggs/leaf	1994 Date	Treatment	Green lacewing eggs/leaf
			July 15	Red+white clover	0.1	June 20	Red+white clover	0.08
				Grass	0.1		Grass	0.04
					NS			NS
			July 18	Red+white clover	0	July 7	Red+white clover	0
				Grass	0		Grass	0
					NS			NS
			Aug 2	Red+white clover	0	July 21	Red+white clover	0
				Grass	0		Grass	0
					NS			NS
			Aug 10	Red+white clover	0.1	Aug 4	Red+white clover	0
				Grass	0		Grass	0.1
					NS			NS
			Aug 18	Red+white clover	0	Aug 15	Red+white clover	0
				Grass	0.1		Grass	0
					NS			NS
						Aug 30	Red+white clover	0
							Grass	0
								NS
						Sept 13	Red+white clover	0
							Grass	0
								NS
						Sept 27	Red+white clover	0
							Grass	0
								NS

[z] NS, * Non-significant (NS).

Table 16. Nitrogen concentration in clover tops, total N and biomass from ground covers in 1992 and 1993.

Location	Treatment	N conc (%)	Total N (kg/ha)	Biomass (kg/ha)
		1992		
Beggs	Red clover	2.34(.08)[z]	56(10)	2475(526)
	Grass	-	-	1824(354)
	Total			4299(880)
	Red+white clover			
	Red	2.71(.07)	8(1)	292(57)
	White	2.77(.12)	19(5)	700(190)
	Grass	-	-	2711(561)
	Total		27(6)	3703(808)
	White clover	2.95(.09)	45(7)	1557(290)
	Grass	-	-	1499(219)
	Total			3056(509)
		1993		
Beggs	Red clover	2.25(0.53)	23(8)	817(289)
	Grass	-	-	1109(254)
	Total			1926(543)
	Red+white clover			
	Red	3.31(.47)	11(3)	370(128)
	White	3.86(.20)	17(3)	471(106)
	Grass	-	-	188(54)
	Total		28(6)	1029(288)
	White clover	3.87(.19)	35(7)	974(209)
	Grass	-	-	757(258)
	Total			1084(249)
Sapulpa	Red clover	2.94(.21)	31(5)	1052(220)
	Grass	-	-	757(258)
	Total			1809(478)

[z] Mean, ( ) standard error.

Table 17. Effect of N application rate on areas with grass ground covers and legume ground covers on Kjeldahl - N and NO_3 - N in soil sampled at two depths in October.

			Kjeldahl - N ($\mu g \cdot g^{-1}$)		NO_3 - N($\mu g \cdot g^{-1}$)	
			Sample depth in (cm)			
Location	Year	Treatment	0-15	15-30	0-15	15-30
Beggs	1992	0	888	706#	1.0	0.5
		56	681*#	436*+#	1.6	0.8*+#
		112	957+	628	3.0	0.8*+#
		168	862	727#	1.3	0.5
		Red clover	897	636	1.3	0.4
		White clover	783	627	1.0	0.3
		Red+white clover	911	584	1.0	0.5
		Significance	C	NS	Q,C	Q
	1993	0	971+	721 +#	1.8 #	0.5 +#
		56	812*#	592*	1.5 #	0.6 +#
		112	686*+#	469 *+#	0.8 +#	0.8 +#
		168	824*#	738 +#	1.3 #	0.5 +#
		Red clover	961	715	1.8	0.8
		White clover	806	601	2.0	2.8
		Red+white clover	964	616	4.9	5.2
		Significance	Q	Q,C	Q	NS
	1994	0	1132+#	715#	3.1#	0.8
		56	1132+#	718#	4.4*#	1.8
		112	1090+#	655#	4.4*#	1.3
		168	1141+#	713#	7.6*+	12.4+#
		Red clover	984	695	1.5	0.6
		White clover	836	645	2.9	1.9
		Red+white clover	1548	893	7.1	4.2
		Significance	NS	NS	L,C	L,Q,C
Sapulpa	1993	0	1115#	700#	1.1#	3.3
		56	1206#	735#	2.5	1.5
		112	1155#	637#	3.9	1.3
		168	1283#	692#	4.8	2.5#
		Red+white clover	1787	1083	3.8	1.1
		Significance	NS	NS	L	L

Table 17. (Continued)

			Kjeldahl - N ($\mu g \cdot g^{-1}$)		NO_3 - N($\mu g \cdot g^{-1}$)	
			Sample depth in (cm)			
Location	Year	Treatment	0-15	15-30	0-15	15-30
Sapulpa	1994	0	1272#	668#	3.1#	1.0#
		56	1368#	790#	6.3#	1.6#
		112	1376#	749#	5.5#	1.1#
		168	1314#	677#	5.2#	1.1#
		Red+white clover	2041	1141	10.7	5.9
		Significance	NS	Q	L,Q,C	NS

$^{\$}$, *, $^{+}$, $^{\#}$ Significantly different from the red clover (*), white clover (+) or then red plus white clover (#) ground cover at 5% level.

^{NS}, ^{L}, ^{Q}, ^{C} Non-significant (NS), or significant linear (L), quadratic (Q) or cubic (C), trends within the four applied N rated at the 5 % level.

Table 18. Pecan fruiting shoots.

		Percent fruiting shoots		
Location	Treatmeant	1992	1993	1994
Beggs	0	34.7[z]	39.3	40.0^{a}
	56	33.0	32.6	53.1^{a}
	112	26.9	31.7	48.2^{ab}
	168	34.0	26.8	35.8^{bc}
	Red clover	25.3	25.3	36.8^{b}
	White clover	7.5	41.8	21.6^{c}
	Red+white clover	34.5	31.3	53.1^{a}
		NS	NS	
Sapulpa	0		34.5	26.4^{c}
	56		38.8	49.8^{a}
	112		35.6	48.8^{ab}
	168		38.5	30.8^{c}
	Red+white clover		53.0	37.4^{bc}
			NS	

[z] Means within columns followed by the same letters are not significantly different at the 5% level by LSD.

Table 19. Pecan leaf N Concentration and Apparent N supplied to the pecan trees by legumes .

Location	Clover	1992	1993	1994
		Leaf N concentration (%)		
Beggs	0	2.07(.12)[z]	2.01(.08)	1.99(.09)
	56	2.09(.11)	2.24(.14)	2.46(.10)
	112	2.08(.10)	113(.05)	2.40(.10)
	168	2.35(.06)	2.23(.06)	2.57(.13)
	Red	2.22(.14)	2.14(.09)	2.21(.15)
	White	2.20(.11)	2.09(.08)	2.11(.08)
	Red+white	2.26(.10)	2.32(.09)	2.31(.11)
	r^2	0.32	0.24	0.52
Sapulpa	0	---	2.45(.08)	2.24(.14)
	56	---	2.51(.10)	2.35(.08)
	112	---	2.54(.12)	2.33(.13)
	168	---	2.53(.12)	2.41(.10)
	Red+white	---	2.53(.12)	2.41(.10)
	r^2	---	0.20	0.35
		Apparent N (Kg/ha)		
Beggs	Red	120(19)[z]	0	66(16)
	White	115(19)	0	47(16)
	Red+white	132(21)	30(35)	83(14)
Sapulpa	Red+white	- - -	93(19)	105(23)

[z] Mean (95% confidence limit).

CHAPTER III

TRICHOGRAMMA PRETIOSUM AS A BIOLOGICAL CONTROL AGENT AGAINST THE PECAN NUT CASEBEARER

Additional index words: *Sitotroga cerealella*, augmentation, dispersal, parasitism, *Acrobasis nuxvorella.*

Abstract: Augmentative releases of *Trichogramma pretiosum* Rilley (Hymenoptera: Trichogrammatidae) were evaluated for parasitizing eggs of pecan nut casebearer *(Acrobasis nuxvorella* Neunzig (Lepidoptera: Pyralidae). *Trichogramma* were released two times at 0, 3000 or 6000 *Sitotroga cerealella* (Oliver) eggs parasitized by *T. pretiosum*/ tree ; 6000 or 12000 total eggs/ tree. Eggs were released on 1 June and 4 June, or 4 June and 7 June. A higher percentage of pecan nut casebearer infestation was obtained on the trees released at 0 or 3000 eggs than at 6000 eggs. Damage by pecan nut casebearer was greatest at the lowest level in the tree canopy followed by the upper and middle levels of the canopy. This may be caused by the preference of *T. pretiosum* for the upper region of the trees or preference of pecan nut casebearer for egg deposition sites at lower canopy heights.

Introduction

The pecan nut casebearer is one of the most important fruit feeding insect pests of pecan (*Carya illinonensis* (Wang.) C. Koch), and greatly reduces yield in many regions of the pecan belt (Harris, 1990). This insect pest has been reported to destroy up to 90% of the pecan crop in some unmanaged native pecans (Boethel et al., 1979). The infestation of this pest varies from year to year depending upon the number and kinds of parasites present, the extent of the crop the previous year and the climatic conditions during March, April, and May (Bilsing, 1926).

The pecan nut casebearer is one of the early season arthropods attacking pecan. This monophagous, multivoltine fruit feeder overwinters as a small larva in a cocoon attached to dormant pecan bud. The larva becomes active coincident with budbreak of the pecan in the spring and grows and develops by first feeding on the buds and then tunneling into the rapidly growing shoot causing it to wilt, turn dark and die. The first generation emerges during late May to early June in Oklahoma, and is much more destructive than later generations because each larva destroys several fruit per cluster; whereas later generations generally only destroy one fruit. Growers normally apply insecticides, usually organophosphate, to control the first generation casebearer and prevent economic damage. But this reduces beneficial arthropods in the orchard and may result in aphid outbreaks. Yellow aphids, primarily the blackmargined aphid, tend to build during May and June after insecticides are applied for the pecan nut casebearer (Harris et al., 1988). Thus, it is imperative to look for an alternative control method for pecan nut casebearer that eliminates the need for pesticide application in pecan orchards. An alternative control for this lepidopterous pest may be the augmentation of existing natural enemies such as *Trichogramma*. *Trichogramma* are parasitiods of lepidopterous eggs. They are the most widely used and commercially available entomophagous insects in many countries (King et al., 1984). *Trichogramma* have been used as a biological control agent for field and vegetable crops, vine yards, and fruit orchards (Li, 1994).

Control with *Trichogramma* is mostly attempted through inundative releases of *Trichogramma*, wherein the crop is flooded with insectary reared parasitiods to raise the field parasitism rate sufficiently to prevent economic injury level to the crop (Olkowski and Zhang, 1990). In addition to direct control, *Trichogramma* releases may also have a preventative effect. They provide benefits not only to the current period of utilization, but also to successive years of production (Li, 1994). In some countries it has been possible to reduce and even abolish the use of agrichemicals by the preventative mass releases of *Trichogramma* to regulate lepidopterous pest species of economic importance in cotton, tomato, soybean and cassava crops (Garcia-Roa, 1990). The *Trichogramma* release program offers economical and ecological advantages. Crops submitted to parasitoid releases present a high level of protection due to the effectiveness of the parasitoid and the combined action of other beneficial agents as well as the integration of cultural and microbiological control measures.

Trichogramma release for control of lepidopterous pests has had mixed results (Hassan, 1981). Kot (1968) identified the significance of factors such as the species and strain of *Trichogramma* released, host density, numbers released and behavior of the parasitiod in modifying the results of inundative releases. Their effectiveness in the field can largely depend on their searching behavior (habitat location, host location), host preference (recognition, acceptance, suitability) and tolerance to environmental conditions (Hassan, 1994).

Trichogramma pretiosum is one of the preferred *Trichogramma* spp. in field and vegetable crops and vineyards. It is commonly used in many countries against a number of insect pests. This species is endemic to southern California and is considered a potential indigenous and augmentative biological control agent (Bai et al., 1992). *Trichogramma pretiosum* was tested against hickory shuckworm of pecan in the laboratory and in the pecan orchard during 1989. It was found effective in the laboratory; however, control was not achieved under field conditions (Calcote and Tedders, 1990). During 1930 to 1935 experiments were conducted with *Trichogramma minutum* Riley against pecan nut casebearer, but successful results were not obtained (Spencer et al., 1949). The purpose of this study was to evaluate the efficacy of *a T. pretiosum* biotype collected from hickory shuckworm eggs in parasitizing eggs of pecan nut casebearer.

Materials and Methods

This experiment was conducted at the Pecan Research Station located near Sparks, OK in June 1994. Trees were 44-year-old ‘Western’ cultivar of uniform size (about 14 m tall). No pesticides had been applied in the orchard during 1994. *Trichogramma pretiosum* were reared in the laboratory on eggs of *Sitotroga cerealella* (Oliver) following the methods of Morrison (1985).

Treatments: Release rates were 0, 3000 and 6000 eggs of *S. cerealella* parasitized by *T. pretiosum*/tree/release; 6000 and 12000 total eggs/tree/release. Eggs were released on 1 June and 4 June, or 4 June and 7 June. Each treatment included five single tree replications in a completely randomized design. Two dixie cups (142 gm) were used to place half of each rate in the cup to be distributed at the release site. The dixie cups were folded shut at the top and stapled. On the side of each cup six holes were cut to let the parasites escape. The two dixie cups containing eggs were hung on limb tips, 2-3 m above the ground on the opposite sides of the tree. Releases were at 4 to 5 p.m. Eggs were inspected to determine if they were attacked by other pests, but no evidence was found.

The minimum and maximum temperature and rainfall records during and after the release period are shown in table 1.

Table 1. Minimum and maximum temperature and rainfall records

Date	Maximum Temp.(^{0}C)	Minimun Temp.(^{0}C)	Rainfall (mm)
June 1	31	21	0
June 2	32	20	0
June 3	32	21	18.8
June 4	32	21	0
June 5	33	19	9.9
June 6	29	19	10.9
June 7	34	24	0
June 8	34	25	0
June 9	27	22	0
June 10	28	21	0.5
June 11	28	21	0
June 12	32	20	0.3
June 13	33	24	0
June 14	32	25	0
June 15	29	24	0
June 16	32	24	0
June 17	32	22	0
June 18	34	22	0
June 19	33	21	0
June 20	35	22	0

Eggs of pecan nut casebearer and fruit entries were counted periodically to detemine the proper release time. Counts at the time of release are in Table 2. Eggs are white when first laid. After two or three days a pinkish color develops and eggs change to a red color, indicating larval emergence is imminent.

Table 2. Number of eggs or enteries of pecan nut casebearer by egg color and number of entries.

Release date	Casebearer eggs /100 fruit clusters			Casebearer entry/ 100 fruit clusters
	white	pink	red	
1 June	2	0	0	1
4 June	4	1	0	1
7 June	5	0	0	1

Treatment evaluation. On 27 June, fruit clusters were examined for evidence of pecan nut casebearer infestation. Fruit clusters were examined at random within three regions of the tree canopy. Number of infested fruits were recorded from 50 fruit clusters at three different regions in the individual tree canopy to examine the effect of parasitism by *T. pretiosum*. The average height of the selected trees was 14 m ±1 m. Sample heights in the upper, middle and lower canopy regions from the ground were 9-15 m, 4.5-9 m and 0- 4.5 m, respectively. A nut was considered to be infested if any of the following were found: immature or mature larva(e), larval feeding damage such as tunnels at the base (even if no larva was found), and frass or web. Parasitized eggs turned brown or black when the parasite pupate inside the egg.

Data were analyzed by analysis of variance (ANOVA) and means separated by LSD.

Results and Discussion

Nut cluster infestation. The percentage of infested fruit clusters by pecan nut casebearer after the release of *T. pretiosum* ranged from 6.5 to 10.2% (Table 2). The infestation percentage of pecan nut casebearer on the trees treated with 0 and 3000 parasitized eggs/ release was significantly higher than for trees treated with 6000 parasitized eggs/ release. The best suppression of pecan nut casebearer damage was 6000 parasitized eggs released on either 1 and 4 June or 4 and 7 June which represented 20 to 33% control. Release of 12000 parasitized eggs (two 6000 egg releases) is the equivalent of 535,000 eggs/ha. There was no significant interaction between the release rates and heights within trees.

Table 3. Percentage of infested fruit clusters at different *Trichogramma* release rates and dates.

Trichogramma per release	Release dates	Infested fruit clusters (%)
3000	1 June, 4 June	10.2 [a] [z]
	4 June, 7 June	10.0 [a]
6000	1 June, 4 June	7.8 [b]
	4 June, 7 June	6.5 [b]
0	- - -	9.7 [a]

[z] Means within dates and columns followed by the same letter are not significantly different at the 5% level by LSD.

Previous augmentation studies showed that large numbers of *Trichogramma* are required to provide a moderate level of egg mortality. Stinner et at. (1974) achieved 33 to

81% parasitism of *Heliothis* eggs in cotton with releases of 469,300 to 957,125 *T. pretiosum* per ha. Oatman and Planter (1978) obtained 53.1 to 85.4% parasitism of *Heliothis zea* (Boddie) in tomatoes with releases of 500,000 to 795,000 *T. pretiosum* per ha. Newton (1990) reported also that high volumes *Trichogramma cryptophlebia* (Nagaraja) released in citrus orchards led to a variable rates of success in reducing damage by false codling moth. These results support our findings.

Fye and Larsen (1969) indicated that the searching capability of female *Trichogramma* can be seriously hampered by complexity of the foliage environment. The density and complexity of the trees and their foliage can effectively reduce the speed and distance of searching by the female parasitoids. Hence, to achieve high parasitism more *Trichogramma* should be released.

Distribution of parasitism. The damage caused by pecan nut casebearer at different heights within trees ranged from 7.9% to 10.8% (Table 3). The infestation percentage of pecan nut casebearer at the lower level was significantly higher than in the middle and upper canopy. The lower level of infestation in the middle and upper part of the tree might be attributed to a higher rate of egg parasitism by *T. pretiosum,* or there were less pecan nut casebearer eggs deposited.

Table 4. Damage caused by pecan nut casebearer in different regions of the tree.

Height within trees (m)	Infested fruit clusters (%)
0 - 4.50	10.8 $^{a\,z}$
4.5 - 9	7.9 ^{b}
9 - 15	7.9 ^{b}

^{z} Means within columns followed by the same letter are not significantly different at the 5% level by LSD.

The dispersal rate and pattern of *Trichogramma* is species specific and dependant on the type of crop in which they are released (Hendricks, 1967; Stinner et al., 1974). *Trichogramma pretiosum* has been reported to prefer the upper region of the crop (Gonzaleth et al.,1970). *Trichogramma minutum* also prefer the upper canopy (Kot, 1964; Smith, 1988), while several other species prefer the lower canopy (Burbutis et al., 1977) and still others show no preference. In general, *Trichogramma* disperse short distances from the point of release, usually less than 20 m (Smith, 1988). *T. pretiosum* dispersed in significant number in all directions away from the release point in spite of wind speeds averaging 3 - 4 m/s. This may support our findings in that the greater preference of *T. pretiosum* for the upper canopy lowered the infestation of pecan nut casebearer.

Dispersal can be inhibited by vegetation such as dense tree crowns in orchards and by high host egg densities (Kot, 1964). In addition, climatic factors such as temperature, humidity and dew, wind and possible duration of photophase significantly affect the movement and efficacy of the parasites. It was reported that when the complexity of the plant increased or the temperature became higher the searching efficiency of *T. minutum* markedly declined, but showed higher rate of parasitism in the upper portion of the plant (Fye and Larsen, 1969). The rate of searching by several female *Trichogramma* spp. increased as temperature increased from 20 to 35 ^{0}C, but declined above 40 ^{0}C (Biever, 1972). At 32 ± 1^{0}C and 60 to 80% relative humidity the highest rate of emergence of *T. pretiosum* from the eggs occurred at time of eclosion, but there was poor emergence above 37°C. Some authors have reported on the effect of wind direction on parasitoid distribution (Hendricks, 1967; Yu et al., 1984). Upperward movement within trees was apparently not influenced by wind direction or speed, possibly because the volume of foliage in individual trees interrupted air movement.

For the parasite to be successful in the orchard, the abundance and distribution of the host is also important. The presence and distribution of host eggs differ from crop to crop and from year to year. Adult parasites that do not find food, host eggs and shelter have a much shorter lifespan and less chance of locating hosts. If an adult has to fly from one tree to another in search of a host, it would need high flying and walking abilities to find a host before exhaustion. These attributes are important both for the survival of the parasite and for its usefulness as a biological control agent (Hassan, 1994).

Conclusions

Results indicate that augmentative releases of *T. pretiosum* to reduce damage caused by pecan nut casebearer appears promising. The highest release rate of *T. pretiosum* has reduced damage from pecan nut casebearer. Egg parasitism in the upper and middle regions of the tree canopies were greater than the lower region. Although conclusive statements may not be made with a single year's results, our findings indicate the potential of *T. pretiosum* to parasitize eggs of pecan nut casebearer. However, even at release rates of 535,000 parasitized eggs per ha damage reduction by pecan nut casebearer was not sufficient to justify trial use by producers.

Factors which may influence the success of this species after augmentation, include poor retention of the parasitoid in the target area and inefficient host searching. The density and complexity of the trees and their foliage may reduce the movement and efficacy of the parasites. Unfavorable weather can also result in lack of success by causing high mortality of the parasites. However, if the most effective release techniques are employed under favorable environmental conditions, the use of *Trichogramma* may become an alternative for reducing damage caused by pecan nut casebearer.

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CHAPTER 1V

SUMMARY

Current management practices in most pecan orchards entail heavy applications of fertilizers and pesticides. Fertilization with N is conducted annually in the spring and is applied to the soil at 100 kg/ha or more. Permanent sods such as bermudagrass have been grown as understory cover in pecan orchards, but compete for moisture and nutrients, thus decreasing growth and yield of pecan trees. Growers also follow intense pest control programs using eight to ten insecticide applications in the southeastern U.S. and five to seven applications in the south central U.S. These intensive management practices have resulted in several problems such as increased input costs, increased resistance development of pests and outbreak of secondary pests especially aphids, greater environmental pollution especially ground water and surface water contamination caused by NO_3 pollution, and growers have also been restricted from grazing their cattle in their pecan orchards due to pesticide applications. These environmental, economical and social problems have led to the development of low-input management systems. One of the alternative approaches to reduced use of chemical inputs would be an integrated management system using legume cover crops, releasing exotic natural enemies or enhancing natural enemies. Legumes or mixtures of legumes grown as ground covers may offer certain advantages compared to perennial grass sod, such as supplying N and increasing beneficial arthropods. Certain legumes harbor large aphid densities that attract aphid predators, and these aphid predators may migrate from the legume ground cover to the pecan canopy, controlling pecan aphids and other pests.

Evaluation of perennial legume ground covers as pure stands or a mixture of 'Kenland' red clover and 'Louisiana S-1' white clover were conducted to determine the N production capacity and aphid infestation rate which attracts beneficial insects to feed up on aphids. Aphid densities counted on the clovers were generally low at both sites because of heavy rainfall. The most abundant beneficial arthropods sampled in the legumes were spiders, lady beetles, predacious stinkbugs and nabids, respectively. The ground cover type, however, did not affect the densities of beneficial species in the pecan canopies. This may be attributed to low pecan aphid densities in the trees and differences in habitat preference.

The most abundant beneficial arthropods in pecan canopies were spiders, lady beetles, and green lacewings, respectively. Spider densities were usually consistent from spring through fall. They are general predators and their feeding activity may not be associated with pecan aphid densities. Green lacewings did not show consistent seasonal patterns during the study period. Both green lacewing adult and larval densities in the trees were not affected by the ground cover. Lady beetle species distribution in pecan canopies was different than in the legumes. In legumes, the predominant lady beetle species during the study period was *Coleomegilla maculata lengi*, but in pecans it was most abundant in 1993 and lowest in 1994. *Olla v-nigrum* and *Harmonia axyridis* were major lady beetles found only in pecan. These two species have more of an arboreal nature, preferring the habitat of the canopy to that of the legumes.

In 1992 and 1993, pecan aphid densities were low and erratic in all the treatments, and did not reach established thresholds for control. In 1994, pecan aphid densities at Beggs were variable among treatments during the sampling dates. At Sapulpa, significantly higher pecan aphid densities were found in the canopies in grass plots than in the legumes. Unhatched lacewing eggs were also very low and showed no significant difference between the grass and the legume plots through out the study period.

A mixture of red clover plus white clover appeared to yield sufficient N to meet pecan requirements at both sites. Pecan roots are not active during fall to readily absorb the accumulated NO_3 from the decomposition of legumes in the soil. Thus, the chance of NO_3 leaching below the root zone might be higher by using legumes than well timed N application. However, this may depend on the intensity and quantity of rainfall, soil type and the amount of N accumulated in the soil.

In general the impact of using certain legume ground covers to increase beneficial arthropods for biological control appears to be small. The primary benefit of these legumes is considered to be the N contribution to pecans.

The second experiment evaluated the efficacy of augmentative releases of *T. pretiosum* in reducing the damage caused by pecan nut casebearer. A higher release rate of *T. pretiosum* reduced damage from pecan nut casebearer. Egg parasitism in the upper and middle regions of the pecan canopies were greater than in the lower regions. No significant interaction was shown between release rates and tree heights. Although damage was reduced at a release rate of 535,000 parasitized eggs per ha, the reduction was not sufficient to justify trial use.

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