

## Chapter 9

## MANAGEMENT OF PREHARVEST INSECTS

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Worldwide, some 10,000 species of insects are pests of man, domestic animals, food and fiber. A substantial industry has developed to produce synthetic insecticides and other pesticides to combat this myriad of pests (Bottrell, 1979). Insecticides have been of tremendous benefit to man but have not been used without deleterious side effects (Luck et al., 1977; Bottrell, 1979; Metcalf, 1980). Boraiko (1980) cites specific aspects of environmental and human health hazards attributed to synthetic pesticide usage. A comprehensive review of the history of insecticide usage and subsequent problems is contained in Metcalf (1980).

Before the late 1930's agriculturists did not have access to many pesticides; thus, they were forced to rely on culturally inherited farming practices for pest control. Such methods (e.g., crop rotation) often unknowingly took advantage of basic ecological principles to reduce pest attack. Today, many agriculturists are directing research efforts toward gaining an understanding of how an agroecosystem functions (i.e., how its components interrelate) so that pest control strategies which are less ecologically disruptive than blanket usage of insecticides can be developed. Efforts to rekindle studies on agroecosystem form and function have necessitated philosophical, as well as scientific, alterations in the way agricultural scientists approach the problems of pest control.

## THE IPM PHILOSOPHY

The latest in a series of philosophies on how to combat pest organisms is called integrated pest management (IPM). Numerous authors (e.g., Smith and van den Bosch, 1967; Huffaker, 1972; Bottrell, 1979; Barfield and Stimac, 1980) define IPM more or less identically as the use of various tactics (chemical, cultural, biological, physical) in an integrated fashion so as to yield predictable economical, ecological and sociological consequences. We shall return to this definition of IPM later to provide specific examples of where the development of IPM in worldwide peanut, *Arachis hypogaea* L., production systems is relative to this definition.

Integrated pest management is synonymous with pest management, and both terms evolved from integrated control which was originally used to describe the use of biological and chemical controls synchronously (Stern et al., 1959). Theoretically, IPM represents a combination of actions (tactics) which can be blended into an overall, balanced attack (a strategy). Realization of the optimum combination of tactics into a strategy for a given crop, pest, or crop-pest complex is not a trivial task. Actual examples show that current IPM programs are in various stages of development (Bottrell, 1979; Barfield and Stimac, 1980).

Barfield and Stimac (1980) critically reviewed IPM from an entomological perspective by (1) focusing on characteristics of agriculture conducive to creating insect pests, (2) identifying characteristics of insects which enable them to become pests, (3) retracing the historical route of insect control up to IPM, (4) elucidating discrepancies between theory and practice of IPM, and (5) identifying relevant problems which must be overcome in dealing with insects as pests and IPM as a philosophical commitment to combatting pests.

Our purpose here is to present the utility of IPM for the peanut agroecosystem. This presentation can best be accomplished in 4 steps. First, we will identify some basic concepts which characterize IPM, then use these concepts as milestones to judge where peanut agriculturists are in relation to the realization of IPM programs. Second, we will identify various approaches to combatting pests and show which, if any, of these approaches is currently utilized in peanuts and how such approaches may change as a function of variables such as crop mix and geographical location. Third, we will provide a conceptual model of the peanut system to serve as a reference for identifying existing and missing information. Fourth, we will place some priority structure on the missing information and justify that structure as relevant to the development of IPM schemes in peanuts.

At least 5 principles of IPM have been identified (Bottrell, 1979). The first and foremost principle is that potentially harmful species will continue to exist at tolerable levels of abundance (Smith and van den Bosch, 1967). Thus, under virtually all situations, pest eradication is not consistent with an IPM program. Second, the ecosystem is the management unit (Smith and van den Bosch, 1967). We shall see later how the focus on management at the individual peanut field level has resulted in uncertainty, particularly in management of mobile, polyphagous insect pests. Third, IPM encourages maximum utility from naturally occurring mortality agents (parasites, predators, pathogens) (Stern et al., 1959). Fourth, any applied control procedure may produce unexpected and undesirable effects (Smith and van den Bosch, 1967). Last, an interdisciplinary systems approach is essential to the development of IPM. Examples will be provided later of ongoing efforts which are aimed at using models as tools to understand the peanut agroecosystem prior to managing it. In short, these are efforts to avoid violation of principles 4 and 5 of Bottrell (1979). These 5 principles will be used throughout this discourse in reference to why specific problems (and potential solutions) seem to exist in development of IPM for specific insects or pest complexes within the peanut agroecosystem. Having reviewed these principles, various approaches to combatting pests are summarized. Afterward, conclusions will be drawn to determine the status of development of IPM programs for insects or pest complexes.

Barfield and Stimac (1980) reviewed 4 distinct approaches to combatting insects. A brief review of these approaches is necessary for identifying how specific peanut insect pests are being dealt with today. The first approach is no action and involves a lack of action in 2 distinctly different situations: (1) in the absence of relevant data and (2) as a decision following analysis of relevant data. Secondly, prevention can be utilized. This approach involves at least 6 categories of tactics: (1) use of resistant plant varieties; (2) manipulation of crop planting date, tillage and row spacing; (3) conservation or introduction of pest natural enemies; (4) crop rotation schemes; (5) use of attractants or repellants; and

(6) preplant application of insecticides. The third approach is suppression, and this approach involves a broad spectrum of actions which may be taken after an insect pest has reached (or is expected to reach) densities considered to be economically important. There are 3 generic categories of suppressive agents: chemicals, parasites and predators, and microbials. The final approach to combatting insects (or other pests) is directed management and involves the use of compatible tactics such that specific consequences, within specified ranges, are understood prior to action. Thus, directed management involves insects as well as other pests and complexes of beneficial organisms. The level of knowledge about the structure and function of a particular agroecosystem needed to achieve directed management appears far superior to the level of knowledge needed for prevention and/or suppression.

The basic concepts of IPM have been outlined and 5 principles identified which must be considered in the development of IPM programs. Further, a summary of 4 distinctive approaches to combatting insect (and other) pests have been provided. Integrated pest management provides the theoretical foundation necessary to deal with pests over sustained intervals of time; however, it is recognized that the multitude of ongoing programs designed to deal with insect pests are in various stages of development. Focus must now be directed toward peanuts as a particular crop plant with numerous pests, many of which are cosmopolitan in distribution. Further, concentration will be on the insect components of that pest complex. Questions relating to which species attack peanuts, where (geographically and in relation to habitat) they attack, when (seasonally and in relation to plant phenology) they attack, and what can be done to lessen the impact of these attacks on a worldwide basis will be addressed. This systematic approach focuses on some basic features of the peanut agroecosystem which contributes to insect pest problems and identifies necessary information for making progress toward the development of IPM programs for peanuts. Lastly, we hope to suggest how features of these IPM programs might vary geographically. Perhaps the initial step should be a conceptual model of the basic features of the peanut agroecosystem.

## CONCEPTUALIZATION OF THE PEANUT SYSTEM

Stimac and Barfield (1979) presented a conceptual model of spatial and pest species hierarchies for soybean which may be applicable to peanuts. Using this conceptual model, we can visualize an analogous spatial hierarchy of how insect pests may arrive in a peanut field. We can then separate pests into pest hierarchies and focus on how insects interact with both the peanut plant and other pests. Within a field, various pests (some of which are insects) attack different peanut plant parts; further, these parts may be attacked at various times in a particular growing season. To design management strategies for economically and environmentally sound production of peanuts, we must focus on 5 aspects of a given peanut field. First, identify what parts of the peanut plant are available for attack, and what magnitude and timing of attack is needed to reduce yields significantly. Second, focus on the characteristics (behavior and biology) of select pests capable of inflicting such damage. Third, identify exactly how these select pests inflict damage, and what can be done to alleviate such

damage. Fourth, evaluate the compatibility of tactics to avoid creating some pest problems while alleviating others. Lastly, identify what knowledge is missing relative to our ability to design viable IPM strategies for peanuts.

Peanuts are attacked by plant intracellular feeders, foliage consumers, insect-transmitted diseases, and insects feeding on roots, pegs and pods. Each type of pest has 1 or more naturally occurring enemies (predators, parasites and/or pathogens) which theoretically can be manipulated for pest suppression. Besides these natural biological controls (or imported ones), at least 3 other categories of management tactics appear to be in use today against peanut insect pests: resistant plant varieties, various combinations of cultural practices, and various insecticides applied at some economic threshold pest density or in a preventative manner. How these 4 general categories of tactics are used depend upon geographical location, particular pests in question, age of crop, and philosophy of the managers.

Now that the general IPM philosophy and field level components (plants, pests, natural enemies, environments and management tactics) in a peanut system have been reviewed, a focus on state of the art for IPM in peanuts worldwide becomes pertinent. Insect pests are divided into 2 major categories based on habitats: foliage inhabitants (consumers, intracellular and insect transmitted diseases) and subterranean inhabitants. The format will include the pest status of each type pest, current management practices used against each, and current information (biological and ecological) about each. This approach will accomplish 2 goals critically important in preparing this book chapter. First, focus is directed on both the similarities and differences in the way IPM strategies for specific pests have materialized around the world. A given insect may be a key pest in 1 location and only an occasional pest elsewhere. Knowledge of why this is true is of central importance in constructing robust management strategies (Barfield and Stimac, 1979). Second, a critically sparse amount of information exists on the ecology of many peanut insect pests, and this paucity has hampered development of better IPM programs for peanuts in many instances. The in-depth discussions of a few well-known species in each of the aforementioned pest groups will provide the evidence that vital ecological and biological information is missing. A final summary will address methodology for overcoming these deficiencies. Toward this end, crop-pest relationships and pest status categories are explored next to provide a conceptual framework for discussing specific arthropod pests and pest groups.

## CROP-PEST RELATIONSHIPS

The central issue in the design of crop protection strategies focuses on 3 critical questions: (1) when is the plant really susceptible to irrecoverable damage?; (2) how much damage does it take to cause true economic loss?; and (3) which organisms (singly or in combination) are capable of inflicting true economic damage? Sufficient data exist for us to explore these questions within the peanut agroecosystem. To accomplish this exploration, we must move beyond general definitions of the economics of crop-pest interactions to the details of specific experimentation on peanuts.

By definition, an insect is considered a pest when its feeding either directly or indirectly causes economic loss. Such loss results from the ecological syn-

chrony in time and space between specific insect pest populations and susceptible crop plants. A more or less static pest density may inflict varying degrees of damage, dependent upon the age of the plant when attacked. The ability of a plant to withstand injury is related to physiological mechanisms (and resulting morphologies) dictated by plant age. The relationship between plant age and the degree of reaction to injury is of paramount importance in understanding how peanuts and various pests interrelate. This age-injury relationship is termed *temporal tolerance*.

Temporal tolerance can be expressed by both gradual and abrupt changes in plant phenology. A gradual change (e.g., seed maturity) may cause exposure to damage over relatively long intervals of time. A more abrupt change (e.g., pod appearance) occurs over a much shorter time interval, and insects feeding exclusively on pods cannot inflict damage until after this change occurs. In gradually maturing plant parts, the amount of real damage inflicted by insects is related to the plant's capability of producing those parts. At certain plant ages, what appears to be significant damage can, in actuality, be replaced by the plant with nonsignificant or no yield reduction. At other plant ages, the

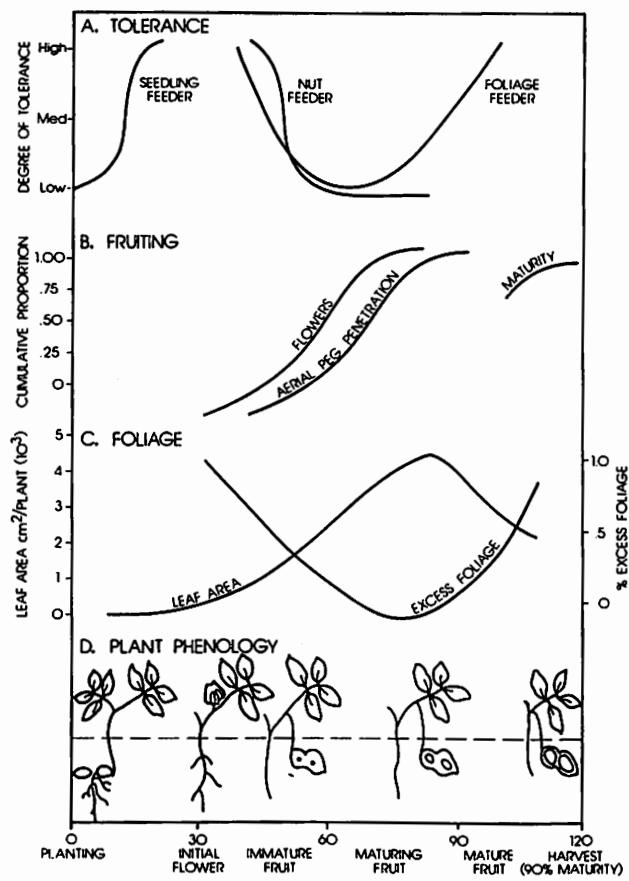


Fig. 1. Spanish peanut phenology and pest tolerance.

same damage will result in significant yield reduction. Several relationships are depicted in Figure 1A (tolerance). Generally, the plant becomes less tolerant in time to nut feeders. The inverse relationship (more tolerant in time) is also possible (Figure 1A-seedling feeder). An insect that injures young peanuts but cannot injure older, mature plants is an example of a case where the plant is more tolerant in time. A third generalized relationship involves pests which consume foliage (Figure 1A-foliage feeder). The plant is less tolerant to defoliation toward mid-season; it is more tolerant of such damage in early and late season.

Another aspect of tolerance is related to the plant part subjected to pest damage. Pest injury to harvestable plant parts (pods) usually causes a more severe reduction in yield than injury by a defoliator. Pests feeding on pods have a much more direct relationship between damage and yield loss than do defoliators whose damage is filtered through plant photosynthetic and partitioning mechanisms. Simply, the plant has a greater propensity for recovering from foliage loss than from pod loss. The ability of the peanut plant to withstand damage, while not reducing yield significantly, is thus related to the distance between the inflicted damage sites and the harvestable sites. That distance is 0 when pods are damaged; thus, maximum loss occurs (eaten pods cannot be harvested). This injury site-recovery ability relationship is termed *spatial tolerance*. In instances where secondary microbial infection or disease transmission occur as a result of insect damage, the concept of spatial tolerance is modified because effects of such infections are realized through internal plant physiological processes. However, when damage is a direct result of insect feeding, spatial tolerance is a valid general concept.

Various published investigations substantiate the general concepts of temporal and spatial tolerance. Williams et al. (1976) reported differential effects of foliage and pod removal in both yield and growth rate of specific plant parts. Plant response to 50 and 75% leaf removal was dependent upon the age at which defoliation was imposed. Pod removal did not change total growth rate; however, yield was obviously reduced. In an effort to quantify the effects of defoliation and disease (*Cercospora* sp.) on peanut plant canopy photosynthesis, Boote et al. (1980) demonstrated that the zone (upper, middle, lower) of canopy damage was important in understanding the peanut plant's reaction to specific injury. The ability of peanuts to recover from various amounts of foliar damage, depending on the plant age at which damage was imposed, was quantified by Jones et al. (1982). These investigators provided quantification of the temporal tolerance of peanuts. Further, they measured how the plant responded in growth, photosynthesis, respiration and yield to impositions of damage. Their approach differs markedly from other studies which imposed damage and measured only yield. This deviation is critically important for gaining greater insight into crop protection schemes aimed at providing protection only when needed. Quantitative descriptions of concepts like temporal and spatial tolerance are not merely useful; they are critical to understanding a crop-pest system, and such understanding is essential to economically efficient protection of the crop.

At least 2 commercial types of peanuts are identifiable: runner (prostrate) and spanish (bunch). Evidence on phenological events and specifics of growth, photosynthesis and response to damage is available for both types. Obviously,

specific growth patterns and responses to insect (and other) damage are conditioned by local agronomic practices and physical environment. Yet, published information (e.g., McCloud, 1974; Williams et al., 1975; Williams, 1979) reveals few real differences in the phenological sequence between runner and spanish type peanuts. We will attempt to describe generally the phenology of peanuts so as to understand when plant parts are available for insect attack and when damage to these respective plant parts is meaningful, rather than concentrate on minor differences between spanish and runner peanuts.

Certain physiological and morphological events inherent to development of peanuts provide the template for the relationship between insect damage, plant growth, and yield. Progressive changes in some of these events are depicted qualitatively (Figure 1D; adapted from Schenk, 1961) for a spanish type peanut. Seedlings emerge in ca. 7 days, and the onset of flowering begins at ca. 30 days. Pod formation and seed genesis occur at ca. 45 days, with a maximum % mature seed at ca. 120 days (maturity). A quantitative description of leaf area growth (Figure 1C; Smith & Barfield, unpublished) and fruit production (Figure 1B; adapted from Gilman, 1975) is shown. Data on flowering and maturity (Gilman, 1975) and peg penetration (Smith, 1950) help define and explain pest relationships with regard to plant phenology.

Flower production begins ca. 30 days from planting with aerial peg penetration of the soil occurring ca. 10 days after flowering (Smith, 1950). Since the peanut is an indeterminate fruiting plant (i.e., fruits continuously until climate terminates growth), it is imperative to determine the plant age where most harvestable fruits arise. Data (Figure 1B) show that pods mature to harvestable yield after 120 days. This time appears to be an asymptote for maturity. Mature, 120-day old seed would have had to penetrate the soil by day 70 since ca. 50 days are required for seed maturity after penetration into the soil (Schenk, 1961). Correspondingly, precursor flowers were produced prior to day 60. In summary, mature pods harvested at 120 days arose from flowers produced during days 30-60 and pegs which penetrated the soil from days 40-70 (Figure 1B). The plant is most sensitive to pests feeding on pod precursors during the 40-70 day period, since any pod formation occurring after this date does not contribute to the harvested product (assuming the crop is harvested at 120 days).

Leaves provide energy to the plant through photosynthate production. Leaf area peaks at 80-85 days (Smith and Barfield, unpublished) which corresponds to the time when the plant is most sensitive to defoliation (Figure 1C, Boote et al., 1980; Jones et al., 1982; Smith and Barfield, unpublished). Sensitivity of the plant to defoliation has been derived experimentally from defoliation experiments and expressed as proportion excess foliage. These 2 curves show that the plant has produced peak foliage area at the same time that foliage is needed most. Results in Boote et al. (1980) and Jones et al. (1982) show similar results under different environmental settings. Similar experimental designs have yielded analogous information from other crop systems (e.g., Ingram et al., 1981). The important physiological event of seed maturity (filling with oil) is thus coincident with peak foliage production (Figure 1C,D). Oil synthesis begins in spanish peanuts when the seed is 14 days old and continues until the seed is ca. 50 days old (Schenk, 1961). If the midpoint for peg penetration is considered as 55 days from planting, and the midpoint for the maxi-

mum rate of oil synthesis requires an additional 32 days, then the calculated peak of oil synthesis is ca. 87 days. The production of lipids represents an energy sink whereby more energy is necessary from photosynthesis to produce lipids for the seed (ca. 60% oil) than to produce carbohydrates for general plant and pod growth. This high energy requirement for oil synthesis is reflected in the foliage growth curve and experimentally verified by the excess foliage curve (Figure 1C). This compilation of certain gross physiological and morphological processes, coupled with the period of plant growth and development when these processes occur, aids in identifying critical damage windows for deployment of crop protection tactics.

Requisites for arthropod pest status include synchrony with a susceptible plant growth stage, and population density or feeding voracity sufficient to inflict injury for which the plant cannot compensate; thus, yield is reduced. Defining an economically damaging population density is difficult, but is approached by weighing the monetary crop loss due to insect damage against the cost of control. For an economic benefit to be obtained, the predicted monetary loss must exceed the cost of control (Smith and Holloway, 1979; Berberet et al., 1979a). The economic threshold (Stern et al., 1959) is the pest density at which control measures should be applied to maintain an economic advantage. This is an extremely variable value, subject to changes in commodity value, control cost, pest density, local climatic condition, etc. As we have pointed out here, the economic threshold is a function of plant age. The concept, however, is important in pest management as it requires knowledge of numerous facets regarding the particular pest and its relation to the plant.

Economic thresholds usually are established on a regional basis and frequently revised. Frequent revision of economic thresholds appears to be the state of the art as insufficient data are available to either evaluate local thresholds or develop tools (e.g., systems models) to predict dynamic thresholds as functions of a vector of input variables such as local weather, market value, pest density and crop age. One of the major problems facing agriculturists, including those working on peanuts, is precisely how to arrive at dynamic, realistic damage thresholds. Barfield and Stimac (1980) argue that systems modeling appears currently to be the most viable tool for accomplishing this goal.

## PEST STATUS

Pest status is an important concept relative to understanding and developing pest management strategies. A phytophagous arthropod may be classified as a key, occasional, secondary or non-pest species. Properly conceived management strategies are focused on the key pest (Smith and van den Bosch, 1967; Bottrell, 1979). A key pest usually causes consistent economic damage annually; whereas, an occasional pest causes economic damage at irregular and unpredictable intervals, usually not annually. A secondary pest is a species which originally was an occasional or non-pest species whose status has changed, for some time interval, to that of a key pest. This change in status usually results from major man-induced changes in the agroecosystem such as crop varietal changes, pesticide use, establishment of pest alternate host plants, changes in planting date, etc. Non-pest species, historically, have never reached economically damaging levels; however, many of these are potentially economically injurious (Smith and Jackson, 1975).

Management strategies usually are directed at the key pest(s), attempting to maintain the population density below the economic threshold. It is imperative that management tactics directed at the key pest do not disturb the extant natural balance that maintains occasional and non-pest species below economically damaging levels. Thus, tactics aimed at key pests also must consider other phytophagous arthropods in the agroecosystem (Barfield and Stimac, 1980).

Pest status can be tempered by regional environmental conditions. The lesser cornstalk borer, *Elasmopalpus lignosellus* (Zeller) and southern corn rootworm, *Diabrotica undecimpunctata howardi* Barber, are key pests of peanuts in the United States, whose pest status in the 3 major peanut production regions (southwest, southeast and Virginia-Carolina) is regulated by climatic and edaphic conditions.

The severity of damage by the *E. lignosellus* is related directly to (although not restricted to) a combination of deep sandy soils and low rainfall (Luginbill and Ainslie, 1917; King et al., 1961; Walton et al., 1964; Smith, 1981). In the southwest where the growing seasons are characteristically hot and dry, soils are deep sands and irrigation is limited; *E. lignosellus* is the key pest and annually causes severe economic damage (Berberet et al., 1979a; Smith and Holloway, 1979). In contrast, in the southeast and Virginia-Carolina where the rainfall is normally more abundant and soils are heavier, the severity of *E. lignosellus* is related to a combination of light soils and length of droughts (Luginbill and Ainslie, 1917; Leuck, 1966; French, 1971). In normal rainfall years, *E. lignosellus* is an occasional pest since rainfall tends to suppress population outbreaks; however, with prolonged droughts, the status may change to key pest.

Climatic and edaphic factors favoring population growth of *D. undecimpunctata howardi* are antithetical to factors favoring *E. lignosellus*. Although *D. undecimpunctata howardi* damage is not restricted to certain soil types, it is more likely to be severe where peanuts are grown in heavier, poorly drained soils (Grayson and Poos, 1947; Fronk, 1950). *Diabrotica undecimpunctata howardi* oviposition, egg eclosion, larval survival and adult longevity are enhanced by relative humidities in excess of 75% (Arant, 1929; Campbell and Emery, 1967). High rainfall and medium textured soils, which result in the moist soils necessary for enhancing population growth, are characteristic of the Virginia-Carolina area and certain areas of the southeast where *D. undecimpunctata howardi* is a key pest (Miller, 1943; Hays and Morgan, 1965; Campbell and Emery, 1967; Chalfant and Mitchell, 1967). In contrast, it is only an occasional pest in the southwest (King et al., 1961; Smith and Jackson, 1975).

The vast majority of phytophagous arthropods inhabiting peanut fields are occasional and non-pest species (Smith and Jackson, 1975). Populations of arthropods in these classifications usually are maintained below damaging levels by climatic factors and/or natural enemies. Biologically perturbing factors, such as unnecessary or non-selective insecticide applications, can disturb the balance between the natural regulating agent and pest, and create conditions conducive for outbreaks of occasional or non-pest species. The twospotted spider mite, *Tetranychus urticae* Koch, and carmine spider mite, *T. cinnabarinus* (Boisduval), provide excellent examples of peanut non-pests changing status. Prior to 1970, these mites were considered non-pest species on peanuts throughout the southern United States peanut belt (King et al., 1961; Smith and Jackson, 1975; Campbell, 1978). The heavy use of insecticides alone and

in combination with fungicides, created conditions conducive to spider mite outbreaks resulting in a change in pest status from non-pest to secondary pest (Smith and Hoelscher, 1975a; Smith and Jackson, 1975; Campbell, 1978).

The specific mechanism(s) involved with spider mite outbreaks cannot be elucidated individually; however, population outbreaks are due most probably to a combination of events that, when occurring simultaneously or in close temporal proximity, release the spider mite population restraining mechanisms. Several factors have been identified which contribute to spider mite population outbreaks. Fungicides can destroy the mite parasitic fungi, *Entomophthora* sp., that help regulate mite densities and thus contribute to outbreaks (Campbell, 1978). Insecticides can reduce arthropod natural enemies of mites, as well as possibly create physiological changes in the mites themselves. Hot, dry climatic conditions also contribute to parameters which cause population change. Mite developmental time is much shorter at high temperatures, resulting in an increased net production of new individuals. Dry microclimates prevent fungal spores from germinating. Regardless, heavy pesticide use must have contributed drastically to the change in pest status of spider mites on peanuts, because relaxation of these disruptive practices has resulted in reverting the pests' status back to the original classification in Texas (Smith and Hoelscher, 1975a). Peanut grower acceptance in Texas of a pest management program which has as one tactic a selective insecticidal application technique which reduces the number of applications and the exposure of nontarget species to insecticides (Smith and Hoelscher, 1975b; Smith and Jackson, 1975), has resulted in spider mites presently being reclassified as non-pests.

## ARTHROPOD PESTS

Phytophagous arthropods reported to attack peanuts worldwide fall into 3 classes: Arachnida, Diplopoda, and Insecta. Further, these arthropods occupy at least 2 distinctly different habitats (foliage and soil) which are of paramount importance in the design of management strategies. Identification of pest habitat provides ecological insight crucial to management activities such as (1) ascertaining relationships between particular pests and complexes of natural enemies within distinct habitats (Johnson and Smith, 1981), (2) directing particular management tactics (e.g., an insecticide) at a particular habitat so as to minimize deleterious side effects within the entire system (Smith and Jackson, 1975), and (3) developing relevant sampling plans (allocation, unit size, numbers) for ascertaining pest densities (Southwood, 1978; Jones and Bass, 1979). Such ecological knowledge is consistent with the methodologies presented by Barfield and Stimac (1980) toward design of reliable management strategies for a myriad of pests.

This section has been designed to illustrate the diversity of pests attacking peanuts. Sufficient space and knowledge of pest bionomics are not available here to develop the biology, natural history, damage caused, and tactics usable against every pest known (or reported) to attack peanuts. This problem has been addressed herein by presenting a tremendous volume of information in tabular form with a relevant literature citation(s) to guide the reader to information sources. Further, specific examples of pests have been selected for concentration on varied biologies and natural histories while illustrating management practices from around the world. This should result in an appreciation for

the diversity of pests which attack peanuts and the various ways with which each may be managed or controlled. An in-depth look at where IPM is relevant to peanut insects worldwide will be summarized in the last section of this chapter.

A list of arthropods attacking peanuts worldwide has been compiled (Table 1). Classes, orders, families, genera and species (unless unavailable) of peanut pests are provided. Geographical distribution is listed as well as habitat occupied within the peanut field. References provided are to the earliest or best available source on the biology of each particular pest and are intended to provide a checklist to facilitate entry into the massive literature on peanut pests. Many of these references provide more localized distribution maps and, to some extent, may deal with management tactics available for that particular pest.

Table 1. A world list of arthropods attacking preharvest peanuts, *Arachis hypogaea* L.

Arthropod	Distribution	Feeding Site <sup>1</sup>	Reference
Class: Arachnida			
Order: Acarina			
Family: Astigmataidae			
<i>Sancassania</i> sp. (= <i>Caloglyphus</i> )	South Africa, USA	S	Aucamp 1969, Shew & Beute 1979
<i>Tyrophagus</i> sp.	South Africa	S	Aucamp 1969
Family: Eupodidae			
<i>Penthaleus major</i> (Duges)	Queensland	F	Smith 1946
Family: Tetranychidae			
<i>Mononychus planksi</i> (McGregor)	Brazil	F	Flechtmann 1968
<i>Oligonychus pratensis</i> (Banks)	USA	F	Smith Meyer 1974
<i>Paraplonobia</i> sp.	Australia	F	Feakin 1973, Passlow 1969
<i>Septanychus</i> sp.	Texas	F	Iglinsky & Gaines 1949
<i>Tetranychus arabicus</i> Attiah (= <i>urticaria</i> Koch) (= <i>telarius</i> (L.))	Egypt, Israel USA, Bulgaria, Argentina, India	F	Smith Meyer 1974, Pietrarelli 1976, Gibbons 1976
<i>Tetranychus cinnabarinus</i> (Boisduval) (= <i>Eotetranychus</i> ) (= <i>cucurbitacearum</i> (Sayed))	Cosmopolitan	F	Hill 1975
<i>Tetranychus desertorum</i> Banks	USA	F	Smith & Jackson 1975
<i>Tetranychus equatorius</i> McGregor	Hawaii	F	McGregor 1950
<i>Tetranychus hypogaea</i>	India	F	Gupta 1976
<i>Tetranychus marianae</i> McGregor	Mauritius	F	Mouria 1958
<i>Tetranychus neocaledonicus</i> Andre (= <i>cucurbitae</i> Rahman & Sapra)	India	F	Smith Meyer 1974
<i>Tetranychus tumidellus</i> Prichard & Baker	Turkey	F	Duzgunes 1959
<i>Tetranychus turkestanii</i> (Ugarov & Nikolski) (= <i>atlanticus</i> McGregor)	Bulgaria	F	Atanasov 1971
<i>Tetranychus</i> sp.	Widespread	F	Hill 1975
Class: Diplopoda			
Order: Julida			
Family: Odontopygidae			
<i>Haplothysanus chapellei</i>	Senegal	S	Gillier 1976
<i>Haplothysanus ealanus</i> Attems	Africa	S	Pierrard 1969
<i>Haplothysanus haplothysanoides</i>	Africa	S	Pierrard 1967

Table 1 (Continued)

<i>Haplothysanus oubanguiensis</i>	Africa	S	Pierrard 1968
Pierrard			
<i>Peridotopyge conani</i>	Senegal	S	Gillier 1976
<i>Peridotopyge perplicata</i>	Africa	S	Roubaud 1916
<i>Peridotopyge rubescens</i>	Senegal	S	Gillier 1976
<i>Peridotopyge schoutedeni</i> Attems	Africa	S	Pierrard 1968
<i>Peridotopyge</i> sp.	Africa	S	Raheja 1975
<i>Peridotopyge spinosissima</i> (Silvestri)	Nigeria	S	Misari 1975
<i>Syndesmogenus mimeuri</i>	Senegal	S	Gillier 1976
<i>Tibiomus ambitus</i> (Attems)	Africa	S	Pierrard 1969
<i>Tibiomus gossypii</i> Pierrard	Africa	S	Pierrard 1969
Class: Insecta			
Order: Orthoptera			
Family: Tetrigidae			
<i>Paratetix carinatus</i> Kirby	Nigeria	F	Yayock 1976
Family: Acrididae			
<i>Acridylus patruelis</i> H. S.	Nigeria	F	Yayock 1976
<i>Austracris guttulosa</i> Walker	Asia	F	Cotterell-Dormer 1941
<i>Chondracris rosa</i> DeGeer	Japan	F	Sonon 1940
<i>Chrotogonus hemipterus</i> Schaum	India	F	Kevan 1954
<i>Chrotogonus rotundatus</i> Kirby	Africa	F	Jepson 1948
<i>Chrotogonus senegalensis</i> Krauss	Nigeria	F	Yayock 1976
<i>Chrotogonus trachypterus</i> (Blanchard)	India	F	Srivastava et al. 1965
<i>Colemania spenarioides</i> , Boliver	India	F	Seshagiri Rao 1943
<i>Comipoda calcarata</i> Saussure	Senegal	F	Roubaud 1916
<i>Krausaria angulifera</i> (Krauss)	Nigeria	F	Oyidi 1975
<i>Locusta migratoria migratoria</i> Reiche and Fairmaire	Africa, Asia, Formosa	F	Vrijagh 1932, Jepson 1948
<i>Melanoplus bivittatus</i> (Say)	USA	F	USDA 1979
<i>Melanoplus differentialis</i> (Thomas)	USA	F	USDA 1979
<i>Melanoplus femur-rubrum</i> (DeGeer)	USA	F	USDA 1979
<i>Melanoplus sanguinipes</i> (F.)	USA	F	USDA 1979
<i>Nomadacris septemfasciata</i> Serville	Africa	F	Jepson 1948
<i>Ortbracis</i> sp.	India	F	Gibbons 1976
<i>Oxya velox</i> (F.)	India	F	Hill 1975
<i>Pyrgomorpha cognata</i> Krauss	Nigeria	F	Yayock 1976
<i>Pyrgomorpha vignaudi</i> Guerin- Meneville	Nigeria	F	Yayock 1976
<i>Romalea microptera</i> Palisor de Beauvois	Florida	F	Watson & Bratley 1940
<i>Schistocera gregaria</i> Forskal	Africa, Spain, Cypress, USSR, Turkey	F	Jepson 1948
<i>Zonocerus elegans</i> (Thunberg)	South Africa	F	Hill 1975
<i>Zonocerus variegatus</i> F.	Nigeria	F	Yayock 1976
Family: Tridactylidae			
<i>Trigonidium cinctinelloides</i> Rambur	Nigeria	F	Yayock 1976
Family: Gryllidae			
<i>Liogryllus morio</i> F.	Africa	F	Jepson 1948
<i>Brachytrypes membranaceus</i> Drury	Nigeria	F	Yayock 1976
Family: Grylloidalpidae			
<i>Grylloidalpa africana</i> Pal.	Nigeria	S/F	Yayock 1976

Table 1 (Continued)

<i>Gryllotalpa eryllotalpa</i>			
var. <i>cafta</i>	Egypt	F	Serry 1976
<i>Gryllotalpa gryllotalpa</i> L.	North Caucasus	S/F	Shchegolev & Weronev 1930
<i>Gryllotalpa hexadactyla</i> Perry	Florida	S/F	Metcalf et al. 1962
<i>Scapteriscus abbreviatus</i>	USA	S/F	Wisecup & Hayslip 1943
Scudder			
<i>Scapteriscus acletus</i>	USA	S/F	Metcalf et al. 1962
Rehn & Hebard			
<i>Scapteriscus vicinus</i> Scudder	USA	S/F	Metcalf et al. 1962
Family: Blattidae			
<i>Blattella</i> sp.	Nigeria	F	Yayock 1976
Order: Dermaptera			
Family: Labiduridae			
<i>Anisolabis</i> (= <i>Euborellia</i> )	Israel	S	Melamed-Madjar 1971
<i>annulipes</i> (Lucas)			
<i>Euborellia stali</i> Dohrn	S. India	S	Hill 1975
Order: Isoptera			
Family: Termitidae			
<i>Allodontermes morogorensis</i>	Tanzania	S	Jepson 1948
Harris			
<i>Amitermes evuncifer</i> Silvestri	Nigeria	S	Feakin 1973
<i>Ancistrotermes crucifer</i>	Gambia	S	Feakin 1973
(Sjostedt)			
<i>Ancistrotermes latinotus</i>	Congo	S	Feakin 1973
(Holmgren)			
<i>Coptotermes formosanus</i> Shiraki	China	S	Feakin 1973
<i>Eremotermes nanus</i>	Sudan	S	Feakin 1973
<i>Eutermes parvulus</i> Sjostedt	Africa	S	Roubaud 1916
<i>Macrotermes belliosus</i>	Africa, Sudan	S	Feakin 1973
(Smeathman)			
<i>Macrotermes natalensis</i> Haviland	Africa	S	Roubaud 1916
<i>Microcerotermes</i>			
(= <i>Microtermes</i> )	Africa	S	Hill 1975
<i>parvulus</i> Sjostedt			
<i>Microcerotermes</i> sp.	Gambia, Nigeria	S	Feakin 1973, Yayock 1976
<i>Microtermes thoracalis</i>			
Sjostedt	Africa, India	S	Weidner 1962, Srivastava et al. 1965
<i>Microtermes</i> sp.	Nigeria, Gambia	S	Feakin 1973
<i>Nasutitermes</i> sp.	Malawi	S	Mercer 1977
<i>Odontotermes anceps</i> (Sjostedt)	Kenya	S	Schumutterer 1971
<i>Odontotermes badius</i> (Haviland)	South Africa	S	Feakin 1973
<i>Odontotermes latericius</i>	South Africa	S	Feakin 1973
(Haviland)			
<i>Odontotermes nilensis</i> Emerson	Sudan	S	Feakin 1973
<i>Odontotermes obesus</i> (Rambur)	India	S	Feakin 1973
<i>Odontotermes vulgaris</i> (Haviland)	Africa	S	Roubaud 1916
<i>Odontotermes</i> sp.	Africa, India	S	Hill 1975
<i>Syntermes</i> sp.	Brazil	S	Bastos Cruz et al. 1962
<i>Trinervitermes biformis</i>	India	F	Feakin 1973
(Wasmann)			
<i>Trinervitermes geminatus</i>	Senegal	F	Feakin 1973
(Wasmann)			
Family: Hodotermitidae			
<i>Hodotermes massambicus</i> (Hagen)	Senegal	S	Gillier 1976
Order: Thysanoptera			
Family: Thripidae			
<i>Caliothrips brasiliensis</i> Morgan	Central America	F	Barral & Velasco de Stacul 1969

Table 1 (Continued)

<i>Caliothrips sumipennis</i>	Sudan	F	Clinton 1962
(Bagnall and Cameron)			
<i>Caliothrips indicus</i> Bagnall	India, Africa	F	Corbett 1920, Panchabhai & Thimmaiah 1973
			Schmutterer 1971
<i>Caliothrips sudaneensis</i>	Sudan	F	
(Bagnall and Cameron)			
<i>Enneothrips flavens</i> Moulton	Brazil	F	Almeida et al. 1977
<i>Frankliniella bipinosa</i> Morgan	USA	F	Morgan et al. 1970
<i>Frankliniella fusca</i> (Hinds)	USA, Brazil	F	Morgan et al. 1970, Almeida et al. 1965
<i>Frankliniella occidentalis</i>	USA	F	Smith & Jackson 1975
(Pergande)			
<i>Frankliniella schultzei</i> (Trybom)	Australia	F	Hill 1975
<i>Frankliniella tritici</i> (Fitch)	USA	F	Morgan et al. 1970
<i>Haplothrips gallarum</i> Priesner	Africa	F	Nonveiller 1973
<i>Scirtotrips dorsalis</i> Hood	India, Sri Lanka, Kenya	F	Hill 1975
<i>Selenotrips rubroinctus</i> Giard	Cosmopolitan	F	Anon. 1977
<i>Sericotripsi occipitalis</i> Hood	Africa	F	Hill 1975
<i>Taeniotripsi distalis</i> Karny	India	F	Hill 1975
<i>Taeniotripsi inconsequens</i> Uzel	USA	F	Watson 1923
<i>Taeniotripsi longistylus</i> Karny	Asia	F	Ramakrishna Ayyar 1929
<i>Taeniotripsi sjostedti</i> (Trybom)	Africa, Nigeria	F	Nonveiller 1973, Yayock, 1976
<i>Tripsi tabaci</i> Lindeman	Egypt	F	Serry 1976
Order: Hemiptera			
Family: Miridae			
<i>Adelphocoris</i> sp. nr. <i>apicalis</i>	Nigeria	F	Yayock 1976
Reuter			
<i>Calocoris angustatus</i> Lethierry	India	F	Ballard 1917
<i>Creontiades pallidifer</i> (Walker)	India	F	Ullah 1940
<i>Creontiades pallidus</i> (Rambur)	Africa, India	F	Hill 1975
<i>Cyrtorhinus caricooides</i> Ghauri	Madagascar	F	Ghauri 1970
<i>Cyrtorhinus rectangulus</i> Ghauri	Madagascar	F	Ghauri 1970
<i>Halticus minutus</i> Reuter	Pescadores Islands	F	Maki 1918
<i>Halticus tibialis</i> Reuter	Africa	F	Hargreaves 1932
<i>Megacestum stramineum</i> Walker	India, Madras	F	Ballard 1917
<i>Psallus</i> (= <i>Pseudatomoscelis</i> )	USA	F	Robinson et al. 1972
<i>seriatius</i> (Reuter)			
<i>Spanagonicus albofasciatus</i> (Reuter)	USA	F	Robinson et al. 1972
<i>Senopterocoris laticeps</i> China	Africa	F	China 1944
Family: Lygaeidae			
<i>Aphanus</i> (= <i>Naphius</i> ) <i>apicalis</i>	Bombay, Africa	F	Scudder 1968
(Dallas)			
<i>Aphanus sordidus</i> (F.)	India	F	
			Desphande & Ramras, 1915, Gibbons 1976
<i>Lygaeus rivularis</i> Germar	Nigeria	F	Yayock 1976
<i>Napbius zavattarii</i> (Mancini)	Africa	F	Scudder 1968
<i>Rhynchosciara littoralis</i>			
Distani	Nigeria	F	Misari 1975
Family: Pyrrhocoridae			
<i>Dysdercus fasciatus</i> Signoret	Asia, Africa	F	Bedford 1937
<i>Dysdercus koenigii</i> (F.)	Africa	F	Hill 1975
<i>Dysdercus superstiosus</i> F.	Africa	F	Hill 1975
Family: Coreidae			
<i>Anoplocnemis curvipes</i> F.	Nigeria	F	Yayock 1976
Family: Alydidae			
<i>Mirperus jaculus</i> Thunberg	Nigeria	F	Yayock 1976
Family: Pentatomidae			
<i>Afrisius figuratus</i> Germar	Nigeria	F	Yayock 1976

Table 1 (Continued)

<i>Cydnus</i> sp.	Tanzania	F	Jepson 1948
<i>Menida loriventris</i> Germar	Nigeria	F	Yayock 1976
<i>Nezara pallidoconspersa</i> Stål.	Madagascar	F	Frappa 1931
<i>Nezara viridula</i> (L.)	Cosmopolitan	F	Hill 1975
<i>Piezodorus pallidus</i> Germar	Africa, Nigeria	F	Jepson 1948, Yayock 1976
<i>Piezodorus rubrofasciatus</i> F.	Nigeria	F	Yayock 1976
<i>Agnoscelis</i> sp.	Nigeria	F	Yayock 1976
<i>Aspavia armigera</i> F.	Nigeria	F	Yayock 1976
Family: Cydnidae			
<i>Cyrtomenus mirabilis</i> (Perty)	Brazil	S	Bastos Cruz et al. 1962
<i>Cyrtomenus ciliatus</i> (Palisor de Beavois)	USA	S	Smith & Pitts 1974
<i>Pangaeus bilineatus</i> (Say)	USA	S	Smith & Pitts 1974
<i>Pangeus congruus</i> (Uhler)	USA	S	Smith & Pitts 1974
<i>Tominotus</i> sp.	USA, Brazil	S	Cavalcante et al. 1977
<i>Tominotus communis</i> (Uhler)	USA	S	USDA 1966
<i>Scaptocoris castaneus</i> Perty (= <i>terginus</i> Schioedte)	Brazil, Argentina	S	Brewer 1972
<i>Sebiurus expansus</i> Signoret	Nigeria	?	Yayock 1976
Order Homoptera			
Family: Cercopidae			
<i>Locris</i> sp.	Nigeria	F	Yayock 1976
<i>Poophilus</i> sp.	Nigeria	F	Yayock 1976
Family: Cicadellidae (= Jassidae)			
<i>Austroasca alfaiae</i> (Evans)	Queensland	F	Passlow 1969
<i>Cicadulina arachidis</i> China	Africa	F	Hill 1975
<i>Cicadulina similis</i> China	Africa, Italian Somaliland	F	China 1928
<i>Cicadulina</i> sp.	Africa	F	Hill 1975
<i>Empoasca alecto</i> Ross & Cunningham	Peru	F	Langlitz 1966
<i>Empoasca dolichi</i> Paoli	Uganda, Nigeria	F	Jepson 1948, Yayock 1976
<i>Empoasca fabae</i> Harris	N. S. & Central America	F	Hill 1975
<i>Empoasca facialis</i> Jacobi	Tropical Africa	F	Anon. 1968, Yayock 1976
<i>Empoasca flavescens</i> (F.)	Asia, Africa, Dutch E. Indies	F	Anon. 1974
<i>Empoasca solana</i> De Long	Hawaii	F	Holdaway 1941
<i>Empoasca sundanaica</i> Bergman	W. Java	F	Bergman 1956b
<i>Empoasca</i> sp.	S. America, Africa, India, USA	F	Hill 1975
<i>Erythroneura tripunctula</i> (Melichar)	W. Java	F	Bergman 1956b
<i>Oruus ablicintus</i> Distant	India	F	Sundararaju & Jayaraj 1977
<i>Orosius argentatus</i> (Evans)	W. Java	F	Bergman 1956a
Family: Delphacidae	S. Africa	F	Hargreaves 1931
Family: Dictyopharidae			
<i>Atronotaria</i> sp.	Nigeria	F	Yayock 1976
Family: Fulgoridae			
<i>Uyanus</i> sp.	S. Africa	S	Feakin 1973
Family: Flatidae			
<i>Caenonia</i> spp.	Nigeria	F	Yayock 1976
Family: Aleyrodidae			
<i>Bemisia tabaci</i> (Gennadius) (= <i>gossypiperda</i> Mistra & Lamba) (= <i>inconspicua</i> Quaint)	Cosmopolitan	F	Hill 1975
Family: Aphididae			
<i>Amphorophora</i> ( <i>Hyperomyzus</i> ) <i>lactucae</i> (L.)	Queensland		Behncken 1970
<i>Aphis craccivora</i> Koch (= <i>laburni</i> Kaltenbach) (= <i>leguminosae</i> Theob.)	Cosmopolitan	F	Hill 1975

Table 1 (Continued)

<i>Apis glycines</i> Matsumura	Indonesia	F	Roechan et al. 1978
<i>Apis gossypii</i> Glover	Cosmopolitan	F	Hill 1975
<i>Longitarsus sacchari</i> (Zehntner)	Africa	F	A'Brook 1968
<i>Myzus persicae</i> Sulzer	Cosmopolitan	F	Hill 1975
<i>Rhopalosiphum maidis</i> (Fitch)	Nigeria	F	A'Brook 1968
<i>Rhopalosiphum padi</i> (L.)	Queensland	F	Behncken 1970
<i>Tetraneura nigriabdominalis</i> (Sasaki)	Nigeria	F	A'Brook 1968
<i>Theroaphis ononidis</i> (Kaltenbach)	India	F	Veeresh 1974
Family: Coccidae			
<i>Monophleboides arachides</i>	Belgian Congo	S	Vayssiére 1957
<i>Vayssiére</i>			
<i>Phenacoccus hirsutus</i> (Green)	New Guinea, SE. Asia, Egypt, India, Malaysia, Indonesia	S/F	Anon. 1959
<i>Vrydagha lepisma</i>	Africa	S	Vayssiére 1957
Family: Pseudococcidae			
<i>Dysmicoccus</i> (= <i>Pseudococcus</i> ) <i>brevis</i> (Cockerell)	Cosmopolitan	S	Hosny 1940, Anon. 1955
<i>Ferrisia virgata</i> (Cockerell)	Asia, Africa, Australia and Pacific Is., N., S. & Central America	F	Anon. 1975
<i>Pseudococcus calceolariae</i> Maskell	Mauritius	S	d'Emmerez de Charmony and Gebert 1921
<i>Planococcus</i> (= <i>Pseudococcus</i> ) <i>lilacinus</i> Cockerell	Asia, Africa	?	Hill 1975
<i>Pseudococcus solani</i>	USA	S	Chaffin 1921
<i>Pseudococcus</i> sp.	Africa, S. & Central America, Australia	S	Hill 1975
Family: Tettigometridae			
<i>Hilda patruelis</i> Stål	Africa	S	Hill 1975
<i>Hypothionella caccia</i>	Rhodesia	S	Rose 1962
China & Fennah			
Order: Coleoptera			
Family: Staphylinidae			
<i>Paedarus sabaeus</i> Erichson	Nigeria	?	Yayock 1976
Family: Scydmaenidae			
<i>Scydmaenus chevalieri</i> Vuillet	Africa	S	Roubaud 1916
Family: Scarabaeidae			
<i>Adoretus cibrosus</i> Harris	Rhodesia	S	Broad 1966
<i>Adoretus umbrosus</i> F.	Africa	S	Roubaud 1916
<i>Anomalus antiquus</i> Gyllenhal	Burma	S	Ghosh 1924a
<i>Anomalus atrovirens</i>	Dutch East Indies	S	van Hall 1917
<i>Anomalus phebeja</i> Oliver	Africa	S	Roubaud 1916
<i>Anomalus</i> sp.	USA, Japan, India	S	Anon. 1959
<i>Catalpa lanigera</i> L.	Virginia	S	Grayson 1947
<i>Cyclocephala immaculata</i> Oliver	Virginia	S	Grayson 1947
<i>Eulepidota mashona</i> Arrow	Africa	S	Hill 1975
<i>Heteroligus claudius</i> Klug	Nigeria	S	Lean 1929
<i>Heteronyx brevicollis</i> Blackburn	Australia	F	Smith 1946
<i>Lachnostenra caudata</i>	Australia	F/S	Smith 1936
<i>Lachnostenra</i> (= <i>Holotrichia</i> ) <i>consanguinea</i> (Blanchard)	India	F/S	Bindra & Singh 1971
<i>Lachnostenra fissa</i> (Brenské)	India	F/S	Hill 1975
<i>Oxyctenota versicolor</i> (F.)	India	F	Bhatnagar 1970
<i>Pentodon idiota</i> Herbst	USSR	S	Shchegolev & Weroneb 1930
<i>Phyllophaga ephilida</i> Say	Americas	F/S	Grayson 1947
<i>Phyllophaga micans</i> Knoch	Americas	F/S	Grayson 1947

Table 1 (Continued)

<i>Podalga</i> (= <i>Crator</i> ) <i>cuniculus</i> Burmeister	Africa	S	Roubaud 1916
<i>Popillia japonica</i> Newman	China, Japan, N. America	F/S	Anon. 1952
<i>Rhopaea magnitorus</i> Blackburn	Australia	S	Hill 1975
<i>Scbizomycha africana</i> Cast.	Africa, Sudan, Egypt	F/S	Roubaud 1916
<i>Scbizomycha</i> sp.	Sudan	S	Hill 1975
<i>Strigoderma arboricola</i> F.	USA	S	Hill 1975
<i>Trissodon</i> (= <i>Isodon</i> ) <i>puncticollis</i> Maclure	Queensland	S	Smith 1946
<i>Xylotrupes gideon</i> L.	Rangoon	S	Ghosh 1924b
Family: Buprestidae			
<i>Sphenoptera perroteti</i> Guerin	India	F	Gibbons 1976
Family: Elateridae			
<i>Conoderus</i> sp.	USA	S	Arthur & Arant 1956
<i>Agriotes gurgistanus</i> Falderman	USSR	S	Feakin 1973
Family: Cantharidae			
<i>Chauliognathus</i> sp.	USA	F	Wolf 1916
<i>Silidius apicalis</i> Waterhouse	Nigeria	F	Yayock 1979
Family: Coccinellidae			
<i>Epilachna corrupta</i> Mulsant	USA	F	Anon. 1934
<i>Epilachna similis</i> var <i>assimilis</i> Mulsant	Nigeria	F	Yayock 1976
<i>Epilachna varivestis</i> Mulsant	N. & Central America	F	Anon. 1954
<i>Epilachna vigintioctopunctata</i> F.	Fiji	F	Lever 1940
Family: Melyridae			
<i>Astylus atracaudatus</i> (Blanchard)	Argentina	F	Venica de Nemirovsky 1972
Family: Tenebrionidae			
<i>Gonocephalum simplex</i> (F.)	Africa	S	Hill 1975
<i>Gonocephalum</i> sp.	Africa	S	Hill 1975
<i>Homala</i> sp.	Senegal	S	Roubaud 1916
<i>Zophosis</i> sp.	Senegal	S	Roubaud 1916
<i>Zophosis congesta</i> Sjostedt	Tanzania	S	Jepson 1948
Family: Lagriidae			
<i>Chrysotragus nearei</i> Borchmann	Nigeria	F	Yayock 1976
<i>Lagria villosa</i> F.	Nigeria	F	Yayock 1976
Family: Meloidae			
<i>Coryna apicicornis</i> Guerin-Meneville	Tropical Africa	F	Hill 1975
<i>Coryna bermanniae</i> F.	Nigeria	F	Yayock 1976
<i>Coryna lanuginosa</i> Gerstaecker	Tanzania	F	Jepson 1948
<i>Decatoma affinis</i> Oliver	Nigeria	F	Yayock 1976
<i>Epicauta cinerea</i> Forester	USA	F	Milliken 1921
<i>Epicauta maculata</i> (Say)	USA	F	Milliken 1921
<i>Epicauta pennsylvanica</i> DeGeer	USA	F	Milliken 1921
<i>Epicauta sericans</i> LeConte	USA	F	Milliken 1921
<i>Epicauta vittata</i> (F.)	USA	F	Wolf 1916
<i>Epicauta</i> spp.	Nigeria	F	Yayock 1976
<i>Epicauta immaculata</i> Say	USA	F	Milliken 1921
<i>Mylabris pustulata</i> Thunberg	India	F	Gibbons 1976
<i>Mylabris trifasciata</i> Thunberg	Nigeria	F	Yayock 1976
<i>Mylabris</i> sp.	Widespread	F	Hill 1975
Family: Cerambycidae			
<i>Derobrachus brevicollis</i> Serville	USA	S	Tippins et al. 1968
<i>Sabra centurio</i> Pascoe	Australia	F	Smith 1946
<i>Zygrita diva</i> Thomas	Australia	F	Hill 1975
<i>Xystroceria marginalis</i> F.	Nigeria	F	Yayock 1976
Family: Chrysomelidae			
<i>Acalymma bivittatum</i> Kirsh	Brazil	F	Santos et al. 1975

Table 1 (Continued)

<i>Apophylia nigricollis</i> Allard	Nigeria	F	Yayock 1976
<i>Apophylia murina</i> Gerstaecker	Rhodesia	F	Jack 1922
<i>Barombia humeralis</i> Lab.	Nigeria	F	Yayock 1976
<i>Buphonella nigrovilacea</i> Jacoby	Nigeria	F	Misari 1975
var. <i>metalica</i>			
<i>Colaspis jansseni</i> (= <i>Macrolaspis</i> ) (Bechyne)	Brazil	F	Almeida et al. 1977, Feakin 1973
<i>Diabrotica balteata</i> LeConte	Americas	S/F	Wolfenbarger 1963, Feakin 1973
<i>Diabrotica speciosa</i> Germar	Brazil	S/F	Christensen 1944
<i>Diabrotica undecimpunctata</i> <i>bouvardi</i> Barber	N. America, Senegal	S/F	Hill 1975, Gillier 1976
<i>Diabrotica</i> sp.	USA, S. America	F	Hill 1975
<i>Ergana bicolor</i> Jacoby	Tanzania	F	Jepson 1948
<i>Hallirbotius africana</i> Jacoby	Tanzania	F	Jepson 1948
<i>Luperodes querternus</i> Fairmaire	Uganda, Nigeria	F	Jepson 1948
<i>Mesoplatus cinctus</i> Oliver	Nigeria	F	Yayock 1976
<i>Monolepta australis</i> (Jacoby) (= <i>rosea</i> Blackburn)	Australia	F	Passlow 1969
<i>Monolepta goldinae</i> Bryant	Nigeria	F	Yayock 1976
<i>Monolepta</i> sp. nr. <i>kraatzi</i> Jacoby	Nigeria	F	Misari 1975
<i>Monolepta nigeriae</i> Bryant	Nigeria	F	Misari 1975
<i>Ootbeca bennigseni</i> Weise	Tanzania	F	Misari 1975
<i>Ootbeca mutabilis</i> Sahlberg	Nigeria, E. Africa	F	Hill 1975
<i>Phaedonina areata</i> F.	Nigeria	F	Misari 1975
<i>Podagrion</i> sp. nr. <i>dilecta</i> Dalman	Nigeria	F	Yayock 1976
<i>Systema elongata</i> F.	USA	F	Bissell 1941
Family: Curculionidae			
<i>Alcidodes dentipes</i> (Oliver)	Tropical Africa	F	Hill 1975
<i>Anaemus fuscus</i> Oliver	Senegal	F/S	Roubaud 1916
<i>Cratopus punctum</i> (F.)	Asia	F	Dove & Williams 1971
<i>Cryotzemus cognatus</i> Marshal	India	F	Charan Singh 1978
<i>Dereodus recticollis</i> Marshal	Africa	F	Jepson 1948
<i>Dereodus vagabundus</i> Faust	Nigeria	F	Misari 1975
<i>Diacoderus</i> sp.	Africa	F	Jepson 1948
<i>Graphognathus leucomoma</i> (Boheman) (= <i>imitator</i> ) (= <i>striatus</i> )	South America, Australia, SE. USA, South Africa, New Zealand	F/S	Hill 1975
<i>Graphognathus peregrinus</i> (Buchanan)	USA	F/S	Bartlett et al. 1968
<i>Graphognathus</i> sp.	SE. USA, S. America, Australia, New Zealand, South Africa	F/S	Hill 1975
<i>Hypsonotus</i> sp.	Brazil	F	Araujo et al. 1977
<i>Ischnotraebelus</i> sp.	Nigeria	F	Misari 1975
<i>Mesoleurus dentipes</i> (Marshall)	Rhodesia	S	Broad 1966
<i>Myllocerus discolor</i> Boheman	India	F	Nath & Pal 1971
<i>Myllocerus viridanus</i> F.	India	F	Ramakrishna Ayyar 1922, Gibbons 1976
<i>Myllocerus</i> sp.	India	F	Brar and Sandhu 1975
<i>Naupactus cincteidorsum</i> Hulst	Argentina	F/S	Brewer & Varas 1973
<i>Nematoxerus acerbus</i> Faust	Nigeria	F	Misari 1975
<i>Pachnaeus azurescens</i> Gyllenhal	Cuba	F	Roig et al. 1923
<i>Pachnaeus costatus</i> Perroud	Cuba	F	Roig et al. 1923
<i>Pachnaeus litus</i> (Germar)	Cuba	F	Roig et al. 1923
<i>Pachnaeus psittacus</i> Olivier	Cuba	F	Roig et al. 1923
<i>Pantomorus glaucus</i> (Perty)	Brazil	F	Cavalcante et al. 1974
<i>Priocnemis basquii</i> Hulst	Argentina	F	Brewer & Varas 1973
<i>Protostrophus hirtiventris</i> Marshall	Africa	F	Marshall 1944

Table 1 (Continued)

<i>Protostrophus ocularius</i> Marshall	Africa	F	Marshall 1927
<i>Scepticus insularis</i> Roelofs	Japan	F	Ishiyama 1920
<i>Sitona crinitus</i> (Hulst)	Israel	S	Plaut 1975
<i>Sitona lineatus</i> (L.)	Israel	S	Plaut 1975
<i>Sphingodes globulus</i> Marshal	Tanzania	F	Jepson 1948
<i>Systates alticollis</i> Marshal	Tanzania	F	Jepson 1948
<i>Systates exaptus</i> Marshal	Rhodesia	S	Broad 1966
<i>Systates</i> sp.	Africa	F	Hill 1975
<i>Trichonaupactus densius</i> Hulst	Argentina	F/S	Brewer & Varas 1973
Order: Lepidoptera			
Family: Limacodidae			
<i>Parasa vivida</i> (Walker)	E. & W. Africa	F	Hill 1975
Family: Pyromorphidae			
<i>Atractomorpha crenulata</i> (F.)	India	F	Srivastava et al. 1965
Family: Pyralidae			
<i>Elasmopalpus lignosellus</i> (Zeller)	N., S. & Central America	S	Hill 1975, Smith 1980
<i>Hedylepta</i> (= <i>Lamprosema</i> )	Mauritius	F	Dove & Williams 1971
<i>indicata</i> (F.)			
<i>Loxostege stricticollis</i> L.	North Caucasus	F	Shchegolev & Weroneb
<i>Loxostege verticalis</i> L.	North Caucasus	F	Shchegolev & Weroneb 1930
<i>Maruca testulalis</i> (Geyer)	Cosmopolitan	F	Hill 1975
<i>Stylopalpia costalimai</i> Almeida	Brazil	F	Bastos Cruz et al. 1962, Almeida 1961.
Family: Olethreutidae			
<i>Epinotia opposita</i> Heinrich	Peru	F	Anon. 1942
Family: Tortricidae			
<i>Amorbia emigrella</i> Busch	Hawaii	F	Holdaway et al. 1941
<i>Tortrix dinota</i> Meyrick	E. Africa	F	Jepson 1948
Family: Gelechiidae			
<i>Anarsia ephippialis</i> (Meyrick)	India, Barbados,	F	Bakhetia 1977
<i>Stegasta bosqueella</i> (Chambers)	N. & S. America,	F	Badar 1972
<i>Stegasta capitella</i> (F.)	Venezuela,	F	Briceno 1971
<i>Stomopteryx subtegicella</i> (Zeller) (= <i>nerteria</i> (Meyrick))	India, SE. Asia	F	Hill 1975, Rai 1976
Family: Geometridae			
<i>Asotis reciprocaria</i> Walker	Uganda	F	Hill 1975
<i>Asotis</i> (= <i>Boarmia</i> ) <i>telenaria</i> Schiffermueller	S. & E. Africa	F	Hill 1975
Family: Arctiidae			
<i>Amsacta albistriga</i> (Walker)	India	F	Kareem et al. 1973
<i>Amsacta lineola</i> (Fabricius)	India	F	Bhardwaj & Kushwaha 1976
<i>Amsacta moorei</i> (Butler)	India, Australia	F	Hill 1975
<i>Creationotus transiens</i> Walker	Malaya	F	Gater 1925
<i>Diacrisia obliqua</i> (Walker)	India	F	Hill 1975
<i>Diacrisia virginica</i> (F.)	USA	F	Mitchell 1919
<i>Estigmene acraea</i> (Drury)	USA	F	Smith & Jackson 1975
<i>Estigmene unipunctata</i> Hampson	Uganda	F	Jepson 1948
<i>Spilosoma investigatorum</i> Karsch	Uganda	F	Jepson 1948
Family: Agaristidae			
<i>Agocera rectilinea</i> Boisduval	Nigeria	F	Youdeowei & Oboite 1972
Family: Noctuidae			
<i>Achaea finita</i> (Guenee)	Africa	F	Hill 1975
<i>Agrotis ipsilon</i> (Hufnagel)	Cosmopolitan	F	Hill 1975
<i>Agrotis repleta</i> Walker	Venezuela	F	Briceno 1971
<i>Agrotis segetum</i> (Schiffermueller)	Africa, Europe, USA, Asia, Taiwan, Japan, Indonesia, Sri Lanka	F	Hill 1975

Table 1 (Continued)

Velvetbean Caterpillar	<i>Anticarsia gemmatalis</i> Hubner	N. & S. America, West Indies	F	Watson 1916, Hill 1975
	<i>Autographa</i> (= <i>Phytometra</i> ) <i>gamma</i> (L.)	Bulgaria, USSR	F	Popov et al. 1972
Cotton Bollworm	<i>Euxoa temera</i> (Hubner)	Bulgaria	F	Popov et al. 1972
	<i>Feltia subterranea</i> (F.)	USA	F	Hill 1975
	<i>Heliothis armigera</i> (Hubner)	Cosmopolitan	F	Anon. 1952
	<i>Heliothis dipsacea</i> (L.)	Bulgaria, USSR	F	Shchegolev & Weroneb 1928, Popov et al. 1972
	<i>Heliothis peligera</i>	USSR	F	Shchegolev & Weroneb 1928
	<i>Heliothis punctigera</i> Walker	Asia, Cocos-Keling Is., Australia, Pacific Is., N., S. & Central America	F	Anon. 1977
	<i>Heliothis virescens</i> (F.)	USA	F	Hill 1975
	<i>Heliothis zea</i> (Boddie)	N., S. & Central America	F	Hill 1975
	<i>Ilattia octo</i> (Guenee)	China	F	Wu 1977
	<i>Mamestra</i> (= <i>Barathra</i> ) <i>brassicae</i> (L.)	Bulgaria	F	Shchegolev & Weroneb 1930
	<i>Mocis repanda</i> (F.)	Brazil	F	Bastos Cruz 1962
	<i>Mocis undata</i> (F.)	China	F	Wu 1977
	<i>Plusia acuta</i> Walker	Tanzania	F	Jepson 1948
	<i>Plusia chalybeus</i> (Esper) (= <i>Chrysodeixis chalybeus</i> Esper)	India, China	F	Wu 1977, Rabindra et al. 1975
	<i>Plusia limbirena</i> Guenee	Kenya	F	Jepson 1948
	<i>Pbytometra</i> (= <i>Plusia</i> ) <i>orichalcea</i> F.	Israel, Ethiopia, India	F	d'Emmerez de Charmoy and Gebert 1921
	<i>Pbytometra gamma</i> L.	USSR	F	Shchegolev & Weroneb 1929
	<i>Plusia signata</i> (F.)	India	F	Srivastava et al. 1965
	<i>Pseudoplusia includens</i> Walker	USA	F	Canerday & Arant 1966
	<i>Seleta dorsalis</i> Butler	W. Africa	F	Vayssiere & Mimeur 1925
	<i>Spodoptera eridania</i> Cramer	Venezuela	F	Briceno 1971
	<i>Spodoptera exempta</i> (Walker)	Asia, Africa, Australia, Pacific Is.	F	Hill 1975
	<i>Spodoptera exigua</i> (Hubner)	Cosmopolitan	F	Hill 1975
	<i>Spodoptera frugiperda</i> (J. E. Smith)	N., S. & Central America, West Indies	F	Anon. 1977, Luginbill 1928
	<i>Spodoptera latifascia</i> (Walker)	Venezuela	F	Briceno 1971
	<i>Spodoptera littoralis</i> (Boisduval)	Europe, Asia, Africa	F	Hill 1975
	<i>Spodoptera litura</i> (F.)	Europe, Asia, Africa, Australia, Pacific Is., Egypt, India	F	Hill 1975, Serry 1976, Gibbons 1976
	<i>Spodoptera ornithogalli</i> (Guenee)	N., S. & Central America, West Indies	F	Anon. 1977
	<i>Trichoplusia ni</i> (Hubner)	Cosmopolitan	F	Anon. 1977
	Family: Liparidae			
	<i>Dasylophia georgiana</i> Fawc.	Nigeria	F	Yayock 1976
	<i>Euproctis fasciata</i> Walker	Nigeria	F	Misari 1975
	<i>Orgyia mixta</i> Snell	Nigeria	F	Misari 1975
	Family: Sphingidae			
	<i>Hippotion celerio</i> (L.)	Nigeria	F	Youdeowei & Oboite 1972
	Family: Lycaenidae			
	<i>Strymon melinus</i> (Hubner)	USA	F	Smith & Jackson 1975
	Family: Pieridae			
	<i>Eurema daira</i> (Godart)	Venezuela	F	Briceno 1971
	Order: Diptera			
	Family: Cecidomyiidae			
	<i>Mycodiplaxis</i> sp.	Japan	F	Yukawa & Tanaka 1976

Table 1 (Continued)

Family: Leptidae	Senegal	S	Roubaud 1916
Family: Empididae	Senegal	S	Roubaud 1916
Family: Platystomatidae			
<i>Rivellia</i> sp.	Africa	S	Seeger & Maldaque 1960
Family: Lauxaniidae			
<i>Homoneura</i> sp.	Nigeria	?	Yayock 1976
Family: Chloropidae			
<i>Hippelates pusio</i> Loew	USA	S	Snoddy et al. 1975
<i>Pachylophus</i> sp.	Nigeria	?	Yayock 1976
Family: Muscidae			
<i>Atherigona</i> sp.	Nigeria	?	Yayock 1976
Order: Hymenoptera			
Family: Formicidae			
<i>Atta capiguara</i> Goncalves	S. & Central America	F	Amante 1967
<i>Camponotus maculatus</i> F.	Nigeria	?	Yayock 1976
<i>Dorylus fulvus</i> Westwood	Senegal	S	Roubaud 1916
<i>Dorylus orientalis</i> Westwood	India	S	Roonwal 1976
<i>Euponera tenuirostris</i> Mayr	Africa	S	Roubaud 1916
<i>Messor</i> spp.	E. Africa	F	Hill 1975
<i>Monomorium bicolor</i> Emery	Africa	S	Roubaud 1916
<i>Solenopsis fugax</i> Latreille	USSR	S	Shchegolev & Weroneb 1930
<i>Tetramorium caespitum</i> L.	USSR	S	Shchegolev & Weroneb 1930
Family: Megachilidae			
<i>Megachile argenteata</i> F.	S. Kazakstan	F	Yakhontov & Rxohtob 1932

<sup>1</sup> Soil (S) and/or Foliage (F)

Table 1 represents the most extensive compilation of information available on pests of peanuts worldwide, with more than 360 species listed from Asia, Africa, Europe, North America, South America and Australia. This large, diverse pest fauna is not unique for peanuts but is rather characteristic of the pest diversity of most agronomic Leguminosae (Singh et al., 1978a; van Emden, 1980). Peanuts ranked tenth from the top in a list of 77 world crops ranked in descending order as to number of pest species (van Emden, 1980). Generally, most peanut pests are highly polyphagous and extremely mobile, resulting in a wide geographical distribution.

The remainder of the present section focuses on 2 general groups of pests: foliage and soil inhabitants. These groups are represented by foliage inhabiting Lepidoptera (foliage consumers); aphids, spider mites, and thrips (intracellular feeders), and *E. lignosellus*, *Diabrotica* spp., and white grubs by soil inhabiting. Specific biologies, natural histories, management tactics and problems induced in peanuts worldwide are detailed for these pests in both habitats. This approach is consistent with needs as outlined in the previous section on IPM philosophy and will be crucial to a discussion on worldwide variations on management approaches for peanut pests.

Both the entries in Table 1 and the more detailed presentation of select representatives from 2 pest groups are designed to focus on the peanut ecosystem. Many of the pests discussed are polyphagous; however, ample space does not exist here to discuss the myriad of plant species attacked nor any resultant complications on the dynamics of individual pests as a result of sequences of host plants fed upon. That many of these pests are mobile and frequently move

among peanuts, other cultured plants and native vegetation is axiomatic to the problem. Such interplant movement undoubtedly effects pest dynamics and subsequent pest status (Stimac and Barfield, 1979; Barfield and Stimac, 1980, 1981); however, the details of these intricate ecological/biological relationships are not dealt with in this section. The last section of this paper will provide the details of what is and is not known about such relationships and will chart a course toward improved management of peanut pests worldwide. First, we must provide adequate details about select peanut pests to set the stage for a compare and contrast approach to IPM worldwide in the peanut agroecosystem.

## FOLIAGE INHABITING PESTS

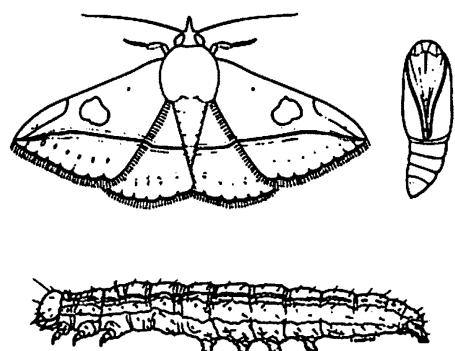
Foliage inhabiting phytophagous arthropods may be divided into 2 groups according to method of feeding and characteristic injury inflicted to the peanut plant. These are (1) foliage consumers which remove foliage with mandibulate mouthparts (orders Orthoptera, Coleoptera, Lepidoptera, Hymenoptera) and (2) intracellular feeders which extract plant cell contents by aspiration with piercing-sucking mouthparts (orders Acarina, Thysanoptera, Hemiptera, Homoptera) (Table 1). Current management strategies, as well as future alternatives, are consistent with this division of foliage pest types. Design of management strategies should be dependent upon pest type, not simply the particular species involved unless divergence in pest biologies dictates species separation. Otherwise, no consistent approaches can result with more than merely local utility.

Arthropods are poikilotherms; thus, these pests are subject to changes in rates of reproduction, development, consumption, movement and mortality as a function of local physical environment. Examples herein cannot explore these intricate, dynamic relationships in more than merely a cursory manner. Nevertheless, sufficient information exists to allow a useful comparison among pests which remove foliage and those that attack cells internally and, in the process, infect the plant with diseases.

### Foliage Consumers

These arthropod pests damage peanuts by removing foliage and thus diminish photosynthetic substrate. Significant yield loss can occur if the plant is in a susceptible phenological stage (temporal tolerance), and the pest population removes a sufficient amount of foliage. The fact that some foliage may disappear does not automatically make the foliage consumer a pest.

**Lepidoptera.** Most of the foliage consuming peanut pests worldwide belong to the insect order Lepidoptera. These varied insects have similar life history strategies but differ in popu-



lation attributes such as developmental time, reproductive rate, consumption rate, longevity, propensity to move, and natural enemy induced mortality. The following examples will serve to highlight the similarities and differences among foliage feeding lepidopteran pests of peanuts worldwide. Initial information on the individual species mentioned is referenced in Table 1.

**Biology.** Species in the lepidopteran families Arctiidae, Noctuidae, Pyralidae and Gelechiidae constitute the major defoliating pests. Most species are polyphagous and host on a wide range of grasses, legumes and/or other plants. The adults characteristically are highly vagile and may move great distances from their pupation sites.

The genus *Spodoptera* contains 5 economically important pest species on peanuts: *S. frugiperda* J. E. Smith, *S. exigua* (Hubner), *S. ornithogalli* (Guenee), *S. littoralis* (Boisduval), and *S. litura* (F.) (Brown and Dewhurst, 1975; Smith and Jackson, 1975). The eggs of all 5 species are laid in scale-covered masses either on the peanut foliage and stems, or the vegetation of host plants adjacent to or within peanut fields. Upon hatching, larvae initially are gregarious and skeletonize the leaf surface. Later instars disperse and become solitary. Larval development requires 2-3 weeks with pupation occurring in the soil. Moths emerge ca. 1 week after pupation with the number of generations per year changing with latitude. In the tropics and subtropics, continuous breeding occurs. Detailed biologies are available for *S. frugiperda* (Vickery, 1929; Luginbill, 1928), *S. ornithogalli* (Crumb, 1929), *S. exigua* (Wilson, 1932, 1934), *S. littoralis* (Hill, 1975) and *S. litura* (Hill, 1975).

Developmental biology of *S. frugiperda* and *S. exigua* fed peanut foliage did not differ drastically from the general format already given. Larval development of *S. exigua* in laboratory experiments was 15 days, and pupal development was 7 days. Eighty-three percent of the larvae pupated and 88% of the pupae emerged as adults (Verma et al., 1974). *Spodoptera frugiperda* developmental time (egg to adult) was ca. 25 days when fed peanut leaves (cv. Florunner) from 45-92 day old plants. However, developmental time increased to 28 days when leaves from 92-120 day old plants were used (Barfield et al., 1980).

*Heliothis armigera* (Hubner), *H. zea* (Boddie), and to a lesser extent *H. virescens* (F.) cause severe, but sporadic defoliation. *Heliothis armigera* is present in the Old World, while *H. zea* and *H. virescens* are New World species. Eggs are laid singly on the foliage, stems and inflorescences with the newly hatched larvae preferably feeding on leaves in the terminal buds. Larvae are extremely variable in color. Larval development on peanut leaves requires 35 and 30 days for *H. zea* at 26 and 30°C, respectively (Huffman and Smith, 1979); and 25 days for *H. armigera* (Pretorius, 1976). Pupation occurs in the soil with the adult emerging in 8-12 days (Isley, 1935; Pretorius, 1976). The entire life cycle lasts about 4-6 weeks on peanuts. Mortality and larval developmental time increased when larvae feed on peanut foliage as compared to other cultivated crops (Pretorius, 1976; Huffman and Smith, 1979).

*Anticarsia gemmatalis* Hubner is a New World pest whose immature stages feed predominately on legumes (Watson, 1916). A fairly complete literature compilation on this insect can be found in Ford et al. (1975) and Moscardi (1979). Mean developmental periods for most life stages of *A. gemmatalis* across a broad range of constant and variable temperatures were derived by Johnson (1980). Mean egg-to-adult development time ranged from 90 days (15.6°C) to

23 days (37.8°C). Studies were conducted using artificial diet. Development and oviposition studies on excised peanut foliage were reported by Nickle (1976).

*Feltia subterranea* (F.) is a New World pest whose larvae are nocturnal feeders. The eggs are laid singly on the peanut plant and are often confused with *H. zea* eggs. *Feltia subterranea* eggs have 36-40 longitudinal ribs (Crumb, 1929), whereas *H. zea* eggs have fewer. Newly hatched larvae feed on the foliage similar to *Heliothis* spp. Larvae soon become nocturnal feeders and hide in the soil or trash beneath the plant during the day (Snow and Callahan, 1968). Larval feeding damage by the later larval instars of *F. subterranea* is easily distinguished from other defoliators because larvae cut the leaflets off at the petiole and feed on the excised leaflets on the soil. Leaf stems appear to be the leaflets snipped off rather than the ragging appearance left by other foliage consumers. The larval stage develops in ca. 24 days (Snow and Callahan, 1968) with pupation in the soil. The pupal stage lasts for ca. 16 days at 25°C (Lee and Bass, 1969). A complete life cycle should take 33-89 days dependent on ambient field temperatures (Lee and Bass, 1969).

The arctiids—*Amsacta moorei* (Butler), *A. albistriga* (Walker), *A. lactinea* (Cremer), and *Diacrisia obliqua* (Walker)—are major defoliators in India (Rai, 1976). Moths of *A. moorei* begin emerging after the first heavy monsoon shower, mate, and oviposit in groups of small rows on the lower surface of the leaves of peanuts and weeds (Ramaswamy et al., 1968). Newly hatched larvae feed gregariously during the early instars (Mathur, 1966). Dispersal to solitary feeding occurs in approximately the 3rd instar. Pupation occurs both in cultivated fields and land adjacent to cultivated fields (Patel and Patel, 1965). A portion of the adults emerge after 6-34 days while the remaining complement delays emergence until the onset of the next monsoon (Rai, 1976). The number of generations vary from 1 to 3 dependent upon geographic location (Singh and Singh, 1956; Bindra and Kittur, 1961; Yadava et al., 1966). The biologies of *A. albistriga*, *A. lactinea* and *D. obliqua* are very similar to *A. moorei* with a few minor exceptions (Sen and Makherjee, 1955; Nagarajan et al., 1957; Pandey et al., 1968; Ramaswamy and Kuppuswamy, 1973; Rai, 1976).

*Stomopteryx subsecivella* (Zeller) eggs are laid 1-2 per leaf and seldom on the stem (Rai, 1976). Newly hatched larvae mine into the stem for 10-15 days before pupating inside the leaf or in leaves folded together by the larvae (Krishnanda and Kaiwar, 1965). Multiple generations (4-5) occur each year as the generation time is less than 1 month (Rao et al., 1962; Yang and Liu, 1966; Gujrati et al., 1973).

Other gelechiids, *Stegasta bosqueella* (Chambers) and *S. capitella* (F.), have been considered pests of peanuts in the New World (Bondar, 1928; Walton and Matlock, 1959; Briceno, 1971; Wall and Berberet, 1980). Larval feeding is restricted to the unopened leaf buds, which causes the unfolded mature leaves to have symmetrical damage on either side of the midrib (Arthur et al., 1959; Wall and Berberet, 1979). Although this damage may be readily apparent, yield losses resulting from defoliation are questionable (Wall and Berberet, 1979).

Development of *S. bosqueella* eggs requires 66.5°C degree days above 12.2°C (Wall and Berberet, 1980). Females laid an average of 16 eggs in the laboratory, a figure probably much below that for field populations. Three genera-

tions occur annually with a generation completed in as few as 23 days when temperatures are high (Wall and Berberet, 1980).

**Movement.** Reference has been made to the highly mobile, polyphagous nature of many lepidopteran pests of peanuts. A problem in dealing with these pests is not the type damage inflicted; rather, it is the severe shortage of information on sources of infestation, causal mechanisms responsible for infestation, and limited research oriented towards understanding how movement and polyphagy affect pest status. Barfield and Stimac (1981) discussed mobility and polyphagy as processes complicating the understanding of population dynamics. An example of how these mobile, polyphagous organisms might filter through a corn-peanut-soybean cropping system was presented by Barfield (1979). Stimac and Barfield (1979) have outlined the role of wide area movement in management decisions made at the individual farm level. Since peanuts are grown either adjacent to or in rotation with other host plants usable by many peanut pests, researchers must begin to understand the role of movement and polyphagy (as well as mortality, feeding, development, etc., already mentioned) in the status of peanut pests.

**Damage.** Foliage consuming pests usually are considered to be occasional, secondary or non-pests, although in some geographic areas they may be key pests. Several defoliating species may individually span all 3 pest categories and often occur simultaneously causing varying levels of plant defoliation. Since the primary plant injury by these pests is physical removal of photosynthetic leaf area (defoliation), the relationship between insect populations, defoliation levels, plant age, and yield at harvest can be examined without specific regard to the pest species. Peanut defoliation by insects and the resulting losses in plant productivity represent complex, dynamic processes which can be best examined by analysis of relevant component parts: plant defoliation and insect foliage consumption.

Insect defoliation of peanuts has been studied predominantly through imposition of defoliation by mechanical methods or infesting plants with varying levels of a given pest species. Three *S. frugiperda* larvae per 20-day old peanut plant caused severe defoliation within 15 days by consuming 1/2 the plant weight (King et al., 1961). Mowing to remove 33% of the plant foliage (cv. Florunner) resulted in lower yields when plants were 70-110 days old (Greene and Gorbet, 1973). On spanish peanuts, removal of 75% of the foliage before first bloom or 50% of the foliage after first bloom did not affect yield or quality adversely (King et al., 1961). Defoliation levels of 50 and 100% (cv. Dodoma Edible) at 4, 6, 8, 10, 12 and 14 weeks after sowing in Tanzania, Africa, produced pod yield reductions (Enyi, 1975). Complete defoliation reduced seed weight and pod number at all age intervals while 50% defoliation during peg formation (8-12 weeks after sowing) only reduced seed weight.

Mechanical simulation of *S. bosqueella* damage to peanut (cv. Spanhoma) terminals showed that plant leaflets compensated for damage (holes in leaves) during growth, especially when the damage was confined to small terminal leaflets (Wall and Berberet, 1979). Feeding by a single individual of this pest during larval development resulted in a 40 cm<sup>2</sup> reduction of mature leaf area (ca. 4 leaflets/individual).

Smith and Barfield (unpublished) hand defoliated 0, 25, 50, 75 and 100% of the leaflets from peanuts (cv. Starr) at weekly intervals beginning when the

plants were 35 days post emergence and continuing until 10 days prior to harvest. Different peanut plots received a designated weekly defoliation with no compounding of defoliation. These data are described in the 3-dimensional response surface depicting the relationships of plant age, % defoliation and % reduction in yield (Figure 2). This response surface supplies a more complete description of defoliation and yield than the defoliation experiments described earlier (King et al., 1961; Greene and Gorbet, 1973; Enyi, 1975), but is in general agreement with previous results. Peanut yield is most susceptible to defoliation from 70-80 days post emergence and practically immune to yield reductions from defoliation prior to bloom initiation and near harvest (Figure 2). This response surface (Figure 2) is in close agreement with Greene and Gorbet (1973), Enyi (1975) and Williams et al. (1976) where peanut yields were most susceptible to defoliation 80-90 days after sowing. Jones et al. (1982) utilized a mobile, infrared gas analyzer and mylar draw-down chamber to measure photosynthetic rate of peanuts when defoliated 0, 25, 50, 75 and 100% at 2-week intervals. Data taken weekly on all defoliation levels measured plant recovery capability and were used to develop a peanut plant growth model sensitive to defoliation, foliar disease and water stress. Much of the data input for model development, as well as initial model structure, is available (Mangold, 1979; Wilkerson, 1980). This approach, while differing markedly from the single defoliation-yield measure design, reaffirmed that 75-85 day old peanuts were most susceptible to yield reduction from defoliation.

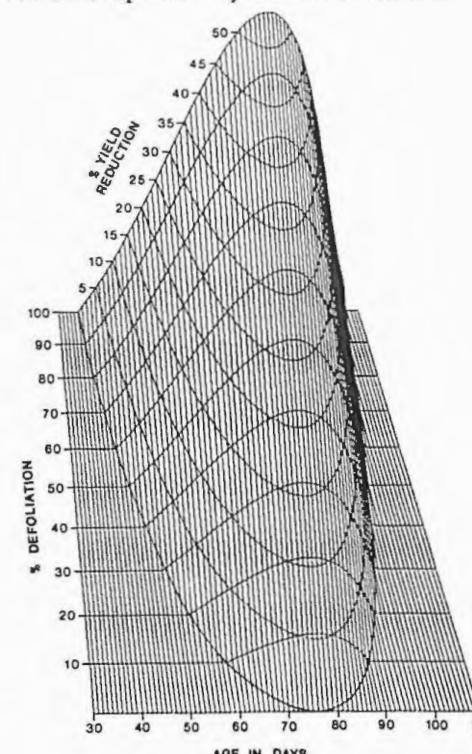


Fig. 2. Response surface depicting the relationship of plant age, % defoliation and % yield reduction for a spanish peanut variety.

Consumption of flowers by foliage consumers should have little effect on pod production and final yield. Dehiscence and pollination occur almost simultaneously with petal expansion in the early morning hours. Breaking the hypanthium near its base 4 hours after anthesis does not prevent fruit development (Smith, 1950). Thus, flower consumption, except during the first 4 hours after anthesis, should not seriously reduce yield. However, inflorescence consumption by insects prior to hypanthium elongation would prevent pollination and could be detrimental to pod set. If insect feeding on inflorescences did prevent normal pod setting, the peanut plant could compensate by producing more flowers (Smith, 1954) as the flowering rate is determined by previous fruit (pod) setting and pollination. Thus, the peanut plant possesses a mechanism to restore lost pods (or pod precursors) by producing more inflorescences; i.e., physiological tolerance. The damage window is limited to 4 hours per day and the plant is temporally most sensitive when inflorescences destined to become fruit are most prevalent.

The relevance of insect consumption of peanut flowers can be further developed by examining data resulting from cross-pollination by native bees and honeybees (Hammons et al., 1963). Observations revealed bees are most abundant in peanut fields in the early morning when the most efficient natural pollination occurs (Hammons et al., 1963). When plants were caged to prevent bee visitation, genetic markers revealed cross-pollination was negligible; while plants available for bee visitation resulted in levels of cross-pollination ranging from 0-2.37% with most cases less than 0.5% (Leuck and Hammons, 1965a, b; Hammons and Leuck, 1966; Girardeau et al., 1975). These data support the concept that pollination occurs rapidly with petal expansion (Smith, 1950), as evidenced by the extremely low level of cross-pollination by bees, and that insect activity, such as flower consumption, would cause only a small yield reduction. Girardeau and Leuck (1967), however, found some evidence that bee tripping of flowers may increase self-pollination and yield in some varieties. This evidence was not substantial enough for changing the current concept that flower consumption by insects does not drastically curb yield.

Insect populations consume foliage at a rate dependent upon species diversity, population density and population age distribution. Different pest species have unique foliage consumption rates and resulting total foliage consumption. Age distribution of a pest population also governs consumption rate. For example, *S. frugiperda* consumes an average of ca. 1.67 cm<sup>2</sup>/day during the medium larval stages and ca. 9.99 cm<sup>2</sup>/day during the large larval stages (Barfield et al., 1980) with total consumption ca. 100 cm<sup>2</sup> (Smith and Barfield, unpublished). *Heliothis zea* larvae consume a total of 176 and 195 cm<sup>2</sup> foliage at 26 and 30 C, respectively (Huffman and Smith, 1979), while *F. subterranea* average 168 cm<sup>2</sup> (Snow and Callahan, 1968). The last 2 instars of *F. subterranea* and *H. zea* consume 73-97% of the total foliage consumed. Daily consumption of peanut (cv. Florunner) foliage by *A. gemmatalis* across 6 constant temperatures demonstrated that most consumption occurred in the penultimate and ultimate instar. Consumption by the last instar ranged from 0.46g/larva/day (21 C) to 0.04g/larva/day (35 C) (Nickle, 1976).

Average cumulative foliage consumption rates by populations of lepidopterous larvae should follow the general shape of the fall armyworm (*S. frugiperda*) consumption rate (Figure 3; Smith and Barfield, unpub. data), but with differ-

ent, species-specific parameters. Young larvae consume small amounts of foliage with foliage consumption increasing as age progresses until the population begins to pupate. Since pupae do not feed, the feeding rate then decreases as an increasing proportion of the population reach the pupal stage. Variability in the duration of larval development (Huffman and Smith, 1979) prevents the abrupt cessation of foliage consumption by a population that would be evident with individuals.

The phenological stage of the plant available when foliage consumption occurs can conversely affect the population dynamics of certain defoliators. Changes in plant phenology are reflective of physiological processes internal to the peanut which, in turn, alter the nutritive value of the foliage. Differences in fall armyworm population parameters (developmental time, consumption, oviposition and longevity) as a function of peanut plant age consumed emphasize the impact of plant phenology on pest dynamics. *Spodoptera frugiperda* larvae fed peanut leaves (cv. Florunner) from plants 67-92 days old (peak pod set and onset of pod fill) had higher consumption rates than larvae fed leaves from 45-70 (vegetative growth and initial flowering) or 92-120 day old plants (peak pod fill and onset of leaf senescence) (Barfield et al., 1980). However, larvae fed leaves from 92-120 day old plants took significantly longer ( $\alpha = .05$ ) to complete development to adults. Adult females resulting from the larvae with the highest consumption rates had a shorter longevity but laid more eggs. Leuck and Hammons (1974a) observed *S. frugiperda* foliage feeding was greatest on the most vigorously growing peanut plants, which could have represented preference for high nutritive value foliage.

Peanut defoliation by insects is dynamic with a multitude of interactive processes occurring simultaneously. Practical utility of a model capturing the essence of this process was developed by Smith and Barfield (unpublished data) in a Peanut Insect Defoliation Model used in Texas IPM programs for peanuts. Data from the plant defoliation response surface (Figure 2) were coupled with age specific feeding rates for larvae of *S. frugiperda* (Figure 3), *S. exigua*, *S. ornata*

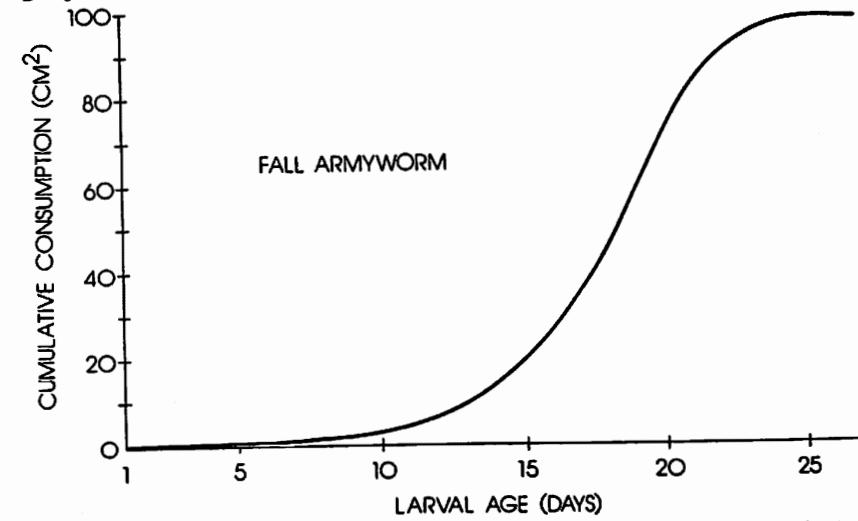


Fig. 3. Spanish peanut foliage consumption by a larval population of *Spodoptera frugiperda* J. E. Smith at 26 C.

*thogalli* and *H. zea* to create a computerized model for predicting yield loss. Inputs into the model (from field scouts) include planting date, date of insect sampling, age distribution and density of defoliating larvae by species and desired prediction interval. The model converts sampling date to peanut phenological time (as chronological time based on planting date), allows the larvae to grow and defoliate the plant, and predicts estimated yield loss (as % yield loss) at desired time intervals following the sampling date. A maximum 7-day prediction interval from sampling is recommended to reduce error due to changes in pest density parameters.

**Management. Natural Enemies.** Numerous listings of natural enemies of lepidopterous defoliators are available. Compiled records are given in Table 2 as a guide to a source; thus, no attempt is made to rewrite a complete list. Although the parasite fauna of foliage consumers is rich, few efforts have been made to evaluate the impact of natural enemies on suppressing foliage consumer populations. Luna (1979) and Collins (1980) made significant progress in understanding natural mortality due to predators of Lepidoptera in soybeans. The techniques appear usable in peanuts, where many of the same pests (e.g., *A. gemmatalis* and *H. zea*) occur.

Two studies in the southwestern United States (Sears and Smith, 1975; Wall and Berberet, 1975) evaluated the impact of extant natural enemies of certain lepidopterous pests. Mean seasonal lepidopteran larval parasitism on peanuts in Oklahoma ranged from 1.5-63.3% with the species and incidence of parasitism (%) as follows: *S. bosqueella* (21.4), *S. frugiperda* (41.8), *S. ornithogalli* (63.3), *S. exigua* (42.9), *Strymon melinus* (Hubner) (62.5), *H. zea* (57.5), *Estigmene acrea* (Drury) (15.6), *F. subterranea* (53.8), *A. gemmatalis* (1.5), *Trichoplusia ni* (Hubner) (38.7) and *Platynota nigrocervina* Walsingham (25.0) (Wall and Berberet, 1975). This 3-year study concluded that larval parasitism had a definite impact on suppressing foliage consumer populations and that conservation of natural enemies was necessary in preventing these pests from achieving key pest status.

Table 2. Resources for information on natural enemies of lepidopterous foliage consumers on peanuts.

Pest Species	References
<i>Spodoptera frugiperda</i>	Bass & Arant 1973, Wall & Berberet 1975, Ashley 1979
<i>S. exigua</i>	Wall & Berberet 1975, Rai 1976
<i>S. ornithogalli</i>	Bottrell 1969, Wall & Berberet 1974, Wall & Berberet 1975
<i>S. litura</i>	Rai 1976
<i>Heliothis zea</i>	Sears & Smith 1975, Wall & Berberet 1975
<i>H. armigera</i>	Rai 1976, Singh et al. 1978b, Stoeva 1973, Bharnagar & Davies 1978
<i>Feltia subterranea</i>	Wall & Berberet 1975
<i>Amsacta moorei</i>	Rai 1976
<i>A. albistriga</i>	Rai 1976, Sundaramurthy et al. 1976
<i>Diacrisia obliqua</i>	Ramaseshiah 1973, Rai 1976
<i>Stomopteryx subsecivella</i>	Rai 1976
<i>Stegasta bosqueella</i>	Badar 1992, Wall & Berberet 1975
<i>Anticarsia gemmatalis</i>	Bass & Arant 1973, Singh et al. 1978b

In Texas, Sears and Smith (1975) made a more detailed study of the population ecology of *H. zea* using ecological life tables. Egg parasitism by *Trichogramma* sp. ranged from 3.3% to a high of 83.3% and increased with suc-

sive generations. A nuclear polyhedrosis virus was responsible for high levels of larval mortality, killing in excess of 90% in some generations. Total generation real mortality ranged from 88.13 - 99.97%. The impact of natural mortality is easily visualized when 666,707 eggs per hectare yield only 3,564 large larvae per hectare (Sears and Smith, 1975). If no mortality had occurred, the large larval population would have exceeded 5.5 larvae per row meter, which is damaging at mid-season. Natural mortality, however, reduced the population to less than 0.3 large larvae per row meter which is decidedly not economically important.

Parasitism of *A. gemmatalis* larvae on peanuts appears to be extremely low in all reports. Nickle (1976) found only 1.5% of *A. gemmatalis* larvae collected in peanuts parasitized, which represented the lowest % parasitism of the 6 Lepidoptera species studied. Berberet (1978a) also reported similar differences between *A. gemmatalis* and *H. zea* parasitism in peanuts. *Heliothis zea* larval parasitism by *Microplitis croceipes* (Cresson) and *Euclatoria armigera* (Coquillett) was 61 and 24%, respectively, whereas *A. gemmatalis* parasitism was nil. *Anticarsia gemmatalis* parasitism was again the lowest of the 10 foliage inhabiting Lepidoptera investigated by Wall and Berberet (1975).

Conservation of extant natural enemies is an important aspect in management of foliage consumers (Smith and Hoelscher, 1975a, b; Smith and Jackson, 1975; Wall and Berberet, 1975; Mangold, 1979). The use of biologically selective insecticides (e.g., *Bacillus thuringiensis* Berliner; Sams and Smith, 1980), ecologically selective application techniques (Smith and Jackson, 1975) and strict adherence to economic thresholds (Smith and Hoelscher, 1975a) helps minimize: (1) nontarget pest resurgence by conserving natural enemies, (2) target and nontarget pest resistance to insecticides by reducing exposure to selection pressures, and (3) changes in pest status.

**Insecticides.** Numerous modern insecticides are very effective in controlling foliage consuming Lepidoptera larvae on peanuts when applied as sprays, baits or dusts (van der Laan and Ankersmit, 1951; Arthur et al., 1959; King et al., 1961; Bastos Cruz et al., 1962; Vittal and Saroja, 1965; Stoeva, 1968; Venkataraman et al., 1970; Morgan and French, 1971; Castro et al., 1972; Bass and Arant, 1973; Feakin, 1973; Kareem et al., 1974; Hill, 1975; Morgan and Todd, 1975; Sinha et al., 1975; Harvey, 1976; Rai, 1976; Sada-kathulla et al., 1978; Urs and Kothai, 1977; Berberet, 1978a, b, 1979; Bass, 1979; Berberet and Guilavogui, 1980; Sams and Smith, 1980). Within the New World, *S. exigua* (Brown, 1961; Cobb and Bass, 1975), *S. frugiperda* (Young, 1979), *H. zea* (Brown, 1968) and *H. virescens* (Nemec and Adkisson, 1969) have shown high levels of resistance to certain insecticides. Insecticidal efficacy against certain species may be dependent upon the status of regional insecticidal resistance; thus, management strategies dependent upon this tactic must be reviewed frequently.

Several organic and inorganic compounds have adverse effects on foliage consumer feeding and biology. Inorganic nutrients applied as foliar sprays reduce foliage feeding damage (Leuck and Hammons, 1974b). *Spodoptera frugiperda* larvae fed peanut leaves treated with sodium chloride (1000 ppm), magnesium oxide (10 ppm) and iron chelate (10 ppm) had retarded weight gains, increased mortality and increased generation time (Leuck and Hammons, 1977). The organotin compound fentin hydroxide (0.5% spray) reduced feed-

ing and resulting damage of *S. subsecivella* on peanuts in India (Kareem and Subramaniam, 1978). Guazatine triacetate (GTA) also reduced insect foliage feeding damage in Alabama (Backman et al., 1977). Both latter antifeeding compounds also have good fungicidal activity against the major leafspot diseases.

Insecticidal applications should be made only in accordance with established economic thresholds. These thresholds usually are based on pest census data coupled with the phenology of plant development. Linker (1980) derived reliable sampling methodologies for several lepidopterous larvae on peanuts and provided calibration constants for translating relative density estimates (e.g., counts from sweep net) into absolute density estimates (e.g., larvae per square meter). Reliable sampling programs for obtaining pest density estimates are requisite for economic use of insecticides in an IPM program. Actual threshold levels and application procedures for specific pests are developed on a regional basis and available through local government offices.

In most instances, economic thresholds are not available, and insecticide use is based either on observable injury or total prophylaxis. Insecticidal prophylaxis is not congruent with the IPM philosophy, as the widespread use of this tactic has resulted in pest resistance, resurgence of nontarget species and changes in pest status. An excellent example of resurgence of nontarget species forcing modification of insecticidal prophylaxis on a foliage consumer is summarized from Teich (1969) for *S. littoralis* on peanuts in Israel. Control for *S. littoralis* consisted of preventive insecticide applications made after each irrigation, resulting in 8-9 applications per season. Reliance on this practice virtually exterminated the natural enemy fauna, caused an upsurge of spider mites and selected resistant *S. littoralis*. Plant defoliation experiments with emphasis on plant phenology, coupled with existing seasonal *S. littoralis* population curves, produced a new approach to insecticidal control based on IPM philosophy. Teich (1969) suggested the growing season of the peanut be divided into 3 phenological periods: (1) vegetative growth, (2) bloom and gynophore formation, and (3) fruit formation. Insecticide application thresholds were set at 20 caterpillars (larger than 10 mm each) per 2-meters row for the vegetative growth period. The destruction of 30 gynophores per 2-meters row during bloom and gynophore formation was considered damaging. Fifty caterpillars (larger than 10 mm) per 2-meters row for the fruit formation period was the third threshold based on plant phenology. Caterpillar censusing should be made twice weekly with population increases calling for action and decreases for relaxation.

**Resistant Cultivars.** Although no specific resistant peanut cultivars have been developed for management of foliage consumers, there is some evidence for sources of resistant germplasm. In the United States, *H. zea*, *S. frugiperda* and *A. gemmatalis* were the common species observed damaging 14 advanced peanut lines in Georgia. The Spanish lines received more damage than either the runner or virginia lines (Leuck et al., 1967). The most preferred cultivar was Starr (Spanish) while Virginia Bunch 67, Florigiant and Southeastern Runner 56-15 were non-preferred. The cultivars NC 6 and Early Bunch possess low to moderate resistance to *H. zea* with antibiosis reported as the resistance mechanism for NC 6 (Campbell and Wynne, 1980).

Laboratory studies have compared *S. frugiperda* biology when fed a non-preferred (cv. Southeastern Runner 56-15) versus a preferred cultivar (cv. Starr).

Southeastern Runner 56-15 increased the mean length of life cycle (egg to egg) an average of 4.3 days and increased larval and pupal mortality, resulting in 12% less adult moth emergence (Leuck and Skinner, 1971). Plant Introduction 196613 was more resistant than Southeastern Runner 56-15, giving 13% fewer *S. frugiperda* moths due to increased larval and pupal mortality (Leuck and Skinner, 1971).

From 98 bunch-type collections evaluated for possible resistance to *S. subsecivella*, 12 cultivars showed some levels of resistance with 16.4 - 20.7% of the leaflets infested (Rao and Sindagi, 1974). Ten cultivars evaluated for *S. subsecivella* revealed that 2 bunch cultivars averaged 12.3% infested leaflets while damage to spreading and semi-spreading varieties ranged from 7.0 - 8.7% (Lewin et al., 1971). These same authors found no evidence for resistance to *A. albistriga*.

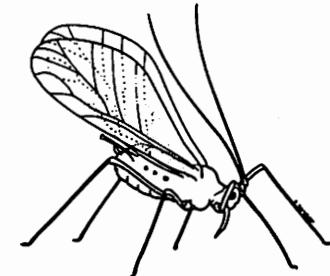
### Intracellular Feeders

These arthropod pests damage peanuts directly by removing cellular contents or indirectly by injecting toxic secretions and vectoring numerous plant pathogens. All types of damage can result in significant yield losses depending on the particular pest species, plant growth stage attacked and local physical environment. Complications exist in deciphering precisely the pest status of many of these organisms, as foliage is not consumed (i.e., holes do not appear in leaves); and quantitative relationships between pest density and probability of transmission of plant pathogens or direct damage to the plant usually have not been derived.

This group is quite large and will be represented by aphids, thrips and spider mites. Sufficient biological knowledge exists on select species of the 3 pest groupings to explore the role of biology and ecology in designing management strategies. Comparisons can be made to foliage consumers and soil inhabitants along at least 3 lines: (1) level of knowledge existing on biology, natural history and dynamics; (2) control tactics used against specific species; and (3) potential, but undeveloped, management strategies which may be better developed in the near future. Such comparisons will be of utility in charting a course for IPM of intracellular feeders worldwide.

**Aphids.** The importance of aphids attacking peanuts is related mainly to a role as vectors of numerous viruses (Table 3). Although the direct feeding of aphids can cause leaf chlorosis and deformation, much lower population densities can create complete devastation of a crop when a large proportion of the immigrant alates (winged form) is capable of virus transmission.

The distribution of aphid-transmitted viruses of peanuts is limited by both the geographical distribution of the aphid vector and the pathogen. For example, *Aphis craccivora* Koch is cosmopolitan in distribution while Gibbons (1977) considers groundnut rosette virus (GRV) to be restricted to Africa,



south of the Sahara, although it has been reported from India, Australia and Indonesia.

Ten aphid species are reported from peanuts (Table 1, Table 3). The role each species plays in virus transmission in peanuts is not always known. *Aphis craccivora*, important in the transmission of several plant viruses, is a key pest of peanuts in several geographical areas and illustrates management primarily based on pest ecology; thus, this section will concentrate on that particular aphid.

Table 3. Peanut viruses and arthropod vectors.

Disease	Vector(s)	Reference(s)
Peanut Mottle	<i>Aphis craccivora</i> Koch <i>Myzus persicae</i> Sulzee	Herold & Munz 1969, Bock 1973, Kuhn & Demski 1975, Paquier & Kuhn 1976
Peanut Stunt	<i>Aphis gossypii</i> Gowers <i>Amphorophora lactucae</i> (L.) <i>Rhopalosiphum padi</i> (Fitch) <i>Aphis craccivora</i> <i>Myzus persicae</i> <i>Aphis cricitola</i> (= <i>spiraecola</i> ) <i>Frankliniella fusca</i> <i>Scirtothrips dorsalis</i> <i>Thrips tobaci</i>	Behncken 1970 Behncken 1970 Behncken 1970 Tolin et al. 1970, Herbert 1967 Herbert 1967, Feakin 1973 Porter et al. 1975 Amin et al. 1978 Bald & Samuel 1931, Ghanekar et al. 1979 Bald & Samuel 1931 Frankliniella schulzei Frankliniella fusca Frankliniella occidentalis
Tomato Spotted Wilt	<i>Aphis craccivora</i> <i>Frankliniella fusca</i> <i>Scirtothrips dorsalis</i> <i>Thrips tobaci</i>	Davies 1972 Bald & Samuel 1931 Bald & Samuel 1931
Groundnut Rosette	<i>Aphis craccivora</i> <i>Aphis gossypii</i> <i>Aphis gossypii</i> <i>Myzus persicae</i> <i>Aphis craccivora</i> <i>Austrogallia torrida</i> Evans (a leafhopper)	Adams 1967 Reddy 1980 Reddy 1980 Dubern & Dollet 1978 Grylls 1954
Peanut Green Mosaic		
Groundnut Eyespot		
Rugose Leaf Curl		

**Biology.** Most aphids overwinter in the egg stage which gives rise to an immature female in the spring. The immature female molts several times and develops into an adult, winged, parthenogenetic, ovoviparous female known as the fundatrix (Eastop, 1977). The fundatrix may give birth to several hundred females which also are parthenogenetic and may be either alate (winged) or apterous. The progeny of the fundatrix are the spring migrants (alates) which disperse from the original host plant. In most aphids, a winged generation is followed by several wingless generations. Overcrowding or unsatisfactory condition of the host plant may cause production of winged forms. Shorter daylengths in the autumn induce the production of sexuparae which, in turn, produce a single generation of sexuals (males and sexually reproducing females). After mating, the female lays eggs which overwinter (Eastop, 1977). Variation in polymorphism is common in aphids and results from external stimuli (temperature, host plant condition, photoperiod, crowding, etc.). These stimuli dictate aphid hormonal balance which, in turn, regulates polymorphism.

In Africa, where *A. craccivora* is important as the vector of GRV (Storey and

Bottomley, 1928; Evans, 1954; Kousalya et al., 1971), no sexual forms have been reported (Real, 1955). *Aphis craccivora* overwinters on numerous uncultivated host plants (mainly the Leguminosae) with the alate migrants from these sources invading cultivated peanuts (Evans, 1954; Booker, 1963; Adams, 1967; Davies, 1972). Local overwintering and alternate hosts of *A. craccivora*, however, are not sources of GRV virus (Adams, 1967; Davies, 1972; Gibbons, 1977). This does not preclude the possibility that wild, uncultivated hosts of the virus do exist (Rossel, 1977).

Pastures planted with *Stylosanthes* could be perennial reservoirs of GRV (Okusanya and Watson, 1966; Gibbons, 1977). Dry season maintenance of GRV could be volunteer peanuts in East Africa but not in Sudan, Nigeria and Malawi (Gibbons, 1977). Viruliferous alates from outside the field provide the initial inoculum of GRV and are responsible for primary infection (or primary spread) of GRV within the field. These immigrant, alate aphids are transported on weather fronts (Davies, 1972).

A higher incidence of primary GRV infection is found in fields with wide spacings between plants as opposed to closely spaced plantings (Hull, 1964). Hull (1964) concluded that immigrant alates were more attracted to sparsely spaced plants due to greater exposure and proliferation of apical buds and young leaves. Young and senescent leaves of sparse plantings increase the exposure and incidence of the color yellow which in turn increases the alighting response of *A. craccivora* alates. The plant parts appearing yellow are fewer and camouflaged by green mature leaves in close plantings. A'Brook (1968) expanded this working hypothesis to include an additional increased alighting response from the contrast of plants and soil, the optomotor response, as well as color attractiveness for sparsely spaced plants. Thus, high density plantings are not attractive to flying aphids searching for a host plant, as they do not receive the correct visual cues (yellow color and contrast between bare earth and plants).

Secondary spread of GRV is attributed to both alate and apterous aphid forms produced from initial colonization within the field (Storey and Ryland, 1955). Widely spaced plantings result in higher aphid populations (Farrell, 1976b) which in turn enhances the very sensitive crowding stimulus for alate production in *A. craccivora* (Johnson, 1965) with 50% alates produced in populations of ca. 200 aphids/m<sup>2</sup> (Farrell, 1976b). Crowding could increase secondary spread through the increased production of alates which emigrate to reduce the population density. However, apterous forms infected 4 times as many plants adjacent to the GRV inoculum source as alates (Storey and Ryland, 1955). Gibbons (1977) reported alates in Nigeria and apterae in Uganda and Malawi most responsible for secondary spread of GRV.

The rate of increase of *A. craccivora* is lower in closely spaced peanut plants (Farrell, 1976b). This phenomenon has been attributed to a lower level of nutrition in the densely planted fields (Real, 1955; Farrell, 1976b). Waghra and Singh (1965) showed decreased fecundity in *A. craccivora* with low levels of nitrogen. Therefore, close plantings not only reduce primary GRV spread, but also lessen secondary spread by reducing aphid buildup and decreasing emigration.

The GRV is transmitted in a persistent manner by *A. craccivora* and requires an acquisition period of ca. 2-3 days (Watson and Okusanya, 1967). Once *A.*

*craccivora* acquires GRV, the virus can remain viable for at least 10 days (Storey and Ryland, 1955). Both immature and adult stages can vector GRV (Storey and Ryland, 1955).

**Damage.** *Aphis craccivora* is considered a key pest in Africa (Hill, 1975) and certain areas of India (Rai, 1976). In Africa the economic importance is related to transmission of GRV, whereas in India direct feeding damage as well as disease transmission is serious.

In India, aphids feed on the succulent vegetative tips prior to the initiation of flowering causing leaf curl and stunted growth. Later they migrate to the floral shoots, seriously reducing pod formation (Rangaswany and Rao, 1964). Yield reductions of 40% are reported (Khan and Husain, 1965) from *A. craccivora* feeding damage alone.

Kousalya et al. (1967) described primary and secondary spread of a rosette disease of groundnuts vectored by *A. craccivora* in India. When the crop was 15 days old, 39% of the plants were infested with *A. craccivora*. By 60 days, 88% of the plants were infested. Maximum disease incidence (0.25% with symptoms) was manifested when the plants were 75 days old. The proportion of apterae was 0.72 at 15 days and 0.94 at 90 days after planting with a positive correlation between % plants infested with aphids and disease incidence.

In Africa, *A. craccivora* is important as the vector of GRV (Hill, 1975). Groundnut rosette virus is manifested by 2 main types of symptoms: chlorotic and green rosette (Gibbons, 1977; Mercer, 1977). Both cause stunting of the plant with severity of the disease related to the earliness of infection. Plants infected early by GRV produce few, if any, pods resulting in virtual crop failure, whereas aphid control in later infections may increase yield by 50% (Eastop, 1977). Crop loss is related to the incidence of the disease (severity) and the phenological age of the plant when infection occurs (temporal tolerance). The later the infection (or the closer to harvest), the less severe the crop loss.

**Management. Natural Enemies.** Numerous general predators are reported to attack *A. craccivora* in India (Patel et al., 1976; Rai, 1976) and USSR (Kesten, 1975), mostly from the families Coccinellidae (Coleoptera) and Syrphidae (Diptera). The aphid parasite, *Lysiphlebus testaceipes* (Cresson), was introduced into India from the United States (1966-67) for suppression of *A. craccivora* (Ramaseshiah et al., 1968). The parasite reproduced readily during the cooler seasons but was successful at temperatures above 32°C only when *A. craccivora* was hosting on *Dolichos lab-lab*. A related aphid species, *L. fabarum* (Marshall), parasitizes 85% of the *A. craccivora* in June in the USSR (Kesten, 1975). For general information on the impact of natural enemies on aphids, see Hagen and van den Bosch (1968).

**Insecticides.** Sprays, granules and dusts are effective against *A. craccivora* in India (Sarup et al., 1960; Vasantharaj et al., 1965; Dorge et al., 1966; Gangadharan et al., 1972) and the United States (Smith and Culp, 1968). Davies (1975) reviewed the current status of insecticides in Africa for *A. craccivora* control and reported on large scale trials with manazon and conventional trials with numerous other compounds.

**Resistant Cultivars.** Brar and Sandhu (1975) evaluated 50 cultivars (19 bunch, 16 spreading and 15 semi-spreading) for resistance to *A. craccivora* in India. Data on differences in aphid reproduction were significant with the bunch cultivars having lower aphid reproduction than spreading or semi-

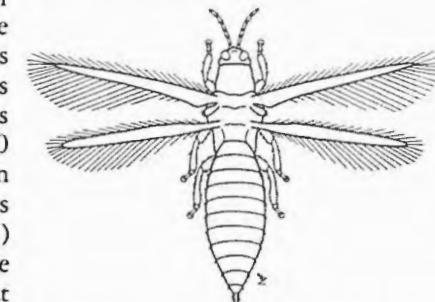
spreading cultivars. Several cultivars showed promise as providing resistant germplasm. Amin and Mohammad (1980) have also shown greatly reduced progeny production by *A. craccivora* when isolated on detached shoots of cultivars of *Arachis chacoense* and *A. hypogaea*. High levels of resistance to GRV have been reported from a group of cultivars from the Ivory Coast and Upper Volta areas of West Africa (Gibbons and Mercer, 1972; Rossel, 1977).

**Cultural.** The major management tactics available for use against *A. craccivora* and transmission of GRV involve sanitation, early planting and uniform, dense plantings of peanuts within a field. Sanitation includes removal of volunteer peanut plants to reduce the GRV inoculum (Gibbons, 1977) and removal of weed hosts (Kousalya et al., 1971). Early planting allows the plant to partially develop either prior to the arrival of viruliferous alates or prior to the buildup of aphids locally (temporal tolerance) (Booker, 1963; Farrell, 1976a). This approach also allows the plant to reach a more mature, unattractive stage and obtain maximum groundcover which tends to reduce primary spread of GRV. Plants 20-30 days old may be infested with 3-5 times as many alate aphids as plants 50-60 days old (Feakin, 1973). Winged aphids invade the crop in large numbers beginning 50 days after 2.54 cm of rain has fallen in the growing season (Feakin, 1973).

A uniform, close spaced planting drastically reduced the alate *A. craccivora* alighting in the field (A'Brook, 1964; Farrell, 1976c). Close spacing also reduces the rate of increase of the colonizing aphids (Farrell, 1976b). Early planting of uniform, closely spaced plants reduces the damage from GRV if the plant is infested, reduces primary spread by deterring aphids from alighting and further reduces secondary spread by reducing aphid reproduction. Densities of 197,600 plants/ha for the late crop and 98,800 plants/ha for the early crop are satisfactory in minimizing GRV infection (Feakin, 1973).

**Thrips.** Seventeen species of thrips (Thysanoptera: Thripidae) have been reported feeding on peanuts worldwide (Table 1). The economics of thrips control with insecticides has been a controversial issue for over 30 years and still remains unresolved on a worldwide basis. The role of thrips as vectors of peanut diseases (Table 3) will receive more attention in the next decade, but such information, at present, is scarce.

**Biology.** Thrips found on peanuts are minute (0.5-2.0 mm long), inconspicuous insects. Adult females oviposit in the plant tissue and all life stages (egg, larva, pupa and adult) are found on the host plant. Hatching gives rise to 2 successive larval stages which feed on the plant tissue, followed by 2 pupal stages which are active but do not feed. All immature stages are wingless but resemble adults otherwise. The metamorphosis of thrips is typical of the paurometabolous insects whose juvenile stages are called nymphs. The terms larva and pupa usually are used to describe the juveniles of the insects having complete metamorphosis; however, the terms larva and pupa are used here to avoid confusion with the thrips literature (Lewis, 1973).



Developmental time from egg to adult varies with temperature and species. Adult tobacco thrips, *Frankliniella fusca* (Hinds), live ca. 30 days and lay 50-60 eggs (Watts, 1934), which require ca. 16 days to hatch during summer (Watts, 1936). Numerous generations occur on cultivated and wild hosts throughout the warm seasons, while in temperature zones thrips generally enter diapause or quiescence during cold climatic conditions as fully grown larvae or adults (Lewis, 1973). In warmer zones, there is a period of inactivity during cool periods (Watson, 1918), but breeding is essentially continuous.

Thrips initially move into fields from wild or cultivated hosts. This movement is most pronounced in the spring when peanuts are in the seedling stage. Peanuts planted downwind of small grain fields readily obtain high densities of thrips (Smith and Sams, 1977). Thrips emigration from the maturing grain crop in the late spring coincides with the planting time for peanuts.

Migrant adults oviposit on peanuts and successive generations occur on the plant throughout the growing season (Tappan and Gorbet, 1979). Populations are highest in the terminal buds during the first 30 days after planting (Smith and Sams, 1977; Tappan and Gorbet, 1979) and decline to a lesser density for the remainder of the season. Phenologically, the plant begins to flower at ca. 30 days after emergence, which coincides with the thrips population decline in the terminal buds (Sams and Smith, 1978; Tappan and Gorbet, 1979). Hammons and Leuck (1966) reported immature thrips to be predominant in flowers and postulated thrips changed microhabitats and food source, moving from the terminal buds to flowers with anthesis. Comprehensive data from Tappan and Gorbet (1979) discounted movement of immatures to flowers, since 90% of the thrips population in terminal buds were immatures throughout the season. Correspondingly, 92% of the population in the flowers were adults. The decline of thrips density in terminal buds at the onset of flowering could result from a dilution of the population from a concentrated few terminal buds early in the season to the numerous flowers and terminal buds produced later in the season as the plants grow.

Thrips found on peanuts are not all phytophagous; some are predaceous and mycophagous. The most commonly encountered predaceous thrips is *Scolothrips sexmaculatus* (Pergande), which preys on several species of spider mites. *Scolothrips sexmaculatus* rarely is abundant on peanuts, although it is quite commonly found on mite infested plants (observation of senior author). The bionomics and predaceous habits of *S. sexmaculatus* are reviewed by Gilstrap and Oatman (1976). *Euphysothrips minozii* Bagnall is a mycophagous species that feeds on spores of peanut rust, *Puccinia arachidis* Spegazzini, in India (Shanmugam et al., 1975).

**Damage.** Adult and larval thrips rasp the leaf tissue and extract the plant juices, preferring the unfolded, developing leaflets in the terminal buds. Feeding is manifested as scarring, chlorosis and deformation of the leaflets. Damage is visually apparent as the leaves unfold. The plant is considered most susceptible to feeding injury by *F. fusca* in the United States from emergence to 30 days old and by *Enneothrips flavens* Moulton in Brazil up to 60 days old (Batista et al., 1973). Severity of damage can range from minimum chlorosis and scarring to leaflet abscission. Approximately 1 thrips per bud can result in injury to 33-80% of the leaflets (Tappan and Gorbet, 1979). The number of damaged leaflets declines within 1-2 weeks after the onset of flowering, lagging behind the

thrips population decline in the buds (Leuck et al., 1967; Tappan and Gorbet, 1979). This lag represents the time delay between the injury occurring in the unfolded leaflets and its appearance on the exposed leaf surface.

Information on the role of the thrips as vectors of peanut pathogens is sparse (Table 3). *Frankliniella fusca* has been implicated as a vector of peanut stunt virus in Virginia (Porter et al., 1975). Several thrips species, including *Thrips tabaci* Lindeman, *Frankliniella schulzei* (Trybom), *F. fusca*, *Scirtothrips dorsalis* Hood and *F. occidentalis* (Pergande) vector tomato spotted wilt virus (Bald and Samuel, 1931; Ghanekar et al., 1979; Amin and Mohammed, 1980; Ananthakrishnan, 1980). Thrips are unusual in comparison to most virus vectors because the larvae must feed on infected plants before either the larva or adult can transmit a virus (Bald and Samuel, 1931; Lewis, 1973).

Detrimental effects of direct thrips feeding on plant growth, anthesis, pod yield and seed quality have been a subject of controversy for several decades. The initial reports by Poos (1945) and Poos et al. (1947) provided the genesis for the thrips control controversy by reporting thrips reduced peanut yields up to 37%. Successive reports by numerous other United States authors (Arant, 1954; Arthur and Arant, 1954; Howe and Miller, 1954; Dogger, 1956; Arthur and Hyche, 1959; Harding, 1959; King et al., 1961; Morgan et al., 1970; Smith, 1971b, 1972a; Minton and Morgan, 1974; Sams and Smith, 1978; Smith and Sams, 1977; Tappan and Gorbet, 1979, 1981) failed to identify yield increases by controlling thrips with insecticides, even with thrips populations as high as 50 per terminal bud (Smith and Sams, 1977) or 92% of the leaflets damaged (Minton and Morgan, 1974). Thrips damage also has not been correlated with seed maturity (Sams and Smith, 1978), rate of flowering or plant growth (Morgan et al., 1970). Close examination of the reports by Poos (1945) and Poos et al. (1947) reveal multiple insecticide applications were used over a long period of plant growth which could have suppressed a nontarget pest species and resulted in spurious conclusions regarding the benefits of thrips control. Existing data clearly show thrips control in the United States is not a sound economic investment (Bass and Arant, 1973; Smith and Sams, 1977; Tappan and Gorbet, 1979).

*Enneothrips flavens* is considered a key peanut pest in Brazil. Control of this thrips on peanuts up to 60 days after plant emergence resulted in absolute yield increases of 790 kg/ha (Almeida et al., 1977) and proportional increases of 39% (Calcagnolo et al., 1974), 50% (Almeida et al., 1965) and 35% (Lara et al., 1975). In several instances, pest species other than thrips were present in these evaluations, but their contribution to yield loss was not readily determined.

In central Africa (Cameroon and Sudan), several species of thrips, *Taeniothrips sjostedti* (Trybom); *Haplothrips gallarum* Priesner; *Sericothrips occipitalis* Hood; *Caliothrips* sp.; *C. sudanensis* (Bagnall and Cameron); and *C. fumipennis* (Bagnall and Cameron), attack the unfolded leaf resulting in chlorosis and deformation on the leaflets (Clinton, 1962; Nonveiller, 1973). The economic importance of thrips on African peanuts is questionable (Hill, 1975).

*Scirtothrips dorsalis* Hood and *Caliothrips indicus* (Bagnall) heavily damage peanut plants in India (Rai, 1976; Sapathy et al., 1977), causing leaf chlorosis and abscission. Control of *S. dorsalis* and *Empoasca* sp. resulted in a yield increase of 1536 kg/ha (Saboo and Puri, 1978).

**Management. Natural Enemies.** Thrips are attacked by both parasites

and predators. General arthropod predators include anthocorid bugs in the genus *Orius* which are cosmopolitan enemies; *Geocoris* sp. (Lygaeidae), *Chrysopa* sp. (Chrysopidae), *Hemerobius* sp. (Hemerobiidae), in the United States; *Cheilomenes vicina* Mulsant (Coccinellidae), *Ischiodon aegypticus* Wied (Syrphidae) in the Sudan, and *Psallus* sp. (Miridae) in India (Lewis, 1973). Insects parasitic on thrips include the families Trichogrammatidae, Scelionidae and Mymaridae which parasitize thrips eggs, and Eulophidae which attack larvae (Lewis, 1973).

**Insecticides.** Thrips are controlled easily with most modern insecticides. Granular systemic, spray and dust formulations give highly effective control (Arthur and Hyche, 1959; Castro et al., 1972; Smith, 1972a; Bass and Arant, 1973; Nonveiller, 1973; Minton and Morgan, 1974; Almeida et al., 1977; Mateus and Gravena, 1977; Rensi et al., 1977; Saboo and Puri, 1978; Sams and Smith, 1978; Rohlfs and Bass, 1980). Campbell et al. (1976), however, have shown insecticidal performance is not independent of peanut variety. Their research produced results which allude to unquantified interactions between soil applied systemic insecticides and cultivars with different growth habits (bunch, runner and intermediate growth types). Insecticides that gave 90% thrips control on one cultivar (or growth type) gave ca. 50% control on another.

**Resistant Cultivars.** Thrips resistant peanut cultivars have been identified in the United States (reviewed by Smith, 1980) and in India (Panchabhai and Thimmaiah, 1973). Resistance to *F. fusca* in the virginia, spanish and valencia type peanuts has been verified by both laboratory and field evaluations. Collectively, the spanish types have the most resistant germplasm. The spanish cultivars Starr and Argentine are immediate sources of agronomically suitable resistant germplasm (Leuck et al., 1967; Young et al., 1972; Kinzer et al., 1973). Plant Introduction 280688, a valencia type cultivar, is non-preferred for larval and adult feeding, has low larval survival (antibiosis) and low levels of oviposition (Kinzer et al., 1972; Kinzer et al., 1973). Cultivars NC 6, Virginia Bunch 67 and PI 290599 are sources of resistant germplasm for virginia type peanuts (Leuck et al., 1967; Young et al., 1972; Campbell et al., 1977; Campbell and Wynne, 1980). Resistance to *Calothrips indicus* (Bagnall) was identified in several cultivars: 21008, 21016, HG-10, 21018 and EC-20979 (Panchabhai and Thimmaiah, 1973). Cultivars 21008 and 21016 were considered the most resistant, with ca. 23% of the leaves showing no damage as compared to 1.39% for the cultivar commonly under cultivation. Fecundity of *F. schultzei* (expressed as eggs/female/24 hours) was greatly reduced on *Arachis chacoense* (0.0), *A. glabrata* (0.0) and *A. duranensis* (0.4) compared to *A. hypogaea* (4.4) (Amin and Mohammad, 1980).

**Cultural.** Several cultural practices are conducive to thrips management. Sanitation of early volunteer seedlings in the spring is important in preventing infiield population increases prior to planting the crop (Bass and Arant, 1973). Peanut fields planted adjacent to winter small grains, especially when positioned downwind, are available for migrant adults when the grain begins to mature (Smith and Sams, 1977).

In India, early planted peanuts usually escape heavy losses from thrips-born bud necrosis disease caused by tomato spotted wilt virus. Infection levels are lowest in peanuts planted at least 6 weeks prior to peak thrips (*F. schultzei*) im-

migration into peanut fields which occurs in August and January. Bud necrosis incidence is related to immigrant thrips (i.e., secondary spread is unimportant). High plant densities result in a lower percentage of infested plants. Early planting dates and high plant densities coupled with properly timed insecticide applications and using less susceptible cultivars are effective in managing bud necrosis disease in India (Amin and Mohammad, 1980).

**Spider Mites.** Several species of soil and foliage inhabiting acarines are associated with the peanut plant (Table 1). The soil inhabiting astigmatids have been implicated in the spread and increase of soil borne fungal diseases of peanut pods (Aucamp, 1969; Shew and Beute, 1979), whereas foliage feeding by the tetranychids causes leaf chlorosis and defoliation. The soil inhabiting astigmatids are presented in this section rather than the soil pest section to maintain biological continuity of pest groups.

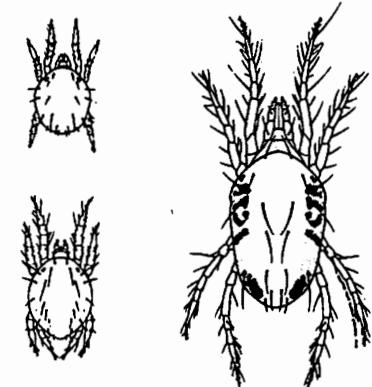
**Biology.** Tetranychid mites develop through the metamorphic stages of egg, larva, protonymph, deutonymph and adult (van de Vrie et al., 1972). The larval stage has 6 legs, and the other stages have 8 legs. Variations in the developmental stages may occur among mite families. For example, the eupodid, *Penthaleus major* (Duges), has a deutovum (prelarval) stage. Length of the life cycle is correlated with temperature, humidity and host plant quality (Watson, 1964; van de Vrie et al., 1972; Jeppson et al., 1975).

Copulation of adults occurs immediately after hatching of the female with diploid eggs giving rise to female progeny and haploid eggs yielding male progeny (van de Vrie et al., 1972). *Tetranychus urticae* (Koch) females produce from 42-204 eggs per female depending upon host plant (van de Vrie et al., 1972) and moisture (Boudreaux, 1958).

The total life cycle for *T. urticae* females is 8-12 days at 30-32 C. Females live ca. 30 days and lay 90-110 eggs (Jeppson et al., 1975). Thus spider mites have a tremendous capacity for increase and can rapidly produce enormous population densities if conditions are optimal.

The dormant stage which passes through adverse environmental conditions is the mated, diapausing female which does not feed until the adverse conditions ease (van de Vrie et al., 1972). Factors inducing diapause include photoperiod, temperature and nutrition (Parr and Hussey, 1966). Tetranychid mites are less host specific than other mite families, as most species have a relatively narrow host range. *Tetranychus cinnabarinus* (Boisduval), *T. urticae* and *T. turkestanii* (Ugarov and Nikolski) have a wide host range including many plant genera (Jeppson et al., 1975).

The astigmatid biologies are poorly known with the exception of species infesting stored grains (Hughes, 1961). *Caloglyphus* life cycle requires 8-9 days at 22 C with adequate moisture with females laying ca. 200 eggs in 24 hours (Hughes, 1961). The biology of *P. major*, Eupodidae, is summarized by Jeppson et al. (1975) and Smith (1946).



**Damage.** Tetranychid mites feed by penetrating the plant tissues and removing the cell contents. Feeding causes the chloroplast to disappear with the small amount of remaining cellular material coagulating to form an amber mass (Jeppson et al., 1975). Continued feeding results in the formation of irregular chlorotic spots on the leaf tissue typical of spider mite damage. Injection of toxins or growth regulators by spider mites was questioned by Jeppson et al. (1975), although the reactions of specific plant species to feeding by the same spider mite species may drastically differ. The severity of plant damage resulting from tetranychid feeding is related to crop species, mite density, local environmental conditions, plant nutrition, moisture stress and phenological growth stage (Watson, 1964; van de Vrie et al., 1972). Spider mites on peanuts are especially destructive during hot, dry weather (Campbell et al., 1974; Osman and Abdel-Fattah, 1975). Significant mite densities may kill large areas of plants (Smith and Jackson, 1975), resulting in considerable yield loss.

The astigmatid genera, *Tyrophagus* and *Caloglyphus*, were isolated frequently from the subterranean parts of the peanut plants in South Africa (Aucamp, 1969). *Caloglyphus micheali* and other *Caloglyphus* species were isolated from field soil, decaying pods and the root and pod zone of healthy plants in the United States (Shew and Beute, 1979). *Caloglyphus* are capable of acting as a disseminating agent for the *Aspergillus flavus* (Link) and *Pythium myriotylum* Drechsler fungi by internal contamination of gut contents with viable spores in defecated fecal pellets (Aucamp, 1969; Shew and Beute, 1979). *Caloglyphus* spp. in South Africa were reported as feeding on the peanut pods and seed, as well as on fungi (Aucamp, 1969); whereas *C. micheali* was reported as totally mycophagous (Shew and Beute, 1979), acting wholly as a fungal disseminating and not a wounding agent. These mites can spread fungi, but for infections to occur, entrance to the seed must be gained. *Caloglyphus* cannot penetrate intact pods and environmental conditions must be in accordance with the ecological requirements of the fungi for germination and fungus growth (Aucamp, 1969). Soil applications of acaricides significantly reduced both peanut pod rot, caused by *P. myriotylum*, and mite populations in both field and greenhouse studies (Shew and Beute, 1979). This report enhances the implication of the role of certain astigmatid mites as vectors of soil fungal pathogens. Other soil pests, *Meloidogyne arenaria* (Neal) and *Diabrotica undecimpunctata howardi* Barber, also have been reported to disseminate and enhance *P. myriotylum* infection of peanut pods (Porter and Smith, 1974; Garcia and Mitchell, 1975).

**Management. Natural Enemies.** Spider mites are attacked by fungi, predaceous mites and insects (Huffaker et al., 1970; McMurtry et al., 1970). The fungi, *Entomophthora* spp. and *E. fesenii* Nowakowski, have been reported infecting *T. urticae* and *T. cinnebarinus* (Carner and Canerday, 1968) with *Entomophthora* spp. mycosis being most effective in hot, humid weather (Campbell, 1978). The destruction of *Entomophthora* spp. fungi by fungicides applied for control of plant pathogenic fungi (leafspots, rusts, etc.) on peanuts may be a major factor in spider mite outbreaks (Campbell, 1978).

Predaceous phytoseiid mites are of major importance in suppression of tetranychids of many crops (McMurtry et al., 1970). Although predaceous phytoseiids have not been reported from peanuts, ca. 10 species have been reported attacking *T. urticae* and/or *T. cinnebarinus* on various other crops in the United States, Egypt, Japan and Canada (McMurtry et al., 1970). Several insects also

are considered important predators of spider mites. The coccinellid genus, *Stethorus*, is restricted to mite predation and has been found in peanut fields in Texas (observation of senior author). Other insect predators and their impact on spider mites are reviewed by Huffaker et al. (1970), McMurtry et al. (1970) and van de Vrie et al. (1972).

**Insecticides.** Spray and granular formulations of insecticides and acaricides effective against *Tetranychus* on peanuts have been reported from Egypt (Attiah and Rizk, 1973; Osman and Rasmy, 1976), India (Gupta et al., 1969), Pakistan (Moiz and Qureshi, 1969), Bulgaria (Atanasov, 1971) and the United States (Campbell et al., 1974; Smith, 1976a; Smith and Mozingo, 1976, 1977).

A preponderance of evidence exists to implicate pesticides (insecticides, acaricides and fungicides) as agents inducing spider mite outbreaks (Teich, 1969; van de Vrie et al., 1972; Smith and Jackson, 1975; Campbell, 1978). The change in spider mite pest status on peanuts in the United States (from nonpest to secondary pest) was discussed earlier under Pest Status. This change in pest status has not been restricted regionally to the United States (Smith and Jackson, 1975; Campbell, 1978) but geographically encompasses worldwide peanut production as evidenced by reports from Pakistan (Moiz and Qureshi, 1969), India (Gupta and Sandhu, 1969; Gupta et al., 1969), Israel (Teich, 1969) and Bulgaria (Atanasov, 1971). The application of certain insecticides and fungicides in combination further exacerbates the spider mite outbreak phenomenon. Campbell (1978) reported multiple applications of carbaryl + benomyl to peanuts resulted in a mite density increase of 344X over the density of the untreated control. The increased use and dependence upon agricultural chemicals as the sole management tactic for peanut pest suppression in many production areas should continue to exacerbate the spider mite problem.

Spider mite populations have a high propensity for developing resistance to insecticides and acaricides. Historically the genus *Tetranychus* has been capable of rapidly developing resistance to a wide variety of toxicants, when repeated toxicant applications provide selection pressure (Jeppson et al., 1975). The problem of a high incidence of pesticide resistance in mites is magnified further by cross-resistance to chemically related and to some unrelated compounds (Smith, 1960). Populations resistant to one organophosphorous (OP) insecticide often show resistance to other OP and carbamate compounds while remaining susceptible to organochlorine compounds (Jeppson et al., 1975). On peanuts, *T. urticae* and *T. cinnebarinus* have developed resistance to some OP compounds as a result of prophylactic OP use (Smith and Jackson, 1975).

**Resistant Cultivars.** Resistance and susceptibility to spider mites have been identified in both commercial cultivars and wild peanut species. In India, *T. urticae* infestations were higher on semi-spreading cultivars (e.g., Exotic 5, C501, Asirya Mwitunde and HG10) than bunch-type varieties (Gupta et al., 1969); thus, erect growing foliage may offer some protection. Campbell et al. (1974) reported the cultivars Va 72R, NC-Fla 14 and NC 17 had the lowest leaf damage while NC 2 had the highest leaf damage resulting from *T. urticae* feeding. Ranges in susceptibility were from 23.8 - 76.9% for the 11 cultivars evaluated in the greenhouse.

Germplasm possessing the highest levels of resistance to spider mites is within the wild species of *Arachis*. Most species in the section *Rhizomatosae*

are highly resistant as wild species PI 338296, 338317, 262840 and 262827 remained virtually free of *T. urticae* (Johnson et al., 1977). Mites feeding on PI 262286 and 262840 had reduced fecundity when compared to the commercial line NC 5 (Johnson et al., 1980). Adult females also exhibited a high degree of non-preference for feeding on both plant introductions. Only a few plant introductions from the sections *Arachis*, *Erectoides*, *Extranervosae*, and *Caulorizae* exhibited resistance (Johnson et al., 1977). Resistance to *T. tumidellus* Prichard and Baker has also been reported (Leuck and Hammons, 1968).

**Cultural.** The microclimate produced by interplanting between rows of citrus and the cultural measures associated with citrus production were presumed as the cause of an outbreak of *T. cinnebarinus* and *T. urticae* on peanuts in Egypt (Osman and Rasmy, 1976). Sprinkler irrigation was effective in suppressing *T. urticae* populations on peanuts in Egypt (Osman and Abdel-Fattah, 1975). Mite populations were 3 to 10X higher under overflow irrigation as compared to sprinkler irrigation. Osman and Abdel-Fattah (1975) suggested sprinkler irrigation washed the mites from the leaves. This observation is supported by research on mites infesting other crops (van de Vrie et al., 1972).

## SOIL INHABITING PESTS

Many soil inhabiting pests characteristically feed upon the fruit (pods) and/or fruit precursors. Secondary infections of fungi and other plant disease organisms may gain entry into the stems, roots and pods as a result of soil arthropod feeding and may further reduce pod production. Information on interactions between soil borne diseases and soil arthropod feeding are limited, but will be presented when available (also see section on spider mites; the soil inhabiting astigmatids). Plants exhibit temporal tolerance to both primary and secondary injury, but spatial tolerance is nil.

Soil inhabiting arthropods are represented in the orders Acarina, Julida, Orthoptera, Dermaptera, Isoptera, Hemiptera, Homoptera, Coleoptera, Lepidoptera, Diptera and Hymenoptera (Table 1). Of the 2 management groups (soil and foliage), soil pests probably represent the key pests of peanuts; however, the ecology, biology, damage and plant phenological relationships are the least understood. Special problems are inherent with soil arthropod research and management which may explain the absence of biological knowledge for understanding pest bionomics and formulating management strategies.

Soil arthropods occupy a cryptic habitat which restricts undisturbed, direct observation of pest life stages, associated behavior and damage inflicted upon the plant. Curbed biological observations hinder hypothesis formulation and data collecting. Data gathering must rely on labor intensive sampling techniques for censusing population density, age distribution and inflicted plant damage. Visual observations of damage to the roots and main stem are delayed temporally until the injury is manifested in vegetative (aerial) plant parts. Pod damage may go totally unnoticed, as secondary infections of soil microorganisms camouflage the arthropod damage at harvest or decompose the pod in the soil. These conditions tend either to mask the importance of soil arthropods or result in the arbitrary assignment of the damage of soil microorganisms.

Edaphic conditions are paramount to the population ecology of soil arthropods. Different pest species require certain soil types as well as optimal envir-

onmental conditions to express their biotic potential. The deep, sandy soils of Texas and Oklahoma share the same foliage consuming lepidopterous pest complex as the heavier soils of North Carolina but the soil pests are different. *Elasmopalpus lignosellus* (Zeller) is an annual, key pest in the southwestern United States (Berberet et al., 1979a; Smith and Holloway, 1979), whereas it occurs infrequently in North Carolina (W. V. Campbell, pers. comm.). *Pseudococcus* sp. (Smith, 1946), *Diabrotica* spp. (Fronk, 1950; Feakin, 1973) and *Dysmicoccus brevipes* (Cockerell) (Feakin, 1973) are pests of poorly drained soils. White grubs are pests of friable, volcanic, red soils in Queensland (Smith, 1946).

Management strategies for many soil pests must be developed on the individual species basis and cannot be classed collectively by plant injury as has been done with foliage inhabitants. The major reasons mandating species divisions in management may include: (1) most soil pests are key pests, thus they are the initial target of management; (2) a good quantitative description of the relationship between pest injury and peanut growth and fruiting is not available, (3) edaphic factors which provide the template for pest population dynamics are more mosaic than the environmental factors which drive foliage inhabitants, and (4) information on soil pest bionomics is limited, restricting the use of biological common denominators.

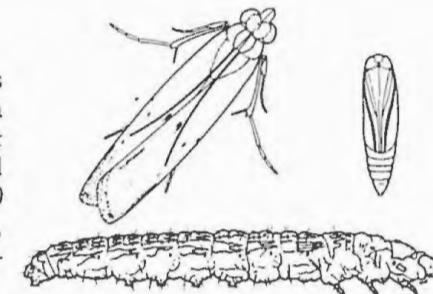
## Lepidoptera

*Elasmopalpus lignosellus* (Zeller) is the only lepidopteran considered a true pest as all life stages, except adult, are soil inhabiting. Several noctuids (e.g., *Agrotis* and *Feltia*) have diurnal soil inhabiting larvae, but most of the damage is from nocturnal foliage consumption.

Recently, *Stylopalpia costalimai* Almeida was reported attacking peanuts in Brazil (Almeida, 1960, 1961; Almeida and Pigatti, 1961; Bastos Cruz et al., 1962). Translation of these reports revealed *S. costalimai* feeds on the foliage and may spend part of the larval stage in or on the soil but is not considered a true soil pest. *Stylopalpia* sp. near *costalimai* has been reported as a key pest of peanuts in Paraguay (Unruh, 1981). This species feeds on pegs and pods and spins feeding webs in the soil which is similar to *E. lignosellus* feeding behavior.

*Elasmopalpus lignosellus* (Zeller). This pest is restricted in distribution to the New World where it attacks peanuts and numerous other legume crops as well as numerous grasses. An ecological equivalent of *E. lignosellus* from peanuts in the Old World has not been reported. The genus *Elasmopalpus* was considered monospecific by Heinrich (1956); however, more recently Gates Clarke (1965) removed *E. angustellus* Blanchard from Heinrich's (1956) synonymy. The larval habitat and host plants of the closely related genera, *Adelphia*, *Tota* and *Ufa* are unknown (Heinrich, 1956) except for *U. rubedinella* (Zeller) which attacks the pods of pigeon pea in the Lesser Antilles (Fennah, 1947).

**Biology.** Adult females oviposit 33-420 eggs (Luginbill and Ainslie, 1917; Walton et al., 1964; Leuck, 1966; Stone, 1968a; Razuri, 1975). Most eggs are laid singly in the soil (less than 2 mm deep) under the drip line of pea-



nut plants. Two % of the eggs may be deposited on the foliage (Smith et al., 1981). Eggs are white the first day, turn red by the second day and hatch the third day (Leuck, 1966). Hatch may require up to 5 days during the cooler fall periods (Luginbill and Ainslie, 1917).

The small, red, first instar larvae crawl across the soil from the oviposition site to the plant or to other edible organic matter. Larvae feed slightly below the soil surface and construct a silken tunnel covered with soil particles which is attached to the feeding site (Luginbill and Ainslie, 1917; King et al., 1961; Bastos Cruz et al., 1962; Leuck, 1966). Larvae feed readily on dead organic matter (Cheshire and All, 1979) as well as numerous plant species (Stone, 1968c). Larval development in the field has been found to require 13-24 days (Leuck, 1966), 11-39 days (King et al., 1961), 24-46 days (Dupree, 1965), or 14-42 days (Luginbill and Ainslie, 1917). Laboratory studies on larval and pupal development by Holloway and Smith (1976b) showed developmental rate dependent upon temperature, with larval-pupal development predicted by the following equation: developmental days =  $191.245 \text{ days} - 5.202 (\text{C temperature})$ . Berberet et al. (1979b) reported 530 degree days above 13 C to be required for development from egg deposition to adult emergence.

The number of larval instars is variable. Leuck (1966) and Dupree (1965) report 6 instars; Razuri (1975), 5-6 instars; and Luginbill and Ainslie (1917), 4-7 instars. The latter author stated that the number of instars increased with temperature. Larvae reared at constant temperatures on artificial diet in the senior author's laboratory have completed 5-9 instars, with 6 instars being most common.

Pupation occurs in the soil, usually at a greater depth than larval feeding (senior author's observation). The pupal chamber is constructed of material similar to the larval feeding tunnel but is of much stronger construction. Pupation in the field lasts ca. 10 days (Luginbill and Ainslie, 1917; Leuck, 1966).

The life cycle from oviposition to adult emergence spans 33-65 days under field conditions. The number of generations per year in the United States is variable with 4 in South Carolina (Luginbill and Ainslie, 1917) and Mississippi (Lyle, 1927), 3 in Arizona (Vorhies and Wehrle, 1946), 3 plus a partial fourth in Georgia (Leuck, 1966) and 3 distinct generations in Texas (Johnson, 1978) and Oklahoma (Berberet et al., 1979b).

Adults are nocturnal (Holloway and Smith, 1975, 1976a), mate the first day after emergence and begin ovipositing the second day (Stone, 1968b). Female moths release a sex pheromone from 0-96 hrs. post emergence which attracts males (Payne and Smith, 1975). Ganyard and Brady (1972) also reported *E. lignosellus* males attracted to virgin female *Plodia interpunctella* (Hubner), *Cadra cautella* (Walker) (both Pyralidae), *Spodoptera frugiperda* and *S. exigua* (both Noctuidae). The active compound for attractiveness was considered to be (Z,E)-9,12-tetradecadien-1-ol acetate. Field experiments by Mitchell et al. (1976) revealed male *E. lignosellus* are not attracted to this compound, but a field permeated with the compound may disrupt mating.

A considerable divergence in the overwintering stage may occur, depending upon climatic conditions. All reports agree that *E. lignosellus* overwinter as a mature larva and/or pupa (Luginbill and Ainslie, 1917; Lyle, 1927; Vorhies and Wehrle, 1946; King et al., 1961; Leuck, 1966). Holloway and Smith (1976b) showed all life stages unresponsive to changes in photoperiod with an

absence of the classical diapause evident in most temperate Lepidoptera with similar host ranges and geographical distributions. Cooler temperatures significantly increased larval and pupal developmental time; thus, these stages negotiate overwintering by a gradual, but prolonged, larval-pupal development period.

*Elasmopalpus lignosellus* is mobile and highly polyphagous, feeding on 62 plant species representing 14 families (Stone, 1968c). It is a pest of many cultivated legumes (Luginbill and Ainslie, 1917; Isley and Miner, 1944; King et al., 1961; Leuck, 1966; Razuri, 1975), rice (Sauer, 1979) and sugarcane (Plank, 1928; Bennett, 1962). The role of wild or cultivated host plants on population outbreaks is unknown but may fit the pattern proposed by Stimac and Barfield (1979).

Population outbreaks of *E. lignosellus* occur during periods of hot, dry weather (Luginbill and Ainslie, 1917; King et al., 1961; Walton et al., 1964; Bertels, 1970; French and Morgan, 1972; Smith, 1981) with high soil moisture inhibiting population outbreaks (Bertels, 1970; All and Gallaher, 1977). Thus, deep, sandy soils with good water percolation favor *E. lignosellus* population increases (Luginbill and Ainslie, 1917; Walton et al., 1964; Dupree, 1965; Leuck, 1966). Edaphic factors undoubtedly are extremely important in *E. lignosellus* outbreaks. Elucidation of the interactions among soil type, climate and *E. lignosellus* population biology is paramount for temporally and spatially predicting epidemic populations. Analysis of a study on the population dynamics of *E. lignosellus* immature stages in peanuts revealed mortality was not density-dependent; thus, abiotic factors were again implicated as dictating population levels (Johnson, 1978).

**Damage.** *Elasmopalpus lignosellus* is a key pest of peanuts in the New World, especially when hot, dry climatic conditions prevail (Bastos Cruz et al., 1962; Walton et al., 1964; Leuck, 1966; French and Morgan, 1972; Smith and Jackson, 1975; Berberet et al., 1979a; Smith and Holloway, 1979). Yield losses in excess of 70% can occur under severe attack (Smith et al., 1975).

Larvae attack all phenological stages of plant growth, feeding on subterranean plant parts. Leuck (1967) described 2 broad types of larval damage to peanut plants. The early instars (1st and 2nd) feed on vegetative buds, flower axils, ground level stems and leaves. These larvae do not consume much plant material during these early instars with damage resulting from scarification of plant parts. Older larvae feed on pegs (gynophores) and pods. Damage to the pegs and pods is considered the most damaging on runner and spanish type peanuts (Leuck, 1967; Berberet et al., 1979a). Smith and Holloway (1979), however, reported spanish type peanuts may be damaged more heavily by larvae scarifying tissue destined to become inflorescences and consuming the minute flower buds concentrated in the plant crown area prior to gynophore and pod formation. They argue that spanish type plants at a chronological age of 28-58 days post planting are phenologically more susceptible to damage from a given larval population than at later stages. During this susceptible plant age, larvae can cause a greater yield loss from a smaller amount of tissue consumed. Leuck (1967) and Berberet et al. (1979a) report *E. lignosellus* populations in Oklahoma and Georgia are very low until most plants begin to form pods, whereas in Texas high larval populations occur much earlier in plant phenology (Smith and Holloway, 1979).

Yield losses from larval feeding have been described quantitatively for Spanish-type peanuts during 2 phenological periods: 28-58 days and 60-110 days post planting. Smith and Holloway (1979) reported that larval populations up to 14,448/ha (10% infested plants) are tolerated by plants 28-58 days post planting without a yield loss. Yield losses from larval densities exceeding 14,448/ha were described in a 3 parameter nonlinear function. Berberet et al. (1979a) described yield reductions 60-110 days post planting as a function of % infested plants using a linear equation. Both studies provide the essential insect density yield loss input data for calculating economic thresholds.

Secondary invasion of larval damaged plants or pods by microorganisms has not been reported. Scarification of pods by larvae (Leuck, 1967) should assist secondary invasion as reported for other soil pests (Porter and Smith, 1974; Garcia and Mitchell, 1975).

**Management. Natural Enemies.** A list of egg-larval, larval and pupal parasites of *E. lignosellus* are presented in Table 4. Levels of *E. lignosellus* parasitism reported from peanuts vary; 2.5-8% (Johnson and Smith, 1981), 5% (Wall and Berberet, 1975), 8% (Schuster et al., 1975) and 13% (Berberet et al., 1979b). However, parasitism of *E. lignosellus* on other crops is substantially higher; 35-61% on cowpeas and soybeans (Leuck and Dupree, 1965) and 12-14% on sugarcane (Beg and Bennett, 1974; Falloon, 1978). This disparity in parasitism between peanuts and other crops was discussed by Johnson and Smith (1981). They postulated the differences were due to *E. lignosellus* larval behavior on certain plants and resulting tachinid parasitism, as tachinids only parasitize exposed larvae. Larvae attacking seedling plants frequently abandon the cryptic microhabitat of the feeding tunnel in search of new hosts as seedlings perish. This behavior is dictated by plant growth stage, as older plants are less likely to perish. *E. lignosellus* larvae usually attack peanuts at a growth stage where plant death is unlikely and larval exposure to tachinid parasitism is limited. Parasitism by Hymenoptera, however, is approximately the same on all crops. The cryptic soil microhabitat of the larvae may further restrict parasitism by concealing the larvae from the numerous general hymenopterous parasites of other lepidopterous larvae inhabiting the foliage (Johnson and Smith, 1981).

The most important hymenopterous larval and pupal parasites are *Orgilus elasmopalpi* Muesebeck and *Invreia* sp., respectively. *Orgilus elasmopalpi* parasitism accounted for 13-37% (Leuck and Dupree, 1965), 33.3% (Wall and Berberet, 1975), 24-100% (Johnson, 1978) and 4-20% (Berberet et al., 1979b) of total parasitism. *Invreia* sp. accounted for 9-77% (Berberet et al., 1979b) and 16% parasitism (Wall and Berberet, 1975) (erroneously reported as *Invreia mirabilis* (Boucek), a valid Old World species (Grissell and Schauf, 1981.) In a 3-year Texas study of *E. lignosellus* natural mortality, Johnson and Smith (1981) reported *Invreia* spp. parasitism to be erratic, ranging from 0.2-7.2% and only occurring in 2 of the 5 generations studied.

Parasitism by *O. elasmopalpi* may be curbed by high temperatures (Johnson and Smith, 1980). Parasite survival from egg to larval emergence from the host was 34% (32.2°C) and 23% (35°C) as compared to a favorable 85% (26.7°C). High levels of parasite mortality also occurred after emergence from the host at the upper temperature extremes; 59% and 0 survival at 32.2 and 35°C, respectively. The levels of field parasitism by *O. elasmopalpi* averaged 3-4% when av-

Table 4. Parasites of *Elasmopalpus lignosellus* (Zeller).

Parasite	Reference
<i>Chelonus insularis</i> (= <i>texanus</i> ) (Cresson)	Wall & Berberet 1975
<i>Chelonus elasmopalpi</i> McComb	Wall & Berberet 1975, Johnson & Smith 1981
<i>Orgilus elasmopalpi</i> Muesebeck	Wall & Berberet 1975, Falloon 1978, Johnson & Smith 1981
<i>Orgilus nitidus</i> Muesebeck	Johnson & Smith 1981
<i>Orgilus</i> sp.	Wall & Berberet 1975
<i>Habrobracon gelechiae</i> (Ashmead)	Johnson & Smith 1981
<i>Bracon mellitor</i> Say	Leuck & Dupree 1965
<i>Agathis rubricinctus</i> Ashmead	Beg & Bennett 1974
<i>Agathis</i> sp.	Johnson & Smith 1981
<i>Apanteles</i> sp.	Wall & Berberet 1975, Johnson & Smith 1981
<i>Microplitis croceipes</i> (Cresson)	Wall & Berberet 1975
<i>Macrocentrus</i> sp.	Beg & Bennett 1974, Johnson & Smith 1981
Ichneumonidae	
<i>Pristomerus spinator</i> (F.)	Wall & Berberet 1975, Johnson & Smith 1981
<i>Eiphosoma dentator</i> (F.)	Falloon 1978
<i>Diadegma</i> sp.	Frank & Bennett 1970
<i>Nythobia</i> sp.	Mercalfe 1965
Chalcididae	
<i>Invreia deceptor</i> Grissell & Schauf	Johnson & Smith 1981
<i>Invreia threa</i> Grissell & Schauf	Johnson & Smith 1981
<i>Invreia usta</i> Grissell & Schauf	Johnson & Smith 1981
Bombyliidae	
<i>Geron aridus</i> Painter	Johnson & Smith 1981
Tachinidae	
<i>Stomatomya floridensis</i> (Townsend)	Wall & Berberet 1975, Johnson & Smith 1981
<i>Stomatomya trinitatis</i> (Thompson)	Beg & Bennett 1974
<i>Stomatomya parvipalpis</i> (Wulp)	Leuck & Dupree 1965

erage maximal temperatures were 36.5°C (Johnson and Smith, unpublished). Extremely high soil temperatures could inhibit both functional and numerical responses of *O. elasmopalpi* (Johnson and Smith, 1980), as well as other *E. lignosellus* parasites.

Predators attacking *E. lignosellus* larvae include: lygaeids, *Geocoris* sp; a carabid, *Philophuga viridicollis* LeConte; and 2 therevids, *Psilocephala acuta* Adams and *Furcifera rufiventris* (LW.) (Johnson, 1978). Although no rates of predation could be assigned quantitatively, these predators were abundant in peanut fields and were often observed feeding on *E. lignosellus* larvae.

Johnson (1978) also reported a virus infecting *E. lignosellus* larvae in peanut fields. Two types of infection were noted: an acute infection which results in rapid death, cuticle disintegration and black cadaver; and a chronic form which results in prolonged larval life, a color change from greenish-blue to pallid red, and the continued structural integrity of the cadaver cuticle. Small (1-2 instar) and medium sized (3-4 instar) larvae succumbed to the acute infection, whereas the chronic infection was primarily in large (5-6 instar) larvae. Viral infection of larvae and the subsequent mortality ranged from 0-19.8% but played a minor role in *E. lignosellus* regulation.

Further studies by Mitchell (1980) identified the *E. lignosellus* virus as an *Entomopoxvirus* with a high level of virulence, attacking the larval and pupal fat

bodies and haemocytes. Infected larvae are active and eat normally until just prior to death. All infected larvae die as larvae, prepupae or pupae. Chronically infected larvae live an average of 48 days beyond the normal developmental period at 27°C.

Partial ecological life tables (Southwood, 1978) constructed on *E. lignosellus* egg, larval and pupal stages in peanuts showed total generation mortality (real mortality) to vary between 87.0 and 99.9% (Johnson, 1978). The greatest age specific mortality occurred in the egg and newly hatched (<1 day old) larval stages (49.7-86.8%) with an average of 60% + mortality occurring prior to the medium larval stage (3-4 instar). Mortality factors were found to act independently of *E. lignosellus* density; thus, biotic factors (natural enemies) were not regulating pest density. Johnson and Smith (1981) suggested that introduction of an exotic natural enemy that is ecologically synchronized with *E. lignosellus* could help regulate populations. Since *E. lignosellus* belongs to a monospecific genus and its distribution is limited to the Americas (Heinrich, 1956), candidate exotic natural enemies for suppression in the United States would need to be obtained from South and Central America or from an ecological equivalent in the Old World.

**Insecticides.** Sprayable and granular formulations of insecticides have shown varying degrees of efficacy against *E. lignosellus* on peanuts (Arthur and Arant, 1956; Cunningham et al., 1959; Harding, 1960; King et al., 1961; Bastos Cruz et al., 1962; Walton et al., 1964; French, 1971; Lee, 1971; French and Morgan, 1972; Corseuil and Terhorst, 1975; Smith et al., 1975; Berberet and Wall, 1976; Sams and Smith, 1979). Insecticidal efficacy is enhanced when either sprays or granules are applied to the soil surface (Cunningham et al., 1959; Smith and Hoelscher, 1975b; Smith et al., 1975). In the southwestern United States, sprays are recommended for dryland peanut production and granules for irrigated production (Smith et al., 1975; Hoelscher, 1977; Berberet and Pinkston, 1978). Adequate soil moisture is fundamental to efficacious results with granular insecticides.

The economics of insecticide use for *E. lignosellus* control on peanuts was originally questioned (Cunningham et al., 1959; Harding, 1960; King et al., 1961). However, larval density-yield studies (Berberet et al., 1979a; Smith and Holloway, 1979) and recent insecticidal efficacy-yield studies (French and Morgan, 1972; Smith et al., 1975; Berberet and Wall, 1976) have shown insecticidal control to be economically efficacious, dependent upon larval population density and plant age. Yield increases of up to 250% (Smith et al., 1975) and 29% (Berberet and Wall, 1976) have been reported in the southwestern United States on dryland peanuts where sprayable insecticides have been used. Under irrigation or adequate soil moisture, granular insecticide formulations resulted in yield increases of 78% in the southwest (Smith et al., 1975) and 45% in the southeast (French and Morgan, 1972).

Economic thresholds for insecticide applications in the southwest are 5% infested plants prior to the initiation of pegging and 10% after the initiation of pegging (Hoelscher, 1977; Berberet and Pinkston, 1978). Thresholds for irrigated peanuts are higher—10 and 15%, respectively—since adequate soil moisture allows for a greater probability for plant damage recovery. Economic thresholds are based primarily upon data from insecticide trials and larval density-yield studies. To date, no efforts have been published which refine existing economic thresholds by including the variables of control costs, produc-

tion values and risk. Smith and Holloway (1979) contend that economic threshold for *E. lignosellus* cannot be further refined until population densities can be predicted accurately in advance of actual losses, thus allowing the calculation of reliable estimates of probable yield loss. Estimates of probable loss are mandatory for balancing probable losses against costs and optimizing the net return (Berberet et al., 1979a; Smith and Holloway, 1979).

Field sampling to estimate pest population densities is inherent to IPM programs in which economic thresholds dictate timing of insecticide applications. Two main sampling plans for estimating *E. lignosellus* larval population densities currently are in use in the United States. In the southwest, individual plants are examined to determine the proportion of plants infested with larvae (Smith and Hoelscher, 1975b; Berberet and Pinkston, 1978); and in the southeast, thresholds are based on the proportion of 0.91 m sections of row infested with larvae (French and Weeks, 1978). Both sampling plans involve a time consuming manual search of the subterranean plant parts and adjacent soil for larvae. Early instar larvae are normally difficult to find; wet soils increase the difficulty of locating all instars (Smith and Hoelscher, 1975b). Large (<1.3 cm) *E. lignosellus* larvae caught in pitfall traps provide as accurate an estimate of field density as sampling 0.91 m sections of row (Jones and Bass, 1979). Pitfall sampling reduced sampling time 33-50% and enabled sampling under wet conditions. All currently used sampling programs for estimating *E. lignosellus* larval densities in peanuts underestimate the actual density because the small, early instars are difficult to census. Sampling also requires destruction of the plants, is time consuming and laborious and is sensitive to soil moisture.

The use of ecologically selective insecticide formulations (i.e., granules) and application techniques which direct the insecticide to the soil habitat of the target organism has been shown to have biological as well as economical benefits. Granular insecticide formulations applied in irrigated peanut cultures for suppression of *E. lignosellus* conserved natural enemies of the numerous foliage inhabiting arthropods (Smith and Jackson, 1975) and increased insecticidal efficacy against the target species (Smith et al., 1975). Basal directed sprays increased insecticidal efficacy when compared to broadcast sprays (Cunningham et al., 1959; Smith and Hoelscher, 1975b; Smith et al., 1975); however, basal directed sprays are not selective enough to conserve the extant, nontarget, foliage inhabiting arthropods (Smith and Jackson, 1975). Basal directed sprays do decrease the total insecticide load on the peanut ecosystem by reducing the number of applications necessary to maintain *E. lignosellus* below established economic thresholds. Reducing the total number of applications aids in conservation of natural enemies during the entire growing season. Ecologically selective pesticide application techniques, used in concert with economic thresholds, help circumvent nontarget pest upset. Problems of conservation of natural enemies, changes in pest status, selection of pesticide resistant populations and economic losses (Smith and Jackson, 1975; Smith et al., 1975) are minimized.

The economic benefits of ecologically selective insecticidal application techniques for suppression of *E. lignosellus* in Texas are dramatic. The major thrust of an IPM program introduced to peanut producers in 1971 utilized ecologically selective insecticide applications and economic thresholds for the key pest, *E. lignosellus*. Prior to 1971, insecticidal application techniques were

mainly broadcast sprays applied by fixed wing aircraft (nonselective). During 1971, aerial application decreased 53% on irrigated fields and 31% on nonirrigated (Smith and Hoelscher, 1975a). Insecticides applied by ground driven equipment (granules and directed sprays) were unchanged. The effectiveness of the program in managing *E. lignosellus* is further illustrated by changes in the proportion of producers using multiple applications. In 1970, 30% of the producers used 5+ applications as opposed to 6% in 1971. Also in 1970, 69% of the producers used 3 or more applications while 72% used 1 to 2 applications in 1971 (Smith and Hoelscher, 1975a). Thus, within 1 year 82% of the growers had adopted the selective application method. The drastically reduced insecticide load on the peanut ecosystem was attributed to adherence to economic thresholds and increased effectiveness of the insecticides on the target soil pest (Smith and Johnson, 1977).

**Resistant Cultivars.** Resistant germplasm has been identified in commercial cultivars, noncommercial cultivars and wild species. Levels of resistance identified have been low, but even those low levels have utility in an IPM program (Smith, 1980; Smith et al., 1980b).

A wide variation in seedling survival among 108 peanut lines artificially infested with *E. lignosellus* eggs provided the first evidence of resistant germplasm (Leuck and Harvey, 1968). Field evaluations of 14 advanced peanut lines, infested in a similar manner but at a more advanced stage of plant growth, failed to show any differences in plant response between lines or types (e.g., spanish, runner and virginia) (Leuck et al., 1967). Several of the lines investigated by Leuck et al. (1967) also were included in the Leuck and Harvey (1968) report. In the latter report, several of the lines reevaluated as seedlings showed a more variable response. Smith et al. (1980 a, b) screened 490 peanut cultivars and identified 81 cultivars which scored significantly less damage than Starr, a commonly grown cultivar in the southwestern United States identified as being the most susceptible of the 490 cultivars. Among these putative resistant cultivars were the commercial cultivars Florunner (tolerance), Early Runner (antibiosis), and Virginia Bunch 67 (tolerance). Further trials with Florunner, Early Runner and Virginia Bunch 67 carried to maturity under field conditions substantiated the greenhouse results. Yield reductions, due to *E. lignosellus* infestations, measured by comparing the same variety with and without an insecticide umbrella, were: Early Runner, 8.4%; Florunner, 19%; Virginia Bunch 67, 19%; Florigiant, 15%; Dixie Spanish, 26%; and Comet, 45% (Schuster et al., 1975). Comet is a commercial cultivar, susceptible to *E. lignosellus* (Kamal, 1973). Florigiant, Florunner and Early Runner also reduced larval survival better than Comet, offering a level of antibiosis. These varieties, Florunner, Florigiant, Early Runner and Virginia Bunch 67, are thought to possess only low but useful levels of resistance to *E. lignosellus* (Smith, 1980).

Schuster et al. (1975) observed parasitism of *E. lignosellus* larvae by hymenopterous parasites to be higher (12.5-18.7%) in prostrate growing cultivars (cv. Florunner) when compared to erect growing spanish cultivars (0.0-1.6%) and suggested that combining intermediate levels of resistance with enhanced parasitism from growth habit could be important in *E. lignosellus* management. Berberet et al. (1979b), however, found that parasitism was not enhanced by prostrate plant growth habit.

**Cultural.** Culturally oriented management tactics include planting dates, sanitation, winter plowing, irrigation and crop rotation. Many cultural practices have resulted from observations on peanuts and other agronomic crops in earlier reports and lack a sound data base.

Removal of crop residues, fall and winter plowing and weed removal reportedly help prevent severe infestations (Luginbill and Ainslie, 1917; Watson, 1917; Guyton, 1918; Box, 1929; Stahl, 1930; Hayward, 1943; Isley and Miner, 1944). In crops such as corn, sorghum and peas, *E. lignosellus* is most destructive when attacking the seedling stage. Many authors (e.g., Isley and Miner, 1944; Wilson and Kelsheimer, 1955; Reynolds et al., 1959) felt crop residues and alternate hosts (weeds) in the fields prior to planting were the sources of damaging larvae on seedling susceptible crops, since seedlings would grow out of this susceptible stage before eggs oviposited in the planted crop could develop into the damaging, older larval stages. Cultivating all infested hosts after planting could force the entire larval infestation to concentrate on the crop seedlings; therefore, sanitation should be practiced several weeks in advance of planting (Reynolds et al., 1959). Dupree (1964) reported that tillage and maintenance of weed free fields 8-10 weeks prior to planting cowpeas gave *E. lignosellus* control equal to insecticides. The level of larval infestation was shown to be different in till (clean field) versus no-till (crop residue) corn (All and Gallaher, 1977; All et al., 1979). The feeding behavior of larvae apparently is modified when crop residues are present (Cheshire et al., 1977), as larval damage to seedling corn was reduced greatly when plant residues were available as a food source (Cheshire and All, 1979).

Flood irrigation readily controlled *E. lignosellus* larvae (Reynolds et al., 1959). Irrigation also increased soil moisture in corn and was an important factor in prohibiting *E. lignosellus* infestations (All and Gallaher, 1977). Increased plant vigor (by eliminating drought stress) and increased soil moisture are both associated with irrigation and should help reduce damage from *E. lignosellus* larvae.

Peanuts planted so that the most susceptible phenological stage(s) will not synchronize temporally with peak *E. lignosellus* populations would minimize damage. In the southeastern United States, peanuts planted prior to mid-April escape peak populations of *E. lignosellus* (Leuck, 1967). Correspondingly, peanuts planted in May in Texas should obtain sufficient maturity to escape heavy damage by the pest populations which peak near mid-August (Johnson, 1978; Smith and Holloway, 1979).

#### Coleoptera

The Coleoptera are a large and diverse group of phytophagous insects on peanuts (Table 1). Many species are foliage consumers as adults and subterranean feeders as larvae. Subterranean feeding larval stages constitute the greatest peanut pest problem, as spatial tolerance is nil. The scarabaeids (white grubs), elaterids (wireworms), tenebrionids (false wireworms), certain curculionids and chrysomelids (*Diabrotica* spp.) are representative of major coleopterous pests where adults and larvae occur in separate habitats. The subterranean coleopterous pests as a group are poorly known, with published information existing as a potpourri of miscellaneous notes.

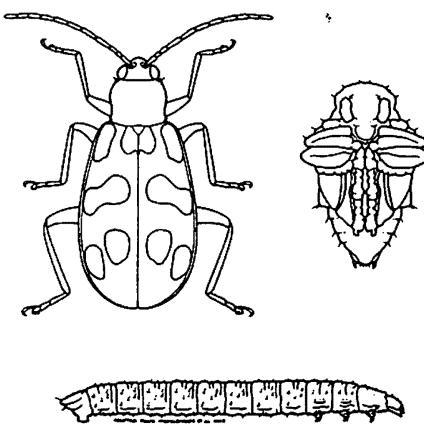
*Diabrotica* spp. *Diabrotica balteata* LeConte, *D. speciosa* Germar, and *D. undecimpunctata howardi* Barber have been reported attacking peanuts. These beetles have a biology different from the other chrysomelid genera listed in Table 1 as adult females oviposit on the soil and the larvae are subterranean feeders. In the United States, *D. undecimpunctata howardi* has been recognized as a pest of peanuts since the beginning of the 20th century (Fink, 1916) and has received considerable attention by entomologists.

The preponderance of literature on *D. undecimpunctata howardi* dictates concentration on this species. However, in certain geographical areas (e.g., United States Gulf Coast area), *D. balteata* may be the dominant species (Feakin, 1973). The difficulty in separating *Diabrotica* spp. larvae taxonomically (Bass and Arant, 1973) may have led to an incomplete understanding of the total species involved throughout the New World.

**Biology.** Overwintered adult female *D. undecimpunctata howardi* oviposit in the soil preferring a shady, moist substratum (Howard, 1926; Campbell and Emery, 1967). Females oviposit over an average of 48 days (Isley, 1929). Field collected females laid from 116-895 eggs (Sweetman, 1926). Oviposition is related to both temperature and relative humidity (Campbell and Emery, 1967). Average eggs laid per female were 0.3, 20.3, 110.6, 106.0, 284.5 and 168.7 at 7, 13, 18.5, 24, 29.5 and 35 C, respectively. Total eggs laid in 7 days (24 C) also increased from 19.7 to 29.0 with relative humidities (RH) of 55 to 97%. Females did not oviposit at a RH of 25%.

Eggs hatch in 6-13 days depending upon temperature (Sweetman, 1926). Eggs will not hatch below 75% RH with peak % hatch occurring above 85% RH (Campbell and Emery, 1967). Larvae feed on the subterranean plant parts for ca. 21 days and then form a pupal cell in the upper 7.6 cm of soil (Sweetman, 1926). The prepupal period requires 6.3 days and the pupal 8.5, with a total of 46.3 days (average) needed to complete a generation (Sweetman, 1926). Under laboratory conditions of 27 C and 60-70% RH, egg incubation was 7 days, larval stage 10 days and prepupal and pupal stages 10 days (Hays and Morgan, 1965). Under laboratory conditions, the life cycle is completed in ca. 27 days (27 C constant), whereas Sweetman (1926) recorded in excess of 2 months for development at a northern latitude.

Three to 4 generations occur each year in the southern United States (Hays and Morgan, 1965); 1 generation occurs in the north (Sweetman, 1926). In the southern states, *D. undecimpunctata howardi* does not enter a completely dormant state during the winter; rather, it overwinters in organic debris near fields as an adult which is active when temperature permits (Hays and Morgan, 1965). According to the observations of Smith and Allen (1932), *D. undecimpunctata howardi* migrates northward during the spring and early summer. Progeny migrate southward during the fall, with no winter survival north of cen-



tral Missouri. Adults are mobile and highly polyphagous, feeding on 208 plant species (Sell, 1916). The larvae attack corn, cucurbits and most agronomic legumes.

**Damage.** Adult *D. undecimpunctata howardi* feed on peanut foliage, preferring the terminal buds (Hays and Morgan, 1965). Economic injury is due principally to larval feeding on subterranean pegs and pods (Grayson and Poos, 1947). Pods are highly susceptible to injury from the time they begin to form until they approach maturity (Grayson and Poos, 1947). Larvae prefer young, soft pods to the older, more mature pods (Fink, 1916). Pegs are also heavily damaged when they first penetrate the soil, prior to enlarging to form pods (Grayson and Poos, 1947). Losses from larval feeding can reduce the yield of whole seed by as much as 80% (Feakin, 1973).

Larval injury predisposes the pod to attack by soil microorganisms (Grayson and Poos, 1947). Pod damage facilitates entry of the pathogenic fungus, *Pythium myriotylum*, which has caused increasing damage to the peanut crop in the Virginia-Carolina area of the United States (Porter and Smith, 1974).

**Management. Natural Enemies.** Information on specific natural enemies of *Diabrotica* spp. is quite limited. Tachinids parasitic on *D. undecimpunctata* adults include *Pseudomyiothyria ancilla* (Walker) (Arnaud, 1978), *Celatoria* (= *Chaetophleps*) *setosa* (Coquillett) (Bussart, 1937) and *C. diabroticae* (Shimer) (Fronk, 1950); the latter are also parasitic on larvae (Arant, 1929). *Celatoria bosqi* Blanch attacks *D. speciosa* Germar in Argentina (Christensen, 1944).

Parasitic nematodes include *Diplogaster* spp., *Neoplectana* sp. and *Howardula benigna* Cobb (Fronk, 1950). Entomogenous fungi have been reported from South America (Christensen, 1944). *Apionerus crassipes crassipes* (F.) (Reduviidae) (Morrill, 1975), and species of *Xantholinus*, *Anisodactylus*, *Agonum*, *Amara* and *Pterostrichus*, (all Coleoptera) (Fronk, 1950) have been observed preying on *D. undecimpunctata howardi*. Only the parasitic fly, *C. diabroticae*, and nematode, *H. benigna*, occurred with regularity, parasitizing 3.7% and 23.6%, respectively, of adult *D. undecimpunctata howardi* (Fronk, 1950).

**Insecticides.** Prior to the early 1960's, the cyclodiene insecticides (e.g., aldrin, heptachlor) were used extensively as soil broadcast and banded treatments for *D. undecimpunctata howardi*. This method of control was so effective and in such widespread, general use as a preventative control tactic that *D. undecimpunctata howardi* was seldom considered a pest (Boush et al., 1963). Aldrin and heptachlor gave season long control and were effective as spray, dust and granular formulations (Boush et al., 1963). These formulations were available in convenient form for preventative treatments (e.g., mixed with fertilizers) (Ritcher, 1953) and were used heavily over a wide geographical area. The result of this pesticide load was *D. undecimpunctata howardi* resistance to cyclodiene insecticides in ca. 10 years (Boush et al., 1963). Cyclodiene insecticides persist in the soil for the entire growing season with high residue concentrated in the pods and seed (Beck et al., 1962; Dorough and Randolph, 1967; Morgan et al., 1967).

Many organochlorine, organophosphate and carbamate insecticides give adequate control of *D. undecimpunctata howardi* larvae (Howe and Miller, 1954; Arthur and Arant, 1956; Boush et al., 1963; Boush and Alexander, 1964; Hays and Morgan, 1965; Smith, 1971a, 1972b, 1976b, 1977a, b). However, the organochlorines, DDT and chlordane, leave undesirable residues in the

peanut pods (Dorough and Randolph, 1967; Morgan et al., 1967). Historically, all insecticides have been used in a preventative manner, applied either at planting or when the plants begin to peg.

Certain peanut varieties (and resulting growth habit) may influence the efficacy of granular insecticides (Campbell et al., 1976). Diazinon and ethoprop were ineffective in controlling *D. undecimpunctata howardi* on NC Ac 15753 (bunch growth habit and *D. undecimpunctata howardi* susceptible), gave moderate control on cv. Florigiant (runner growth habit and susceptible) and gave excellent control on NC 343 (intermediate growth habit and resistant). Cultivars resistant to *D. undecimpunctata howardi* (e.g., NC 6, NC 343) require less insecticide (as much as 75% less) to achieve adequate control (Campbell et al., 1976; Wynne et al., 1977).

**Resistant Cultivars.** After screening 2,500 peanut cultivars for resistance to *D. undecimpunctata howardi*, Boush and Alexander (1965) concluded that spanish cultivars were more resistant than valencia, with virginia cultivars being the most susceptible. A highly resistant spanish cultivar (PI 262048) was crossed with 2 virginia cultivars in an attempt to transfer resistance (Alexander and Smith, 1966). Resulting F<sub>1</sub> plants were more resistant than the susceptible parents (virginia cultivars) but were more susceptible than the resistant parents.

Survival of *D. undecimpunctata howardi* larvae was measured on germinating seeds of 172 cultivars under laboratory conditions (Chalfant and Mitchell, 1967). Resistance (antibiosis) was identified in those cultivars with larval survival  $\leq 10\%$ , which comprised ca. 5% of the total cultivars. Selected putative resistant and susceptible cultivars were further evaluated in field experiments (Chalfant and Mitchell, 1970). Results from field evaluations were not always congruent with laboratory data. For example, PI 221068 was susceptible in the laboratory and resistant in the field trials. The reverse was true for cv. Georgia Station Runner. Attempts to explain variability in pod damage by planting according to pod maturity (plant phenology), as opposed to a single planting date, were unsuccessful. The authors did, however, explain that resistance evaluations based on % damaged, immature and undamaged pods could result in spurious conclusions since maturity and peg formation rates are highly variable among cultivars. The rate of pod maturity may also be an extremely important resistance factor. Fink (1916) reported that peanut plants in which the pods are matured or are maturing rapidly were either free from larval damage or decidedly less injured. Spanish cultivars mature faster than most virginia or runner cultivars, which may explain the general result of spanish cultivars possessing resistance (Boush and Alexander, 1965; Hays and Morgan, 1965).

The original observation of Fronk (1950) that plant growth habit, pod size and seasonal insect and plant development were related to resistance were considered by Smith (1970) in developing a greenhouse screening technique for identifying *D. undecimpunctata howardi* resistance to peanuts. Smith (1970) measured larval damage to both immature and mature fruit and found no preference for either. Cultivar NC 10211 sustained the least damaged fruits (9.2%) while cv. Argentine and NC 343 were the highest damaged (28.5 and 34.0%, respectively). Nine cultivars considered possibly resistant (Smith, 1970) were reevaluated by subjecting the cultivars to 3 pest levels: 25, 50 and

100 larvae (Smith and Porter, 1971). Differences between % infested fruit occurred at the low levels of infestation (25 and 50 larvae), but no differences were discernible at the high infestation level. The authors concluded that high levels of resistance were not available in the cultivars evaluated, and that demonstrated levels of resistance would not give commercially acceptable control.

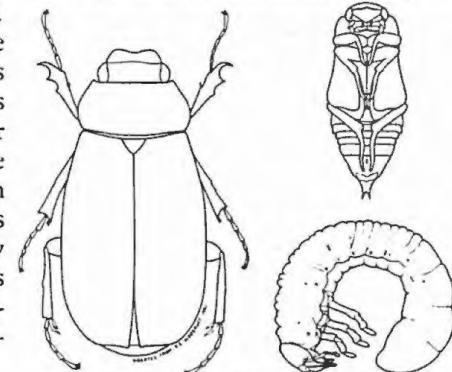
It is interesting to note that NC 6, a commercial virginia type cultivar resistant to *D. undecimpunctata howardi*, was developed from a cross of NC 343 x Va 61R (Campbell et al., 1971; Wynne et al., 1977). Both parents of NC 6 were considered previously susceptible to larval feeding in greenhouse studies (Smith, 1970; Smith and Porter, 1971). The absence of the expression of resistance by NC 6 parents in the greenhouse tends to indicate resistance is related to multiple factors expressed under field conditions and not in the laboratory. Also, Boush and Alexander (1965) and Hays and Morgan (1965) stated that spanish type cultivars are sources of resistant germplasm. This could be related to rates of change in plant phenology and growth characteristics as speculated by Fronk (1950). NC 6 is especially adapted to heavier soil types where *D. undecimpunctata howardi* is consistently a problem. As additional positive qualities, it possesses moderate resistance to thrips (*Frankliniella fusca*), *Emblepsa fabae* Harris and *Heliothis zea* (Campbell and Wynne, 1980).

**Cultural.** Moisture content of the soil is probably the most important single environmental factor in determining population levels of *D. undecimpunctata howardi* (Grayson and Poos, 1947). High soil moisture and a roughened soil surface make a preferred oviposition substratum for adult females (Howard, 1926). Avoidance of heavy soils with high organic matter content in poorly drained areas helps prevent larval population buildup (Fronk, 1950).

Larval injury is greater on crops that follow winter cover crops of green manure but injury may also be severe in winter fallow fields (Bissell, 1936, 1940; Feakin, 1973). Injury may be greater when peanuts are grown annually on the same field (Fink, 1916). Fronk (1950) found no differences in *D. undecimpunctata howardi* injury between 3 different dates of planting in Virginia. Plant spacings of 15, 23 and 30 cm apart and row widths of 0.61 and 0.84 m had no effect on plant injury.

**White Grubs.** Numerous species of white grubs (Scarabaeidae), attack peanuts throughout the world (Table 1). Some species are pests as both larvae and adults, while others are pests as either adults or larvae. Many species are highly polyphagous and have 1 or 2 year life cycles. Long white grub life cycles present special problems in crop rotation especially when peanuts are planted behind new land or weedy fallow. In general, the life histories are poorly understood and information relating damage and plant phenology is nil.

**Biology.** The biology of *Lachnostenus consanguineus* (Blanchard) in India has been studied by several authors (Kalra and Kulshrestha, 1961; Desai and



Patel, 1965; Patel et al., 1967; Rai et al., 1969; Srivastava et al., 1971a). The following biology is a compilation of these reports.

The adult beetles are polyphagous, nocturnal feeders of the foliage and inflorescence of numerous plants. Adults emerge from the soil pupation site, leaving behind a round hole on the soil surface. Maximum adult emergence and mating coincides with the onset of the rainy season. Beetles emerge from the soil sharply between 7:30 and 8:00 p.m. and fly to adult host plants to feed. Sharply between 5:00 and 5:30 a.m., the beetles return to the soil. Caged beetles also exhibit this distinct periodicity in activity. Adult beetles prefer to feed on the foliage of numerous trees adjacent to the cultivated crops and fallow fields where larvae developed rather than cultivated crops.

Adult females lay eggs singly in the soil up to a depth of 10 cm. Oviposition coincides with the onset of the monsoon. Eggs incubate for 7-12 days. White grubs complete 3 instars feeding on the subterranean plant parts for 8 to 10 weeks. The grubs remain in the upper 15 cm of soil when the soil is moist, descend deeper (up to 0.75 m) during dry periods and return close to the surface when soil moisture returns. The grub population tends to be denser in the raised portions of infested fields. Full grown grubs descend deeper in the soil (up to 0.75 m) to pupate. Pupation requires 13 days at 27°C. The adults hibernate in the soil until the onset of the next monsoon. Only 1 generation occurs annually.

In Australia, the scarabs *Rhopaea magnicornis* Blackburn, *Heteronyx brevicollis* Blackburn and *Trissodon* (= *Isodon*) *puncticollis* MacLure (Smith, 1946) are pests of peanuts. Only *R. magnicornis* and *H. brevicollis* larvae are damaging with *T. puncticollis* feeding on peanuts only as adults.

Adult *R. magnicornis* are active between November and January with females ovipositing in the soil. Larvae hatch within 21 days and feed for approximately 16 months. The last larval instar is a voracious feeder and occurs in November of the second year. Pupation occurs in an earthen cell deep in the soil. The resulting adult emerges with the onset of the rains. This scarab has a 2-year life cycle and attacks peanuts planted on previously fallow or pasture land where the adult oviposited the year prior to the crop being planted. The adults also tend to be active on friable, volcanic, red soils (Smith, 1946).

Adults of *H. brevicollis* oviposit at the base of peanut plants and weeds. Adults prefer oviposition sites which offer a high degree of physical cover and are attracted to organic manure (Passlow, 1969). In Africa, *Eulepida mashona* Arrow is attracted to organic manure for oviposition with outbreaks more prone to occur in areas where *Brachystegia* spp. trees are dominant (Rose, 1962; Broad, 1966). The adults are abundant for the first few weeks of the rainy season, feeding on the foliage of *Brachystegia* spp. and related trees.

*Strigoderma arboricola* F. is a common, widely distributed, white grub feeding on the pods of peanuts in the eastern United States (Miller, 1943; Grayson, 1947). This insect has 1 generation per year with the third instar overwintering at a soil depth of 18-20 cm (Grayson, 1946). Adult females lay an average of 26.3 eggs which require 8-21 days to hatch. The first 2 larval instars last 12-30 and 10-36 days, respectively, with the third and final instar lasting 238-292 days (Grayson, 1946). Peanuts grown in dark soil, high in organic matter and where the plants are large, are more prone to attack (Miller, 1943).

**Damage.** Adults are either defoliators of peanuts (e.g., *Heteronyx* spp.,

Passlow, 1969; *H. brevicollis*, Smith, 1946) or feed on the main plant stem severing the top of the plant from the roots (e.g., *T. puncticollis*, Smith, 1946). Adults of several species in which the larval stages are also pests prefer to feed on the foliage of trees (e.g., *L. consanguinea*, Rai et al., 1969; Srivastava et al., 1971a; *E. mashona*, Rose, 1962; Broad, 1966).

Larvae feed on the roots, pods and nitrogen nodules on the roots (Miller, 1943; Smith, 1946; Rai et al., 1969; Mercer, 1978b). The tap root system of peanuts make it particularly susceptible to root feeding (Rai et al., 1969; Srivastava et al., 1971a). Dry weather exacerbates the wilting caused by larval root pruning. Larvae feeding on root systems and pods may facilitate secondary invasion of fungi (Mercer, 1978a).

**Management. Natural Enemies.** *Lachnostenia* spp. grubs are parasitized by the scoliid, *Scolia aureipennis*, a fungus, *Metarrhizium anisopliae* (Kalra and Kulshrestha, 1961) and a milky disease, *Bacillus* spp. (Patel et al., 1978). *Anthia sexguttata* (F.), a carabid (Rai et al., 1969), *Bufo melanostictus*, a toad and *Gecko gecko*, a gecko (Kalra and Kulshrestha, 1961) prey on adults. Birds may be an important larval predator, especially in newly plowed fields (Yadava et al., 1975). Prasad (1961) reviewed the importance of natural enemies for regulating white grubs in India and suggested the introduction of *Bufo marinus*, the Surinam toad, to bolster predation.

The natural enemies of the white grubs in the genus *Leucopholis* were reviewed by Leefmans (1915). Numerous scoliid wasps were reported to parasitize *Leucopholis* with 1 species, *Dielis thoracica*, parasitizing 26% of the larvae.

**Insecticides.** Insecticidal control of *L. consanguinea* in India has received a great deal of attention (Desai and Patel, 1965; Kaul et al., 1966; Patel et al., 1967; Ravindra and Thobbi, 1967; Rai et al., 1969; Sharma and Shinde, 1970; Srivastava et al., 1971b; Bindra and Singh, 1972; Yadava and Yadava, 1975; Prasad, 1977; Yadava et al., 1977; Yadava et al., 1978). Control has been targeted toward both adults and larvae. Grub control with a granular insecticide has increased yields by 794 kg/ha when the insecticide treated area had 24% plant mortality and 68% grub control (Prasad, 1977).

Applying an insecticide to the trees where the adult beetles congregate to feed has had some success (Yadava et al., 1977). It is imperative that the timing of the sprays coincide with peak adult emergence after the monsoon onset. Control must be applied on an areawide basis.

**Cultural.** Species of white grub adults attracted to organic manure (e.g., *E. mashona*, Rose, 1962; Broad, 1966; *Heteronyx* spp., Passlow, 1969) for oviposition could be managed by avoiding the practice of deep burial or organic manure immediately after application. White grubs with 2-year life cycles (e.g., *R. magnicornis*), where oviposition originates in weedy fallow or pasture lands, are manageable by planting peanuts on land previously under cultivation for 1 or 2 years (Smith, 1946).

## OVERALL PERSPECTIVE

The body of knowledge in population ecology comprises the theoretical basis for the use of integrated pest management (IPM), and violations in ecologically founded management principles have caused severe constraints in design of pest management strategies (Bottrell, 1979; Barfield and Stima, 1980).

Worldwide peanut crop production and protection have not been immune to mistakes made in other agricultural production systems. The present chapter has summarized pest types, particular pests, variant pest biologies and protection practices for insect pests of peanuts worldwide. Several observations can be made concerning this summary which will allow analysis of where improvements can be made in the design of peanut crop protection strategies worldwide.

An overview of tactical approaches to insect management in peanuts worldwide (Table 5A) reveals that, by far, chemical pesticides comprise the mainstay protection practice; yet, published research depicts a tremendous potential (Table 5B) for development of multitactic IPM strategies sufficiently adaptable to particular sites. Further, select insect pests vary from key pests to non-pests (Table 6) over geographical space. This obviously reflects the effects of site-specific environments (biotic and abiotic components) on pest dynamics. Thus, site specific IPM strategies must be structured consistent with local pest dynamics. Only then can we begin to decipher the generality of a given management strategy.

Tables 5A, B reflect a basic problem seemingly common to most all current crop protection programs: almost unilateral dependence upon pesticides often applied in a prophylactic manner. Economic thresholds, sampling methodologies and systems level design, inherent to utilizing pesticides under the IPM philosophy, are generally lacking. Within these 3 areas lie the keys to improving peanut crop protection strategies based on the pesticide tactic.

Table 5. Current usage of select IPM tactics and sampling for select groups of peanut insect pests worldwide (A) and availability of tactics and sampling plans (B) whether or not actually used. Large denomination (X) represents significant use; small (x) represents relatively minor use. Blank spaces represent non-availability.

	Host Plant Resistance	Cultural Practices	Importation	Conservation	Augmentation	Insecticides	Scouting Program Used	Economic Threshold
<b>A.</b>								
Foliage Feeders								
Lepidoptera				x		x	x	x
<i>Aphis craccivora</i>			x	x	x			
<i>Elasmopalpus lignosellus</i>	x				x		x	x
<i>Diabrotica undecimpunctata</i>	x	x				x		
Spider Mites						x		
Thrips	x	x				x		
<b>B.</b>								
Foliage Feeders								
Lepidoptera	x		x	x		x	x	x
<i>Aphis craccivora</i>	x	x	x	x		x		
<i>Elasmopalpus lignosellus</i>	x	x	x	x		x	x	x
<i>Diabrotica undecimpunctata</i>	x	x		x		x		
Spider Mites	x	x	x	x		x		
Thrips	x	x	x	x		x		

Table 6. Pest status of select peanut insect pests worldwide.

	United States	South America	Africa	Asia
Thrips	Non-pest	Key pest	(unknown)	(unknown)
Aphids	Non-pest	Non-pest	Key pest	Key pest
Spider Mites	Secondary pest	(unknown)	Secondary pest	Secondary pest
<i>Empoasca</i>	Non-pest	(unknown)	(unknown)	(unknown)
Foliage Consumers	Occasional pest	Occasional pest	Occasional to Secondary pest	Occasional pest
<i>Elasmopalpus lignosellus</i>	Key pest	Key pest	Does not occur	Does not occur
<i>Diabrotica</i> spp.	Key pest (regionally)	Occasional pest	(unknown)	(unknown)
White Grubs	Occasional pest	Occasional pest	Occasional pest	Occasional pest
Termites	Non-pest	Non-pest	Key pest	Key pest

The concept of an economic threshold is not new to agriculture and has been reiterated earlier. Despite much dialogue on the concept, researchers simply have not addressed the complexity of construction of realistic, dynamic thresholds. Peanut researchers are no exception. The literature is clogged with discussions that true economic thresholds are functions of pest density, crop stage, physical environment, market value, pest combinations, etc. Yet, the fact remains that, in actuality, economic thresholds (where used at all) are used as static values for single pests. Until agricultural researchers (in this case, for peanuts) address the construction of more realistic thresholds, crop protection schemes will continue to be based more on experience than on the dynamics of specific ecological/sociological/economic stages. Barfield and Stimac (1980) argue that systems models offer the most promising tools currently available for derivation of these thresholds.

As a whole, peanut (as other) IPM practitioners have a poor perception of the relationships among sample allocation, sample unit size, numbers of samples and target pest dispersion. In virtually all cases, no reliable relationships have been derived to relate relative pest densities to absolute densities. Since actual damage density relationships are functions of absolute pest densities, this remains a legitimate problem. Despite much dialogue on sampling, researchers still have focused on sampling methodologies more for convenience than for reliability. So-called practical sampling plans have been given priority over first addressing reliable sampling procedures, then extracting practical features. This shortcut has resulted, in most cases, in a lack of ability to evaluate management strategies. How can threshold levels be established or the results of a new tactic be evaluated if reliable estimates of absolute, not merely relative, density cannot be made? By and large, peanut researchers have not dealt with this problem. Linker (1980) is the best exception. If improvements in using the pesticide tactic are to be realized, attention must be given to the design of reliable sampling methodologies.

The proper use of pesticides (i.e., dynamic economic thresholds, reliable sampling methodologies, target pest selectivity, etc.) requires more technology on the user level than other IPM tactics (e.g., host plant resistance, cultural practices, biological control). In developing countries of the world where user level technology, mechanization and desirable pesticides are limited, IPM strategies must rely more on tactics other than pesticides. This of course does not mean that the more developed countries should continue their reliance on pesticides, but that these constraints are inherent to subsistence and small farm agriculture. Figure 5B reveals that several tactics are available for peanut insect management but are yet to be implemented either because the tactics have remained in a developmental or research stage or have been ignored for the more convenient pesticide. Resistant peanut germplasm has been identified for each pest discussed (Table 5B) but has only been sparsely utilized (Table 5A).

Possibly the most neglected tactic in peanut insect management has been biological control, especially in the use of exotic natural enemies (importation, Table 5A, B) and augmentation of extant natural enemies. Most pest species spend part of their life cycle on alternate hosts or occur as pests by immigration from alternate hosts. Successful biological control on alternate hosts (the more stable habitat) could reduce the immigrant inoculum level below economic levels. Natural enemies following the pest movement into the field would further reduce pest population increase.

Stimac and Barfield (1979) illustrated both pest and spatial hierarchies for a soybean crop production system. Conceptually, peanuts can be depicted identically. Most peanut researchers appear to recognize that the dynamics of insect pests may be driven by biotic or abiotic influences outside the peanut field. Yet, crop protection scenarios continue (at least in the United States) to be developed unilaterally to system level understanding. This, in large part, reflects the infancy of knowledge on how polyphagy and mobility affect pest dynamics (see Barfield and Stimac, 1981). The pulse of insects into a peanut field from sources outside that field (crop and noncrop) may be more important in determining pest status than the conflicting forces of development and mortality occurring in the field. In short, pest status is a function of the crop production system, not merely the crop. While most researchers, when pressed, recognize this fact, necessary research to address such a complex problem is almost totally lacking. This must be overcome if significant progress is to be made in designing peanut crop protection strategies.

Finally, we are convinced that the potential exists (see Table 5B) to design robust production and protection strategies for peanuts. Reallocation of resources to address management of pest arthropods with a multitude of tactics is paramount to upgrading IPM in peanuts.

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