Higley, L.G., and R.K.D. Peterson. 1994. Initiating sampling programs. In L.P. Pedigo and G.D. Buntin (eds.). Handbook of Sampling Methods for Arthropods in Agriculture. CRC Press, Boca Raton, FL.

Chapter 7

INITIATING SAMPLING PROGRAMS

Leon G. Higley and Robert K. D. Peterson

TABLE OF CONTENTS

I. Introduction 120

II. Deciding When to Begin Sampling 120
   A. Timing and Monitoring 120
   B. Host, Pest, and Environmental Considerations 121

III. Techniques 121
   A. Initiation Based on Life History and Environment 121
      1. Calendar Date 121
      2. Migration 122
      3. Thermal Development 124
         a. Why Does Temperature Matter? 124
         b. Approaches for Estimating Development 127
         c. Limitations 129
         d. Using Estimates of Thermal Development in Sampling 129
   B. Initiation Based on Ongoing Monitoring 130
      1. Hosts 130
      2. Alternate Hosts and Attraction Sites 131
      3. Trapping 131
      4. Surveys 132
      5. Remote Sensing 132
   C. Initiation Based on Pest Impact 133

IV. Practical Considerations 134

References 134

ISBN 0-8493-2923-X/94/$0.00+.50

I. INTRODUCTION

Initiating sampling programs involves questions of efficiency and of risk. It is inefficient to begin programs too early, before sufficient numbers of insects are present to allow accurate estimates of population density. On the other hand, waiting to sample until pest populations reach their highest numbers presents the risk that significant injury will occur before sampling and management action can be taken. Indeed, the use of economic injury levels (EILs) is based on the premise that injury is preventable; EILs are not accurate when used where substantial injury has occurred already.[1] Thus, sampling programs must be initiated before significant pest injury can occur but not much before pest populations are established.

Generally, issues involving the timing of sampling programs are less studied than other aspects of sampling. For many situations, timing is a simple question that does not require much effort. For other instances, timing may be a critical consideration. We present some of the general approaches for determining when to begin sampling and discuss some considerations of host, pest, and environment that pertain to this question. Next, we explore some of the techniques associated with initiating sampling programs. In particular, because of its overriding importance to many sampling programs, we discuss in detail measuring thermal development of insects to predict pest occurrence. Finally, we conclude with a discussion of practical considerations in initiating sampling programs.

II. DECIDING WHEN TO BEGIN SAMPLING

A. TIMING AND MONITORING

The two fundamental approaches for determining when to initiate sampling programs are timing and monitoring. Timing refers to activities used to predict the occurrence of pests, independent of direct observations or counts of pests. Monitoring, as we define it, refers to broad, often qualitative, sampling of pests or their effects used to trigger more precise, quantitative sampling programs. Timing includes considerations of seasonality, weather, and especially temperature. Monitoring includes surveys, trapping, and possibly remote sensing. These approaches are not mutually exclusive. For example, the identification of favorable weather systems for insect migration is a timing approach, using pheromone traps to confirm insect migration from such systems is a monitoring approach.

Methods for initiating sampling based on timing are common in regions with strongly variable climates. Probably the most common timing procedure is the simple use of calendar dates to begin sampling. Such an approach often succeeds because of the strong relationships between many insect life histories and seasonal cycles. Similarly, temperature is crucially important in determining insect development times; therefore, using temperature to predict the occurrence of damaging pest species is one of the most important considerations in timing sampling programs for many species.

Because monitoring is a form of sampling, most of the issues and considerations of sampling discussed elsewhere in this book apply. Indeed, some entire sampling programs, such as those used to detect new pest species introductions, represent little more than ongoing monitoring. In the context of initiating sampling programs, monitoring is a low cost, qualitative approach used to trigger more quantitative, and expensive, sampling efforts. Evaluation criteria from monitoring may include pest occurrence, pest impact, estimates of pest density, or the recognition of specific pest forms, such as gregarious phenotypes of grasshopper species or alate aphids. (The occurrence of such forms is an indication of high population levels.) Typically, monitoring is of greatest utility for pests whose occurrence is not predictable from seasonality, weather patterns, or estimates of thermal development. Additionally, monitoring is valuable as a confirmation of predictions made from timing approaches.

Both of these methods for determining when to start a sampling program operate within a universe set by specific attributes and relationships of the pest, its host, and the environment. All pest management activities, including sampling, depend upon the interactions of these three factors.

B. HOST, PEST, AND ENVIRONMENTAL CONSIDERATIONS

Host and pest characteristics have an important bearing on sampling. Host susceptibility to pest attack is the most basic parameter. Usually, sampling is only required when the host is at a stage susceptible to the pest. Typically, seedling and reproductive stages are most severely affected by pest injury, therefore, sampling may be critical at these times.[2] In addition to host phenology, pest life history characteristics, particularly synchrony between injurious life stages of the pest and susceptible stages of the host, are important. Sampling may be initiated when such synchronies arise. For example, phenological delays in plant development may render plants susceptible to pest attack that ordinarily might be avoided.

The environment influences various aspects of host and pest relationships. Water stress frequently alters pest and host phenologies and renders plants more susceptible to pest injury. Indeed, the occurrence of drought may itself be an indicator of the need to monitor pest species such as grasshoppers and spider mites. Another environmental influence is the use of cultural practices which may reduce or increase the likelihood of pest occurrence. For example, crop rotations or certain tillage practices may eliminate the need for sampling some potential pests. In contrast, other practices, such as incorporation of a cover crop or poor weed control may increase pests and trigger sampling efforts.

Interactions between these factors can alter host susceptibility to pest attack, pest population levels, host or pest phenologies, or pest occurrence in a host. If these relationships are sufficiently predictable, the occurrence of a given situation (late season water stress, for instance) may be sufficient to initiate sampling efforts for some species. More commonly, this interplay of host, pest, and environment represents a moving surface upon which timing and monitoring approaches for sampling initiation must contend.

III. TECHNIQUES

A. INITIATION BASED ON LIFE HISTORY AND ENVIRONMENT

1. Calendar Date

Pest life history events and their consistent correlations to calendar dates often provide an indication of when to initiate sampling. The timing of biological events (e.g., oviposition or appearance of first instars) and their fidelity to dates has often been determined through several years of observation. In many instances, the phenological information has been gained without intensive research. Moreover, using calendar dates is one of the most inexpensive techniques for determining when to begin sampling.

In Nebraska, recommendations to begin sampling for second-generation European corn borer, *Ostrinia nubilalis*, are based, in part, on the periodicity of adult flight activity—approximately mid-July each year.[3] For corn rootworm, *Diabrotica* spp., management and sampling for adults are initiated in early July, when adults typically begin to emerge.[3]

In many instances, using calendar dates is the only viable technique available to determine when to begin sampling. Monitoring techniques such as trapping or surveying may be impractical or prohibitively expensive. Although calendar dates may be adequate for predicting seasonality, degree-day accumulations typically are more precise than calendar dates. However, degree-days have not been determined for many pests or for some pest life stages. Therefore, calendar dates often are used because degree-day information is nonexistent or incomplete.

2. Migration

Migration is an important biological phenomenon for many arthropods, including several pest species. Arthropod migration can be defined as an adaptive behavior that results in the displacement of large groups of individuals.[4] In the Noctuidae alone, several important pests migrate very long distances each year and cause significant damage to crop plants in regions where they cannot overwinter. Further, migration within Noctuidae may be more widespread than previously reported.[5]

The characterization of arthropod migration continues to be an active research area. In recent years, the ability to accurately predict both the temporal and spatial aspects of migration has greatly enhanced the capacity to manage these pests. Moreover, arthropod migration events, when predicted or observed, are used to initiate sampling programs.

Insect migrations have been characterized using a number of meteorological techniques.[6] Probably the simplest method is to observe persistent airflows from specific directions. These airflows can indicate that migration events are likely to occur (Figure 1). Migration also can be predicted when appropriate synoptic weather systems occur.[7-9] Synoptic weather systems are atmospheric circulation patterns that influence the weather over an area of 1000 km or greater.[6] Trajectory analysis, the projection of the path of an air parcel, provides a sophisticated technique for predicting the movement of migratory insects that are transported above their typical activity layer.[9,10] In recent years, trajectory models have been developed that are very accurate in projecting migratory movements within specific time intervals.[9-11]

Regardless of the meteorological methodology used, monitoring of individuals usually is needed to verify that migration has occurred. Monitoring of migrants typically is accomplished by trapping or observing individuals on hosts or attraction sites. However, unconventional techniques, such as radar imaging and observation by plane or helicopter, also have been used to identify migrants and to predict infestation locations. Direct observations of insects that migrate over relatively short distances also have been used to initiate sampling. Examples include chinch bug, *Blissus leucopterus leucopterus*, Mormon cricket, *Anabrus simplex*, and true armyworm *Pseudaletia unipuncta*.[4]

In the midwestern U.S., some of the most important insect pests of field crops are migratory species. Species such as the black cutworm, *Agrotis ipsilon*, the green cloverworm, *Plathypena scabra*, and the potato leafhopper, *Empoasca fabae*, cannot overwinter in the upper Midwest. These species recolonize northern areas by migrating from southern overwintering regions each spring. Migrants, or, more commonly, their progeny can significantly injure crops from localized areas to entire regions, depending on the magnitude of the migration. The observation of immigrant green cloverworm moths in Iowa has been used to initiate a time-sequential sampling program developed by Pedigo and van Shaik[12] to determine if subsequent generations will cause economic damage to soybean. Similarly, the observation of potato leafhop-

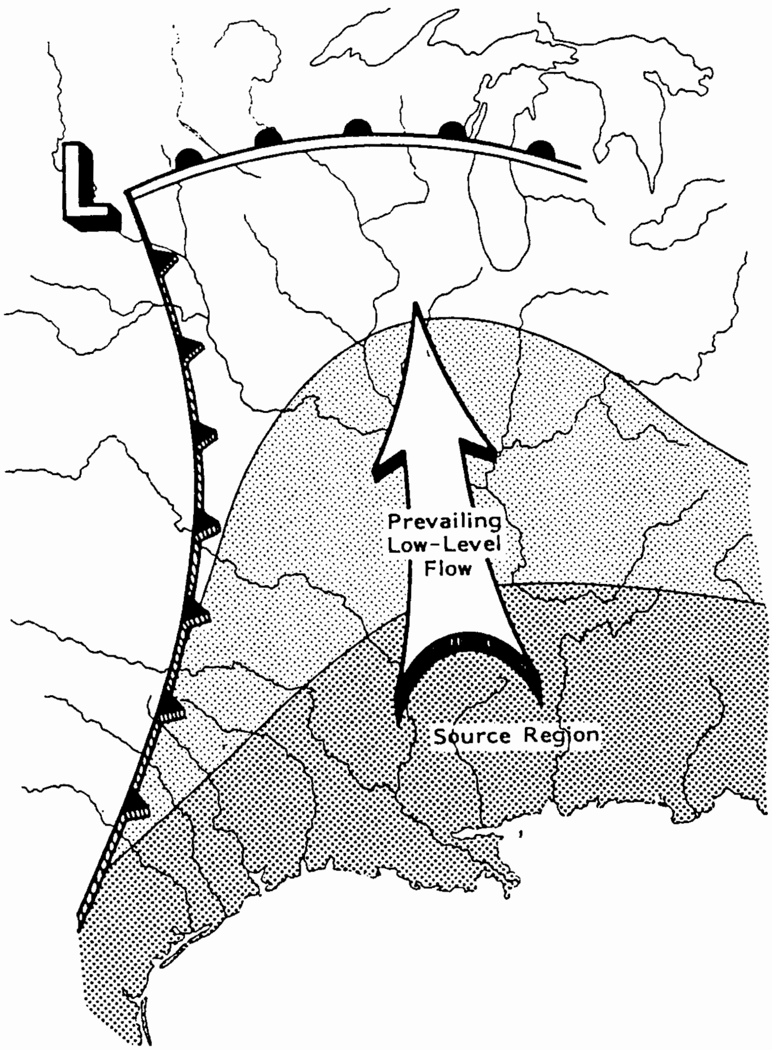

FIGURE 1. A typical weather pattern exhibited during insect migration events in the Spring in the U.S. A northward-moving airflow (arrow) is east of an approaching cold front (line with triangles) and south of a warm front (line with semicircles). Insects are transported to the midwestern and northern U.S. from source regions in the southern U.S. (Reprinted with permission of the Entomological Society of America, from *Bull. Entomol. Soc. Am.*, 34, 9, 1988.)

per immigrants late in the first growth cycle of alfalfa may initiate a sampling program during the second and third growth cycles.[6]

An advanced system of migration and outbreak prediction has been developed for the black cutworm in the midwestern U.S.[6,13] A meteorologically based trajectory model has been used to effectively predict black cutworm outbreak locations. Moreover, the model predicts the magnitude of the outbreaks using a numerical rating system based on trajectory proximity to black cutworm source regions in the southern and southeastern U.S.[6,13] Predictions of black cutworm immigration have made it possible to initiate and intensify sampling at relatively specific locations.

Ongoing research will lead to a better understanding of arthropod migration as an ecological phenomenon and will result in better predictions of migration events. More accurate predictions will then improve decisions to initiate sampling programs for immigrant arthropods.

3. Thermal Development

Because pest management concerns are typically directed at the injurious stage in an insect's life cycle, it is important to predict when this stage will be present. Ideally, sampling will be started at the first presence of the injurious insect. Consequently, it often is necessary to track common developmental events such as emergence or activity after overwintering, oviposition, egg hatch, and larval development. These developmental events may be observed through ongoing monitoring or sometimes predicted based on calendar dates. However, these approaches may be infeasible or inappropriate for many situations.

Insects are poikilothermic organisms; their body temperatures depend upon ambient temperatures. Homeothermic organisms, such as humans and other mammals, also have temperature-dependent development, but by maintaining constant body temperatures the impact of temperature on development is constant. Describing insect development often requires a description of temperature as well as time. This principle of using time and temperature to describe poikilothermic development has been known since the early 1700s,[14] but many fundamental aspects of these relationships are uncertain. Remaining questions include: what is the physical basis for effects of temperature on development, what are appropriate mathematical models for describing temperature development relationships, how does environmental variability, especially fluctuating temperature, influence development, and what are the most accurate methods for estimating development in the field?

Proper timing of sampling programs for many insects depends upon accurate estimates of insect development based on ambient temperatures. Although some questions mentioned above can be neglected in practical applications, others may not. In practical applications, we need only employ that degree of complexity necessary to provide a suitably accurate prediction of development. Estimates of development for timing sampling programs typically need to achieve 85 or 90% accuracy for most insect species. Also, relating temperature and development is of trivial importance if temperatures are constant or developmental events are driven by other factors, such as photoperiod. Thus, establishing temperature-development relationships typically is most important for early season insects in temperate regions, where temperatures are variable and limit development.

There is a large volume of literature on questions of temperature and insect development. We present key issues as they relate to initiating sampling programs and refer to major reviews that can guide the reader to additional information as desired. The points we consider are (1) the physical basis for temperature-development relationships, (2) approaches for estimating development, (3) limitations to existing methods, and (4) using estimates of development to start sampling programs.

a. Why Does Temperature Matter?

This seemingly simple question actually encompasses substantial biological complexity. The basic answer is that growth depends on temperature because biochemical reactions that are essential to growth also depend on temperature. However, the details of how temperature affects physiology and growth is more complicated.

At a macro level, the relationship between temperature and growth rate is well established. As indicated in Figure 2, at low temperatures there is a curvilinear response; at higher temperatures growth rates increase linearly with temperature; at still higher temperatures, the relationship becomes curvilinear again and maximum

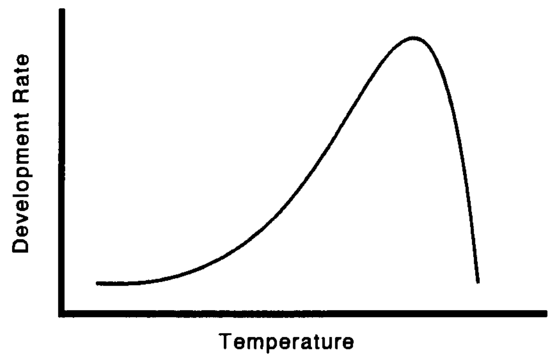

FIGURE 2. The thermal development curve—the generalized relationship between temperature and rate of development.

rates are achieved; and finally, at temperature beyond the optimum, growth rates decrease precipitously. Usually, temperatures at the upper limit of development are near the lethal temperatures for a species.

Why the temperature development curve has this shape is more problematic. Undoubtedly, the extremes of the curve represent situations in which temperature restrains development. At high temperatures, respiration may be increased, potential dessication may be greater, and enzyme activities may be diminished. At low temperatures, membrane permeabilities may be reduced, insufficient heat may be available for ready formation of enzyme-substrate complexes, and, again, enzyme activities may be suboptimal.

One explanation suggested for the shape of the low temperature portion of the curve is that nonlinearity is a reflection of genetic differences between individuals and that this, coupled with low temperature mortality, produces an apparently nonlinear relationship.[15] Lamb[16] addressed this question directly by examining development rates of pea aphid, *Acyrthosiphon pisum*, clones at low temperature. Lamb demonstrated that curvilinear responses between temperature and development rates are not a function of genetic differences. Instead, temperature and development rates have a nonlinear relationship at low temperatures, which seems to be a reflection of the intrinsic physiology of development.

A simple model for understanding temperature and growth begins by recognizing that growth requires the production of new cellular material. (Strictly speaking, growth involves differentiation and cell enlargement, as well as the production of new cells, but these other aspects of growth follow from increases in cellular material.) Production of new materials depends, in turn, on synthetic biochemical reactions. These reactions require heat; they are temperature dependent. Besides heat of activation, rates of diffusion for enzymes and substrates also depend on temperature, as may membrane permeabilities, which also influence substrate and enzyme availability. Temperature also may influence metabolic rates and partitioning of resources between growth and maintenance. In addition to temperature, any factor that influences substrate availability or enzyme availability and function will have a corresponding influence on development rates. This observation helps explain why nutrition, photoperiod, stress conditions, and similar factors all influence development rates.

Some approaches to modeling thermal development are based on the assumption that one or more enzyme-catalyzed reactions are "rate limiting" for growth,[17] which tends to ignore the impact of temperature on other factors such as membrane function. This notion of rate-limiting reactions has provided a loose physical explanation for some models of insect development. More sophisticated explanations have been proposed and are the basis for some influential approaches to modeling thermal development. Sharpe and DeMichele[18] proposed a biophysical model for poikilothermic development based on the premise that reversible inactivation of control enzymes occurs at the upper and lower temperature limits for a species. This model was subsequently reparameterized to improve its ease of fit and other statistical properties.[19]

The Sharpe and DeMichele model has been very influential and widely used to describe development.[20] Adoption of the Sharpe and DeMichele model did not arise out of ease of use, because determining parameters for the model is a somewhat involved procedure (although a computer program has been published for making these estimates[20]). Instead, its appeal probably derives from its theoretical basis in reflecting a biochemical mechanism for describing development. Unfortunately, as Lamb[16] has pointed out, the biophysical basis of the Sharpe and DeMichele model is unfounded. The premise of the Sharpe and DeMichele model is that upper and lower temperature control enzymes regulate development through reversible thermal inactivation. However, Lowry and Ratkowsky[21] showed that this assumption is not true for some bacterial strains. Hilbert and Logan[22] argued that describing development through a rate-controlling enzyme ignores the important influence of temperature on phase changes in cuticular waxes. Also, Lamb et al.[23] pointed out that different life stages of insects have different developmental rate parameters, which would not be the case were a single rate-controlling enzyme operating. There is no direct evidence that specific enzymes regulate developmental rates at temperature extremes. Consequently, although Sharpe and DeMichele's model may provide a good empirical fit to some data, it does not reflect an underlying biochemical reality, as some of its users propose.[20]

A further complication to our understanding of how temperature affects development arises when we consider development under fluctuating temperatures. Typically, development is faster for fluctuating temperatures around a low temperature mean than under constant conditions at the same mean temperature. In contrast, development is slower for fluctuating temperatures around a high temperature mean than under constant conditions at that temperature.[24,25] Why this is so, involves mathematical, and possibly, physiological explanations.

The mathematical aspect is a reflection of the shape of the temperature-development curve (Figure 2) in what is called the rate summation effect.[25] Tanigoshi et al.[24] provide a lucid explanation of the effect; essentially, fluctuations around a low temperature mean will have the effect of producing rapid acceleration in development rates because of the concave shape of the curve. (Figure 3 presents this effect graphically.) In the low temperature portion of the development curve, the magnitude of the decrease in rates with temperature fluctuations below the mean is much less than the magnitude of the increase in rates associated with fluctuations above the mean. Therefore, temperature fluctuations above a given mean will increase development more than comparable fluctuations below a mean decrease development. The reverse is true for the convex, higher temperature portion of the development curve. On the linear portion of the curve, fluctuations above and below a mean temperature cause equally offsetting changes in development.

The other possible explanation for changes in development rate is that temperature fluctuations may cause physiological changes that stimulate or retard development. Although various mechanisms have been suggested, after reviewing the available evidence, Worner[25] concluded that present experimental findings could reflect

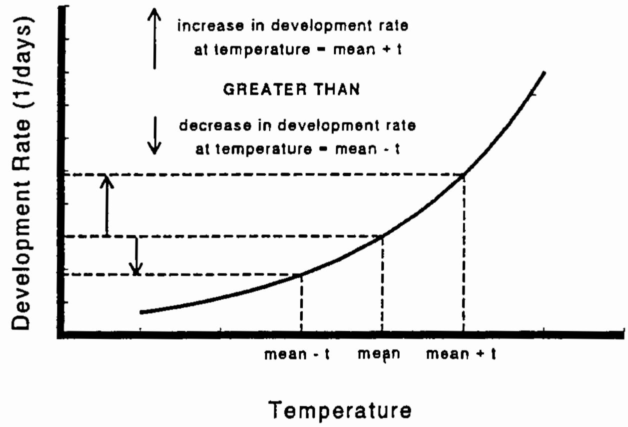

FIGURE 3. The rate summation effect at low temperatures. Temperature fluctuations (t) above and below a given mean produce a development rate greater than that predicted by the mean temperature, because fluctuations above the mean increase development rates proportionally more than fluctuations below the mean reduce development rates.

inadequate developmental models, currently unknown physiological mechanisms, or a combination of both factors.

As this discussion illustrates, we cannot as yet describe in a detailed, physiological sense how temperature influences development. Temperature-development curves have been described for many insect species, albeit based on constant temperature development. Undoubtedly, better physiological understanding of how temperature influences development would offer the prospect of improved models for thermal development. However, the absence of such understanding is not an impediment to developing acceptable development models for use in sampling.

b. Approaches for Estimating Development

Three basic methods have been used for modeling development: physiologically based models; degree-day, or linear, models; and curvilinear models. The one example of a physiologically based model is that of Sharpe and DeMichele,[18] but as we pointed out, its physiological basis is not well supported. Consequently, the Sharpe and DeMichele model probably is better characterized as a curvilinear model. Both the linear and curvilinear approaches are descriptive rather than explanatory. They are mathematical expressions of the temperature-development curve.

Degree-day, or linear summation, models are the most widely used for insect sampling. They have the virtues of being easily developed and easily used. They have the failing of being obviously incorrect. The temperature-development curve clearly is a curve, rather than a straight line. Because the degree-day approach is based on a linear relationship between temperature and development, the lower and upper portions of the development curve are discounted. However, this inaccuracy may not matter if daily temperatures do not often enter the curvilinear portions of the curve. Additionally, the degree of curvilinearity differs among species. Often degree-day models provide sufficient accuracy for practical uses in timing sampling. As we will discuss in more detail, weighing complexity and biological accuracy against simplicity and practical acceptability is an important issue in modeling thermal development, as well as other approaches for timing sampling programs.

Degree-days (also called heat units, thermal units, or growing degree-days) represent a measure of physiological time through a combination of time and temperature.

Degree-days are the number of degrees above the minimum temperature acceptable for growth multiplied by time in days. Ten degrees above the minimum for 5 d represents 50 degree-days (10° × 5 d) just as does 2° above the minimum for 25 d (2° × 25 d). Both instances are taken to represent the same amount of physiological time; an insect would have grown the same amount under either circumstance.

Using degree-days to estimate development requires that a minimum developmental threshold, the lowest temperature at which development will proceed, be determined. Additionally, the total degree-day requirements for specific life history events (such as egg hatch, larval development, pupation, adult emergence, and preoviposition period) must be calculated. For some species, a maximum developmental threshold also may be determined.

At least three important approximations are associated with degree-days. First, degree-days are based on constant relationships between temperature and development, in other words, linearity. Second, degree-days do not address changes in development associated with fluctuating temperature. Third, degree-days use an estimate for the minimum developmental threshold. The first two of these approximates are intrinsic to the technique. However, approaches for improving estimates of the minimum developmental threshold for a species are possible. Commonly, a minimum threshold is established by extending the linear portion of the development curve to where development is zero (the *x*-intercept method).[26] Another approach is to iteratively test thresholds and identify that threshold producing the least variability in estimates of development times.[5,26] Lamb[16] proposed a new procedure based on relationships between low temperature development and maximum development. He presents evidence indicating that a minimum developmental threshold can be set as that temperature where development is 8% of the maximum rate, which both improves the estimate and avoids conventional difficulties in determining threshold temperatures.

The practical use of degree-days involves making estimates of daily temperature accumulations. These represent the degrees above the minimum threshold and below the daily temperature cycle. In practice, the actual daily temperature cycle rarely is available; instead, it is approximated based on maximum and minimum temperatures. Higley et al.[27] summarize approaches for estimating temperature accumulations and limitations to their use. In brief, daily accumulations may be calculated as a rectangle (average temperature-minimum developmental threshold) or as the area under a sine wave and above the minimum developmental threshold. Although sine wave estimates frequently are held to be more accurate, both approaches involve assumptions about the shape of daily temperature cycles that are not always valid (particularly when the movement of weather fronts produces rapid temperature shifts).[27]

Many nonlinear models have been developed to describe thermal development, and Wagner et al.[20] reviewed most of these approaches. Other than the Sharpe and DeMichele model, no individual model has been widely adopted. The key problems with curvilinear models are the substantial research required for their development and greater complexity in use. Also, the validity of any given model tends to vary depending upon the individual species being modeled. Nevertheless, curvilinear models clearly represent a more accurate representation of thermal development curve than linear approaches.

Models that reflect the influence of fluctuating temperatures on development represent an extension of basic curvilinear models. Hagstrum and Milliken[28] summarize efforts to develop such models and present their own method for describing development under fluctuating temperatures. Worner[25] presents an insightful, comprehensive discussion on the problems of modeling development under fluctuating temperature regimes. Her analysis suggests that existing approaches based on laboratory and field data are largely inadequate. She concludes that more research is needed, particularly with field data under extreme climate conditions for a species, before accurate predictions of development under fluctuating temperatures is likely to be possible.

c. Limitations

The most obvious limitations in techniques for predicting development relate to their biological validity. None of the techniques, including more advanced approaches, provides a definitive, accurate description of development across most environmental conditions. However, models do differ in their accuracy, although absolute accuracy is not a requirement for initiating sampling programs. Beyond the theoretical validity of any given model, a number of additional factors will limit the accuracy of virtually all models. Indeed, variability from extrinsic factors may overwhelm intrinsic differences between models.

Higley et al.[27] discuss many of these limitations. Among these are effects on development of insect nutrition, availability of water, and hormonal regulation of development (modified by mechanisms of diapause and physiological responses to stimuli such as photoperiod).[29] Approximations in laboratory estimates of development, particularly constant temperature estimates, and estimates of developmental thresholds for degree-days are other potential sources of error. Also, development curves and thresholds differ among the life stages of an insect species, but most often development curves are averaged across life stages for ease of use. This is one aspect of thermal development where simulation models are useful to provide stage-specific development estimates.

Probably the most important limitation to all types of thermal development estimates are the temperature data used to drive those estimates. Short of directly measuring insect body temperatures, field estimates of ambient temperature only provide a gross picture of the actual thermal environment an insect experiences. Baker et al.[30] provide a valuable discussion of the many factors producing discrepancies between weather station temperature data and actual temperatures in the field. These factors include observation time, latitude, surface structure, topography, and urbanization. Additionally, many insect species can actively modify their body temperatures through thermoregulation.[31]

Some limitations may preclude the use of predictions of thermal development for a species. More often, these factors add to variability in developmental estimates. Consequently, accuracy in predictions of thermal development is a function not only of the choice of estimation technique, but also of the extrinsic circumstances in which an estimate is made.

d. Using Estimates of Thermal Development in Sampling

Predictions of pest occurrence based on thermal development usually are of greatest value where temperature limits development. In temperate regions, spring development of insect pests is a common situation where thermal estimates of development are of value. Just as such estimates are needed for initiating sampling activities, so must estimates for thermal development be initiated. Typically, degree-day accumulations are started by calendar date, but specific incidents, such as oviposition or a migration event, also may start thermal development estimates.

Choice of a specific estimation technique reflects a consideration of acceptable accuracy weighed against increasing complexity. The great virtue of the degree-day technique over others is its simplicity of calculation and use. Despite its many theoretical failings, the degree-day approach may be the method of choice, if it can provide sufficient accuracy for a given pest situation. However, degree-days do not work with many species and for these curvilinear models are a necessity. Similarly, the need to consider fluctuating temperatures will differ among species and environments.

Often, complexity of a developmental model mistakenly is assumed to provide greater accuracy. For example, the assumption that simulation models provide better estimates of development than mathematical models is based on a misconception. Actually, simulation models do not represent an alternative to degree-day or curvilinear approaches. Instead, such models incorporate degree-day or curvilinear approaches within the simulation (although simulation models do allow for the use of stage-specific developmental thresholds). Even between degree-day and curvilinear approaches to modeling development, increasing complexity of the model is of itself a poor guide to the accuracy of the model. For instance, degree-day models have proved as good or better predictors than curvilinear models in a number of instances.[32-34] Occam's razor (the principle that given two explanations that fit the data the simple should be chosen over the complex) clearly applies to models of insect development. Consequently, the guiding principle in selecting models to describe insect development is to begin with the simplest model or procedure possible and only add complexity as a specific pest situation requires.

B. INITIATION BASED ON ONGOING MONITORING

1. Hosts

Monitoring of arthropods on their hosts is a direct and commonly practiced technique to determine if sampling should begin. The level of resources devoted to monitoring the host often is related to its economic value and the type of feeding injury. For example, apple usually is monitored more intensively than soybean.

Host monitoring can occur throughout the season, or during a short time interval, depending on pest biology and host susceptibility to injury. In the midwestern U.S., alfalfa is monitored for alfalfa weevil larvae, *Hypera postica*, only during the first growth cycle, and possibly during the early regrowth of the second cycle because adults or larvae are not injurious on subsequent cuttings. Sorghum is monitored for greenbug in Nebraska to indicate arrival of migrants and to indicate time of sampling for assessing mid-to-late season populations.[3] Soybean often is monitored throughout the growing season in the southern U.S. because of the ubiquitous presence of several pest species.

Occasionally, only specific hosts or areas of fields need to be monitored. For example, first generation European corn borer moths initially prefer to oviposit on the tallest corn plants in an area; therefore, monitoring for egg masses may be limited to fields where the corn stand is taller than surrounding areas of the field or surrounding fields.[3] Similarly, corn plants that have green silks may be monitored for second generation European corn borer egg masses because females prefer to oviposit on these plants. Grasshoppers initially feed in weedy, grassy field margins, so monitoring this type of field margin is a common technique to determine if sampling should begin.[35]

Several examples have been provided regarding monitoring of insects on plant hosts. However, host monitoring is not limited to plant hosts. Indeed, pests such as the stable fly, *Stomoxys calcitrans*, face fly, *Musca autumnalis*, and horn fly, *Haematobia irritans*, are observed on cattle throughout the summer. In some cases, medical pests, such as various species of ticks, tsetse flies, *Glossina* spp., and mosquitoes have been monitored on human hosts.[36]

2. Alternate Hosts and Attraction Sites

Many arthropod species use alternate hosts or attraction sites for one or more activities, such as resting, mating, oviposition, or feeding. Consequently, the presence of a species on an alternate host or attraction site may initiate sampling on the primary host. Moreover, the technique for monitoring the alternate host may be similar to monitoring the primary host.

In the spring, newly eclosed European corn borer adults are attracted to grassy or weedy areas at field margins. These areas, termed action sites, have been used to provide an indication that oviposition on corn will soon begin; therefore, moths at action sites indicate that sampling of corn should begin for first-generation egg masses, first instars, or first instar feeding injury.[3] Black cutworm moths prefer to oviposit in low, wet portions of fields that are weedy or grassy. Developing larvae cut crop stalks in these areas or move to crop plants after the weeds have been destroyed by cultivation or herbicides. Monitoring of these areas is important for sampling and management programs.

Chinch bugs overwinter in attractive sites, and feed on alternate hosts, such as wheat, oat, rye, and barley before migrating to corn and sorghum, where they are most injurious and economically damaging. The observation of chinch bugs in alternate host areas (e.g., in wheat fields adjacent to corn or sorghum fields) may initiate sampling to estimate potential economic populations.[3]

3. Trapping

Traps are used to provide direct evidence of the presence of pest species in a specific area. Additionally, the number of individuals captured may indicate the likelihood that an economic population currently exists or may exist during the season. Specimens captured in traps may indicate the emergence of adults or immatures from overwintering sites, the emergence of adults from immature stages, or the immigration of individuals from other regions.

Traps used to capture arthropods generally are left unattended and specimens are collected by humans at specific time intervals.[4] Because they do not require continuous human labor to operate, they usually provide a very cost-efficient technique for arthropod monitoring. Traps vary greatly in size, shape, and especially in mode of collection. In general, traps are divided into two main categories: those that incidentally capture individuals, and those that attract and then capture individuals.[36] Some traps, however, both attract and randomly capture individuals, so the delineation between the two types can be obscure. Traps that randomly capture arthropods include Malaise, window, cone, and pitfall. Traps that attract and then capture arthropods include light, pattern and/or color, Manitoba, shelter, carbon dioxide, tethered animal, boxed animal, bait, kairomone, and pheromone. Traps that both randomly capture and attract arthropods include sticky, color, and water. Detailed descriptions of arthropod traps are provided by Southwood[36] and in Chapter 5 of this book.

The different trapping mechanisms vary in their ability to attract specific arthropod species. For example, most conventional light traps attract many night-flying insects from several orders. Indeed, light trap receptacles can become so cluttered with specimens that identification of the species of interest may be difficult. Conversely, some traps use pheromone lures that are specific to only one species and one sex.

The presence of individuals in traps can lead to the decision to initiate sampling. The decision to begin sampling for several noctuid pests is dependent on their presence in light traps. Pheromone traps are used to assist in the decision to begin sampling or to use a management tactic for pests such as European corn borer, codling moth, *Cydia pomonella*, Mediterranean fruit fly, *Ceratitis capitata*, oriental fruit fly, *Dacus dorsalis*, melon fly, *Dacus curcubitae*, and corn earworm, *Helicoverpa zea*.[4]

4. Surveys

Surveys and surveillance programs usually are organized programs with detailed, established protocols for monitoring arthropods.[4] The programs may either be intensive or extensive, but, at the very least, they rely on established protocols. Federal, state, and private organizations conduct surveillance programs to identify arthropods, and sometimes to quantify pest populations. At the federal level in the U.S., the USDA, through APHIS and PPQ, detects agricultural pests introduced into the U.S., determines the spread of introduced pests into new regions, and determines the abundance of established pests.[4] Individual states also conduct pest surveys, using state entomologists and Cooperative Extension Service specialists. Private consulting companies also engage in survey activities that identify important pests.

Surveillance programs often use methods such as monitoring hosts, monitoring alternate hosts, monitoring attraction sites, and trapping (described above). Arthropods surveyed in the U.S. include gypsy moth, *Lymantria dispar*, Japanese beetle, *Popillia japonica*, *Ixodes damini*, and the Asian tiger mosquito, *Aedes albopictus*. After a pest, whether introduced or indigenous, is identified in a surveillance program, decisions typically are made to begin more formalized and thorough sampling, or to initiate a management tactic.

5. Remote Sensing

Remote sensing in entomology is a relatively new field, initiated by advances in radar, satellite, and optical technologies. Remote sensing, in a broad definition, is the "observation of a target by a device some distance from it".[37] The uses for remote sensing in entomology include the direct observation of insects, the detection of insect-induced injury to plants, and the characterization of environmental factors that affect insects. Remote sensing techniques include photography, videography, satellite photography, multispectral scanning, thermal imaging, radar, and acoustic sounding. Radar imaging, photography, and videography for entomological applications have been conducted both from the ground and from aircraft. Riley[38] provides an extensive review of remote sensing techniques used in entomology.

Remote sensing techniques have yet to be used extensively for determining favorable conditions to begin sampling. However, for many years, forest entomologists have used remote sensing methods to rapidly identify stands of trees injured by several insect pests, such as hemlock looper, *Lambdina fiscellaria fiscellaria*, spruce budworm, *Choristoneura fumiferana*, bark beetles, *Dendroctonus* spp., gypsy moth, and forest tent caterpillar, *Malacosoma disstria*.[38] After injured trees are discovered by remote sensing techniques, sampling then can be initiated to determine if management actions are necessary.[39] Additionally, plants that experience significant abiotic stress, such as drought stress, may be detected by optical and thermal imaging. Because host plants typically are more susceptible to insect injury when stressed by other abiotic and biotic factors, early detection of these conditions with remote sensing techniques may indicate where and when sampling for pests should begin.

In fruit orchards, aerial remote sensing has been used to indirectly detect infestations of insects by identifying injured trees or products produced by insects, such as honeydew.[38] For example, infestations by the European red mite, *Panonychus ulmi*, were identified in peach orchards because low-altitude infrared photographs detected differences between normal and mite-induced chlorotic leaves.[40] Defoliation of pecan by the walnut caterpillar, *Datana integerrima*, has been detected by infrared techniques.[41] In citrus groves, infestations of brown soft scale, *Coccus hesperium*, were detected with aerial photography from the blackened foliage caused by a sooty mold fungus that uses honeydew as a growth medium.[42]

In field crop agroecosystems, the effects of insects have been detected successfully by aerial remote sensing using color and infrared film. Infestations of corn leaf aphids, *Rhopalosiphum maidis*, and sweetpotato whiteflies, *Bemisia tabaci*, have been identified by the photographic detection of sooty mold on leaves.[43,44]

Environmental factors that influence insect pest abundance and location can be detected by aerial and satellite remote sensing techniques. This has been especially useful in detecting short-lived environmental characteristics that dramatically influence pests. For example, both satellite and aerial techniques have been used to identify ephemeral vegetation used by the desert locust, *Shistocerca gregaria*, in Africa.[38] Mosquito breeding habitats in Africa and the U.S. have been readily identified using aerial photography.[45,46] Rainstorms that are responsible for the concentration and deposition of African armyworm adults, *Spodoptera exempta*, have been detected using thermal imaging techniques from satellite systems.[47]

Direct observation of insects with remote sensing has been successful primarily because of advances in radar technology.[38] Migrating moths, locusts, aphids, planthoppers, and leafhoppers have been observed using radar systems. Although radar imaging of migrating insects has not been used to determine when and where to begin sampling, remote sensing of frontal systems responsible for the transport of migrating pests has been used in this manner.

Remote sensing offers tremendous potential for pest management programs. Specifically, by remotely sensing insect activity, the effects of insects, and the environmental factors that affect insects, it is possible to determine when and where to begin sampling. An overriding consideration regarding the use of remote sensing to initiate sampling is that evidence of insect activity or favorable environmental factors is necessary before economic injury occurs.

Remote sensing technology is progressing at a rapid rate.[38] Currently, most remote sensing techniques are relatively expensive, precluding substantive use on all but the most devastating insect pests. Moreover, satellite-based systems have not, as yet, proven as beneficial as aerial-based systems for entomological uses.[38] Advances in optical, thermal, and audio technology undoubtedly will improve the precision and cost-effectiveness of remote sensing systems used for entomological purposes.

C. INITIATION BASED ON PEST IMPACT

Using symptoms of pest impact to trigger sampling and other pest management actions commonly occurs, particularly with occasional pest species. Because the occurrence of such pests is unexpected, monitoring programs for injury often are likely to be the only proactive procedures used. The risk in using injury symptoms to initiate sampling is the prospect that excessive injury will occur before a management decision is made. Therefore, successfully initiating sampling based on pest impact is largely a function of injury rates. If the development of injury is slow, initiating sampling based on symptoms of injury may be acceptable but not if the injury rate is rapid.

In Europe, spruce bark beetle, *Ips typographus*, infestations initially are identified based on symptoms of injured trees. The beetle vectors the fungal pathogen, *Ceratocystis polonica*, which may cause substantial tree mortality. Stands of discolored trees that are dropping their needles and/or the presence of dead trees may indicate an outbreak population. Identifications of injured trees lead to intense sampling activities over a larger area to determine the extent of the outbreak.

For insect larvae, injury rates (consumption rates) are closely tied to development rates. Additionally, injury rates for young larvae are much lower than for later stages.

For example, more than 90% of injury by most defoliating caterpillar species occurs in the last two larval stages.[48] Consequently, monitoring injury or numbers of young larvae can be successful because relatively little injury occurs at this stage. However, environmental conditions that rapidly accelerate development (warm temperatures) or extremely high numbers of insects, even young larvae, dramatically increase injury growth rates. A further potential problem is that young larvae and their injury may not be readily apparent in a field. Some insect behaviors may help conceal both insects and their injury. For example, yellow woollybear, *Spilosoma virginica*, larvae are gregarious in early stages, so their presence is more difficult to detect than if they were widely dispersed.[49] Older, more injurious, larvae rapidly disperse, and sampling must be initiated before this dispersion occurs.

Direct measures of pest impact on a host of itself may be valuable in triggering sampling or pest management action. For example, a key impact of insect defoliation of soybean is to reduce the efficiency of canopy light interception.[50] Consequently, monitoring light interception or canopy leaf area development (which is highly correlated with light interception) would provide a mechanism for identifying when sampling for defoliators might be initiated. Monitoring light interception or canopy development has the potential virtues of relating more directly to potential yield losses then insect numbers and of potentially being an inexpensive form of monitoring (because instrumentation, including some remote sensing procedures, can be substituted for labor).[50]

IV. PRACTICAL CONSIDERATIONS

Choices between timing and monitoring approaches, and between individual techniques, depend upon the specific host, pest, and environmental conditions. Certain hosts or pests will necessitate a given procedure. Other host and pest combinations may present more flexibility.

As is true in all sampling, the more accuracy demanded, the greater the cost. Consequently, the guiding principle for initiating sampling programs is to employ only that degree of effort necessary to start sampling before pest injury becomes unacceptable. The key question for initiating sampling programs is how much accuracy is needed? This in turn directly depends upon how susceptible the crop is to pest attack. Susceptibility is a function of the value of the host and the injuriousness of the pest. Less accuracy is needed for lower value hosts and for less injurious pests. Additionally, sampling is conducted amidst considerable background variability: variability in environmental conditions, particularly weather, variability in host phenology, and, most importantly, variability in pest occurrence and numbers. Therefore, whatever approach is chosen, the approach must accommodate this natural variation. Fortunately, timing and monitoring do not need to provide precise estimates of pest densities, that is, the function of the sampling programs they initiate. Consequently, approaches for starting sampling programs tend to be more forgiving and less quantitative than other features of sampling.

REFERENCES

1. Pedigo, L. P., Hutchins, S. H., and Higley, L. G., Economic injury levels in theory and practice, *Annu. Rev. Entomol.*, 31, 341, 1986.
2. Funderburk, J. E. and Higley, L. G., Managing arthropod pests, in *Sustainable Agriculture: The New Conventional Agriculture,* Hatfield, J. L. and Karlen, D. L., Ed., CRC Press, Boca Raton, FL, 1993.
3. Wright, R. J., Danielson, S. D., Witkowski, J. F., Hein, G. L., Campbell, J. B., Jarvi, K. J., Seymour,

R. C., and Kalisch, J. A., Insect Management Guide for Nebraska Corn and Sorghum, Nebraska Cooperative Extension Service EC 92-1509, Lincoln, 1992.

4. Pedigo, L. P., *Entomology and Pest Management*, Macmillan, New York, 1989.
5. Peterson, R. K. D., Higley, L. G., and Bailey, W. C., Phenology of the adult celery looper, *Syngrapha falcifera* (Lepidoptera: Noctuidae), in Iowa: evidence for migration, *Environ. Entomol.*, 17, 679, 1988.
6. Hutchins, S. H., Smelser, R. B., and Pedigo, L. P., Insect migration: atmospheric modeling and industrial application of an ecological phenomenon, *Bull. Entomol. Soc. Am.*, 34, 9, 1988.
7. Pienkowski, R. L. and Medler, J. T., Synoptic weather conditions associated with long range movement of potato leafhopper, *Empoasca fabae*, into Wisconsin, *Ann. Entomol. Soc. Am.*, 57, 588, 1964.
8. Shaw, M. W. and Hurst, G. W., A minor immigration of the diamond-back moth, *Plutella xylostella* (L.) (*maculipennis* Curtis), *Agric. Meteorol.*, 6, 125, 1969.
9. Domino, R. P., Showers, W. B., Taylor, S. E., and Shaw, R. H., Spring weather pattern associated with suspected black cutworm (Lepidoptera: Noctuidae) introduction to Iowa, *Environ. Entomol.*, 12, 1863, 1983.
10. Wolf, R. A., Pedigo, L. P., Shaw, R. H., and Newsom, L. D., Migration/transport of the green cloverworm, *Plathypena scabra* (F.) (Lepidoptera: Noctuidae), into Iowa as determined by synoptic-scale weather patterns, *Environ. Entomol.*, 16, 1169, 1987.
11. Reap, R. M., Techniques Development Laboratory Three Dimensional Trajectory Model, Technical Procedures Bulletin 255, U.S. Department of Commerce, NOAA, National Weather Service, Washington, D.C., 1978.
12. Pedigo, L. P. and van Schaik, J. W., Time-sequential sampling: a new use of the sequential probability ratio test for pest management decisions, *Bull. Entomol. Soc. Am.*, 30, 588, 1984.
13. Smelser, R. B., The Use of Trajectory Analysis to Project the Long-Range Migration of the Black Cutworm Moth into the Midwest, M.S. thesis, Iowa State University, Ames, 1986.
14. Reamur, R. A. F. de, Observation du thermomètre, faites à Paris pendant l'année 1735, comparees avec cells qui ont été faites sous là ligne à Isle de France, à Alger et an quelque-unes de nos isles de l'Amerique, *Mem. Acad. Sci., Paris*, 545, 1735.
15. Campbell, A. B., Fraser, B. D., Gilbert, N., Gutierrez, A. P., and Mackauer, M., Temperature requirements of some aphids and their parasites, *J. Appl. Ecol.*, 11, 431, 1974.
16. Lamb, R. J., Development rate of *Acyrthosiphon pisum* (Homoptera: Aphididae) at low temperatures: implications for estimating rate parameters for insects, *Environ. Entomol.*, 21, 10, 1992.
17. Barnes, T., *Textbook of General Physiology*, P. Blakiston & Sons, London, 1937.
18. Sharpe, J. H. and DeMichele, D. W., Reaction kinetics of poikilotherm development, *J. Theor. Biol.*, 64, 649, 1977.
19. Schoolfield, R. M., Sharpe, P. J. H., and Magnuson, C. E., Nonlinear regression of biological temperature-dependent rate models based on absolute reaction-rate theory, *J. Theor. Biol.*, 88, 719, 1981.
20. Wagner, T. L., Wu, H., Sharpe, P. J. H., Schoolfield, R. M., and Coulson, R. N., Modeling insect development rates: a literature review and application of a biophysical model, *Ann. Entomol. Soc. Am.*, 77, 208, 1984.
21. Lowry, R. K. and Ratkowsky, D. A., A note on models of poikilotherm development, *J. Theor. Biol.*, 105, 453, 1983.
22. Hilbert, D. W. and Logan, J. A., Empirical model of nymphal development for the migratory grasshopper, *Melanoplus sanguinipes* (Orthoptera: Acrididae), *Environ. Entomol.*, 12, 1, 1983.
23. Lamb, R. J., Gerber, G. H., and Atkinson, G. F., Comparison of development rate curves applied to egg hatching data of *Entomoscelis americana* Brown (Coleoptera: Chrysomelidae), *Environ. Entomol.*, 13, 868, 1984.
24. Tanigoshi, L. K., Hoyt, S. C., Browne, R. W., and Logan, J. A., Influence of temperature on population increase of *Tetranychus mcdanieli* (Acarina: Tetranychidae), *Ann. Entomol. Soc. Am.*, 69, 712, 1976.
25. Worner, S. P., Performance of phenological models under variable temperature regimes: consequences of the Kaufmann or rate summation effect, *Environ. Entomol.*, 21, 689, 1992.
26. Arnold, C. Y., The determination and significance of the base temperature in a linear heat unit system, *Proc. Am. Soc. Hortic. Sci.*, 74, 430, 1959.
27. Higley, L. G., Pedigo, L. P., and Ostlie, K. R., DEGDAY: a program for calculating degree-days, and assumptions behind the degree-day approach, *Environ. Entomol.*, 15, 999, 1986.
28. Hagstrum, D. W. and Milliken, G. A., Modeling differences in insect development times between constant and fluctuating temperatures, *Ann. Entomol. Soc. Am.*, 84, 369, 1991.
29. Tauber, M. J., Tauber, C. A., and Masaki, S., *Seasonal Adaptations of Insects*, Oxford University Press, New York, 1986.
30. Baker, D. G., Kuehnast, E. L., and Zandlo, J. A., Climate of Minnesota. Part XV. Normal temperatures (1951–80) and their application, *Univ. Minnesota Agric. Exp. Stn.*, AD-SB-2777.

31. May, M. L., Insect thermoregulation, *Annu. Rev. Entomol.*, 24, 313, 1979.
32. Hochberg, M. E., Pickering, J., and Getz, W. M., Evaluation of phenology models using field data: case study for the pea aphid, *Acyrthosiphon pisum*, and the blue alfalfa aphid, *Acyrthosiphon kondoi* (Homoptera: Aphididae), *Environ. Entomol.*, 15, 227, 1986.
33. Stinner, R. E., Rock, G. C., and Bacheler, J. E., Tufted apple budmoth (Lepidoptera: Tortricidae): simulation of postdiapause development and prediction of spring adult emergence in North Carolina, *Environ. Entomol.*, 17, 271, 1988.
34. McClain, D. C., Rock, G. C., and Stinner, R. E., San Jose scale (Homoptera: Diaspididae): simulation of seasonal phenology in North Carolina orchards, *Environ. Entomol.*, 19, 916, 1990.
35. Danielson, S. D., Wright, R. J., Witkowski, J. F., Hein, G. L., Campbell, J. B., Jarvi, K. J., Seymour, R. C., and Kalisch, J. A., Insect Management Guide for Nebraska Alfalfa, Soybeans, Wheat, Range, and Pasture, Nebraska Cooperative Extension Service EC 92-1511, Lincoln, 1992.
36. Southwood, T. R. E., *Ecological Methods with Special Reference to the Study of Insect Populations*, 2nd ed., Chapman & Hall, New York, 1978.
37. Barrett, E. C. and Curtis, L. F., *Introduction to Environmental Remote Sensing*, 2nd ed., Chapman & Hall, New York, 1982.
38. Riley, J. R., Remote sensing in entomology, *Annu. Rev. Entomol.*, 34, 247, 1989.
39. Ciesla, W. M., Operational remote sensing for forest insect and disease management—some challenges and opportunities, *Proc. Remote Sensing Nat. Resour., Moscow, Idaho, 1979,* 1980, 39.
40. Payne, J. A., Hart, W. G., Davis, M. R., Jones, L. S., Weaver, D. J. et al., Detection of peach and pecan pests and disease with color infrared aerial photography, *Proc. Bienn. Workshop Color Aerial Photogr. Plant Sci.*, Gainesville, FL, 1971, 216.
41. Harris, M. K., Hart, W. G., Davis, M. R., Ingle, S. J., and Van Cleave, H. W., Aerial photographs show caterpillar infestation, *Pecan Q.*, 10, 12, 1978.
42. Hart, W. G. and Ingle, S. J., Detection of arthropod activity on citrus foliage with aerial infrared color photography, *Proc. Workshop Aerial Color Photogr. Plant Sci.*, Gainesville, FL, 1969, 85.
43. Wallen, V. R., Jackson, H. R., and MacDiarmid, S. W., Remote sensing of corn aphid infestation, 1974 (Hempitera: Aphididae), *Can. Entomol.*, 108, 751, 1976.
44. Nuessly, G. S., Meyerdirk, D. E., Hart, W. G., and Davis, M. R., Evaluation of color-infrared aerial photography as a tool for the identification of sweetpotato whitefly induced fungal and viral infestations of cotton and lettuce, *Proc. Bienn. Workshop Color Aerial Photogr. Plant Sci.*, Weslaco, Tex., Am. Soc. Photogramm. Remote Sensing, 1987, 141.
45. Muirhead-Thomson, R. C., The use of vertical aerial photographs in rural yellow fever mosquito surveys, *Mosq. News*, 33, 241, 1973.
46. Wagner, V. E., Hill-Rowley, R., Norlock, S. A., and Newson, H. D., Remote sensing: a rapid and accurate method of data acquisition for a newly formed mosquito control district, *Mosq. News*, 39, 283, 1979.
47. Garland, A. C., The Use of Meteosat in the Analysis of African Armyworm Outbreaks in East Africa, Ph.D. thesis, University of Bristol, Bristol, U.K.
48. Hutchins, S. H., Higley, L. G., and Pedigo, L. P., Injury equivalency as a basis for developing multiple-species economic injury levels, *J. Econ. Entomol.*, 81, 1, 1988.
49. Peterson, R. K. D., Higley, L. G., Buntin, G. D., and Pedigo, L. P., Flight activity and ovarian dynamics of the yellow woollybear, *Spliosoma virginica* (Lepidoptera: Arctiidae), in Iowa, *J. Kan. Entomol. Soc.*, 66, 97, 1993.
50. Higley, L. G., New understandings of soybean defoliation and their implications for pest management, in *Pest Management of Soybean*, Copping, L. G., Green, M. B., and Rees, T. R., Eds., Elsevier, Amsterdam, 1992.