Agriculture is an essential part of our economy on which we all depend for food, feed, and fiber. With the increased agricultural productivity in this country as well as abroad, the general public has taken agriculture for granted while voicing its concern and dismay over possible adverse effects of agriculture on the environment. The public debate that has ensued on the subject has been brought about, in part, by the indiscriminate use of agricultural chemicals and, in part, by misinformation, based largely on anecdotal evidence.

At the national level, recommendations have been made for increased research in this area by such bodies as the Office of Technology Assessment, the National Academy of Sciences, and the Carnegie Commission on Science, Technology, and Government. Specific issues identified for attention include: contamination of surface and groundwater by natural and chemical fertilizers, pesticides, and sediment; the continued abuse of fragile and nutrient poor soils; and suitable disposal of industrial and agricultural waste.

Although a number of publications have appeared recently on specific environmental effects of some agricultural practices, no attempt has been made to approach the subject systematically and in a comprehensive manner. The aim of this series is to fill the gap by providing the synthesis and critical analysis of the state-of-the-art in different areas of agriculture bearing on environment and vice versa. Efforts will also be made to review research in progress and comment on perspectives for the future. From time to time methodological treatises as well as compendia of important data in handbook form will also be included. The emphasis throughout the series will be on comprehensiveness, comparative aspects, alternative approaches, innovation, and worldwide orientation.

Specific topics will be selected by the Editor-in-Chief with the council of an international advisory board. Imaginative and timely suggestions for the inclusion in the series from individual scientists will be given serious consideration.

Published Titles

- Environmentally Safe Approaches to Crop Disease Control
- Soil Amendments and Environmental Quality
- Soil Amendments: Impacts on Biotic Systems
Environmentally Safe Approaches to CROP DISEASE CONTROL
To our parents and our family for their love, support, and patience.
Environmentally Safe Approaches to CROP DISEASE CONTROL

Edited by
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Plant diseases are one of the major causes of crop loss worldwide. It has been estimated that billions of dollars worth of crops are lost each year due to plant diseases. Historically, chemicals have been relied upon extensively as the primary crop management tool for preventing and controlling diseases. Recently, the use of chemicals in agriculture as well as in the home garden has come under sharp criticism by those who fear their improper use and adverse effect on the environment. This concern, coupled with the pressure from increased regulation of the pesticide industry, has prompted researchers to investigate alternative approaches for managing crop diseases. This reference book provides a balanced overview of the state-of-the-art environmentally safe approaches to crop disease control and the current research trends in this area. Each chapter was written by an expert in the respective field. This publication should be a useful resource to students and professionals in the fields of plant pathology, agronomy, horticulture, and environmental sciences, as well as those concerned with environmental issues in agriculture.

The editors wish to thank the individual contributors for the time and effort they put into the preparation of their chapters. Special thanks are also due to the CRC Press staff and editorial board for their general guidance and assistance.

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Part I
Environmental Aspects
CHAPTER 1

The Influence of Environment on the Development and Control of Disease

H. Alastair McCartney

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I. INTRODUCTION

Crop disease epidemics are the result of the complex interaction between the pathogen (or pathogens), the host, and its environment. What is meant by “environment” in the context of disease epidemiology? Environmental factors can have either a direct effect on the pathogen, for example, many fungal spores need water to germinate, or an indirect effect through their influence on the physiological status of the crop. For soil-borne pathogens the physical and chemical nature of the soil is obviously of paramount importance, but for foliar pathogens the microclimate of the crop plays the dominant role through its effect on leaf surfaces. When considering the dispersal of pathogens we may need to include the atmosphere near the ground in our definition of the appropriate “environment.” In the rest of this chapter the definition of “environment” will usually be restricted to that which has direct effects on pathogen development.
Since the Second World War crop disease in Western agriculture has largely been controlled with fungicides, pesticides, etc. Over the last few years there has been increasing pressure to reduce the use of phytochemicals in agriculture, both from environmentalists and from governments, for example, in Scandinavia and in Holland the aim is to halve their use by the year 2000. There are several reasons for this. Fungicides are perceived to be dangerous for human health either through direct exposure (during application or by spray drift) or through residues in drinking water or food. Thus, for high-value food products there is a demand for low levels of fungicide and pesticide residues and, as detection instrumentation sensitivity increases, the acceptable levels will continue to decrease. Since 1975 the European Union has issued several directives on the reduction of pesticide levels in drinking water (Regulations 75-440/EEC, 79-869/EEC, and 80-778/EEC). Intensive use of phytochemicals is considered to be detrimental to the environment. Pimentel et al. (1991) estimated that in the U.S. the costs of indirect damage due to long-term environmental pollution from agriculture was approximately $10^9 (U.S.). In Europe, where intensive farming is common, the costs are probably at least equal to if not greater than this. The environmental consequences of phytochemical use include adverse effects on nontarget organisms including beneficial fungi pathogenic to insects; direct and indirect effects on insects, fish, and mammals; subtle perturbations on natural ecosystems; and the development of resistant strains of pathogenic organisms. The pressure to reduce the use of phytochemicals, particularly in the U.S. and Europe, is likely to intensify toward the end of the millennium.

The best way to control disease in crops is to eliminate the disease by growing resistant cultivars. A significant proportion of the effort of plant breeders is devoted to developing disease resistance in crops; but even with the advent of new molecular genetic techniques, it is likely that for the foreseeable future crop protection will still rely heavily on the use of phytochemicals. Thus, crop protection strategies need to be developed which minimize the use of phytochemicals while maintaining acceptable economic returns for the farmer. This means either the development of novel control measures, for example, based on biological control, or more efficient use of control chemicals. There are several options available for developing such control strategies, each requiring knowledge of the interactions between the pathogen, host, and environment. For example, reduction in phytochemical use can be achieved by applying control measures only at critical times during the development of an epidemic. This requires an understanding of how epidemics develop, including knowledge of the relationship between microclimate and pathogen growth. Alternatively, agronomic practices may be devised based on disease avoidance, for example, by the choice of a sowing date to avoid periods of weather which favor disease development or spread. Knowledge of the interactions between the pathogen and environment may suggest methods of manipulating crop microclimate through breeding or agronomic practice, which reduces the chances of disease. The reduction in phytochemical use could also be achieved by more efficient application of sprays, for example by targeting specific parts of the crop vulnerable to attack. Environmental factors play an important role in spray application and in the retention of spray on crop surfaces.

Although there is usually little we can do to control or manipulate those environmental factors which regulate disease development in crops, except perhaps for protected crops, it is clear that an understanding of how environmental factors affect crop pathogens is essential for the development of effective control strategies. The rest of this chapter will consider the effects of the major microclimatic factors on the development of plant pathogens, the crop environment, and how this information can be used to control disease. The chapter will concentrate on foliar diseases caused by fungal pathogens, but the general principles should find applications in other crop/disease systems.

II. THE PATHOGEN AND ENVIRONMENT

The physical environment affects all stages in the life cycle of plant pathogens and much of the effort in plant pathological research is devoted to the study of these changes. Temperature and moisture are the two most important microclimatic factors in the direct interaction of host and pathogen, although radiation (light) can have also have an influence, while wind and rain play
dominant roles in dispersal processes (including dispersal by insect vectors). The exact relationship between the pathogen and environmental parameters differs for each pathogen/host system and a comprehensive summary would fill several volumes. This section will illustrate some of these interactions, using selected examples taken mainly from foliar fungal pathogens of arable crops.

The interaction between fungal pathogens and host broadly follows three phases: infection, development, and sporulation — each of which may be limited by environmental factors.

Infection usually includes spore germination followed by penetration and invasion of the host tissue. Some spores, referred to as resting spores, remain dormant for a period after their formation (Hawker and Madelin, 1976; Manners, 1982). In some cases, dormancy can be broken in response to environmental cues. For example, alternate wetting and drying or freezing and thawing will stimulate the germination of teliospores of *Puccinia graminis*, while spores of *Tilletia* species require exposure to temperatures below 10°C before they will germinate (Manners, 1982). Many spores do not have a dormant period as their function is to propagate the pathogen quickly. Nevertheless, their germination is often mediated by environmental factors. However, it must be remembered that spore germination can be stimulated or inhibited by other factors related to the host and the pathogen (Macko, 1981).

As with all biological functions, spore germination is temperature dependent. For all spores there is a minimum and maximum temperature range, outside of which the spores will not germinate (Table 1). The rate of germination tends to increase to a maximum at an optimum temperature, then decline as the temperature increases. For example, Harthill and Cheah (1984) found that germination of conidia of *Pyrenopeziza brassicae* on cauliflower (*Brassica oleracea*) increased almost linearly from 6°C to about 16°C before declining with higher temperatures until ceasing at 26°C; similar results were found by Figueroa et al. (1995a) on oilseed rape (*Brassica napus*). Rust basidiospore germination usually takes place over a wide range of temperatures, but thermal death occurs in some species at temperatures greater than 30°C (Gold and Mendgen, 1991). Temperature can also affect the rate of penetration of the pathogen into the host. In epidemiology this is often quantified as the incubation period: the time taken between spore inoculation and appearance of visible symptoms (Manners, 1982). The temperature response of incubation periods tends to reflect that for germination, with the shortest periods found near the optimal temperatures for germination. Further development of the pathogen within the host can be quantified, in epidemiological terms, by the latent period: the time between inoculation and the production of spores (Manners, 1982). Both infection and latent periods are influenced by temperature (Table 1). Latent periods tend to decrease with increasing temperature, providing other factors are favorable. For example, the latent period for *Erysiphe graminis* decreases from about 14 days at 5°C to 3 days at 18 to 25°C, and that for *Puccinia hordei* decreases from 60 days at 5°C to 6 days at 25°C (Polley and Clarkson, 1978). The effects of temperature on pathogen/host interactions may be moderated by other environmental factors such as light and moisture.

Light can stimulate germination in some fungi and inhibit it others (Leach and Anderson, 1982), but the mechanisms are poorly understood. Most basidiospores are released at night or early morning; however, darkness is not necessary for germination and when other conditions are favorable light has either no effect or a marginal negative effect (Gold and Mendgen, 1991). The effects of light on germination can be complex: for example, germ-tube elongation of *Alternaria linicola* conidia on linseed under wet conditions was interrupted by exposure to light periods of up to 12 h immediately after inoculation, but resumed on return to darkness (Vloutoglou et al., 1996). However, if the light period started 6 h after inoculation germ-tube elongation was unaffected, suggesting that the germ-tubes had started penetration and were no longer sensitive to light. Similarly, exposure to light reduced the probability of infection of wheat leaves by *Mycosphaerella graminicola* (*Septoria tritici*) early in the infection process but stimulated it later (Shaw, 1991). Schuh (1993) found that 12 h dark/12 h light was more conducive to germination of *Cercospora kikuchii* on soybean than, in descending order, 24 h dark or 12 h light/12 h dark or 24 h light. The importance of light on the development of epidemics in the field has been little studied and is poorly understood.

Moisture is probably the most crucial environmental parameter affecting spore germination and infection of fungal pathogens. Most fungal spores require high relative humidities to germinate
### Table 1a  Some Recent Studies on the Effects of Environmental Factors and Pathogen Development: Infection Processes

<table>
<thead>
<tr>
<th>Pathogen (Crop)</th>
<th>Infection/Incubation Period (ip)</th>
<th>Temperature</th>
<th>Wetness</th>
<th>Latent Period</th>
<th>Source</th>
</tr>
</thead>
<tbody>
<tr>
<td>Arthuriomyces peckianus (raspberry)</td>
<td>Germination: optimum 20°C</td>
<td>min 6 h @ 20°C</td>
<td></td>
<td></td>
<td>Truxall et al. (1995)</td>
</tr>
<tr>
<td>Ascochyta fabae f. sp. lentis (lentils)</td>
<td>Infection frequency &gt; @10 and 15°C than @ 25°C</td>
<td>&gt;24 h</td>
<td>6–7 day @ 20°C</td>
<td></td>
<td>Pedersen and Morrall (1994)</td>
</tr>
<tr>
<td>Aspergillus niger (fig)</td>
<td>Optimum 35°C</td>
<td>&gt;97% RH</td>
<td>13–14 day @ 10°C</td>
<td></td>
<td>Subbarao and Michailides (1995)</td>
</tr>
<tr>
<td>Botrytis cinerea (strawberry)</td>
<td>Optimum 20°C (32 h wet)</td>
<td>min 12 h</td>
<td>5–7 day @ 5°C</td>
<td></td>
<td>Sosaalvarez et al. (1995)</td>
</tr>
<tr>
<td>(black spruce)</td>
<td>Zero @ 4 and 36°C</td>
<td></td>
<td></td>
<td></td>
<td>Zhang and Sutton (1994)</td>
</tr>
<tr>
<td>Cercospora arachidicola (groundnut)</td>
<td></td>
<td></td>
<td></td>
<td></td>
<td>Wadia and Butler (1994a)</td>
</tr>
<tr>
<td>Fusarium moniliforme (fig)</td>
<td>Optimum 30°C</td>
<td>&gt;97% RH</td>
<td>40 h @ 30°C (wild isolate)</td>
<td></td>
<td>Subbarao and Michailides (1995)</td>
</tr>
<tr>
<td></td>
<td>Range 8–34°C</td>
<td>min 20 h</td>
<td>38 day @ 12°C</td>
<td></td>
<td>Wadia and Butler (1994a)</td>
</tr>
<tr>
<td>Phaeoisariopsis personata (groundnut)</td>
<td>Optimum 20°C</td>
<td>Range 8–34°C</td>
<td>13 day @ 25°C</td>
<td></td>
<td>Butler et al. (1994)</td>
</tr>
<tr>
<td></td>
<td></td>
<td>min 20 h</td>
<td>26 day @ 33°C</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Puccinia arachidis (groundnut)</td>
<td></td>
<td></td>
<td>49 day @ 12°C</td>
<td></td>
<td>Wadia and Butler (1994a)</td>
</tr>
<tr>
<td></td>
<td>Range 4–30°C</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Pyrenopeziza brassicae (oilseed rape)</td>
<td>ip, opt wetness 21–11 day @ 12°C</td>
<td>min 13 h</td>
<td>22 day @ 5°C</td>
<td></td>
<td>Figueroa et al. (1995a,b)</td>
</tr>
<tr>
<td></td>
<td>14–18 day @ 18°C</td>
<td></td>
<td>14 day @ 15°C</td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>Time for 50% germination: 40h @ 5°C; 18 h @ 15–20°C</td>
<td></td>
<td>17.5 day @ 20°C</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Rhynchosporium secalis (winter barley)</td>
<td></td>
<td></td>
<td>(RH 100%)</td>
<td></td>
<td>Davis and Fitt (1994)</td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>24 day @ 5°C</td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td></td>
<td></td>
<td>19 day @ 10°C</td>
<td></td>
<td></td>
</tr>
</tbody>
</table>
### Table 1a (continued)  Some Recent Studies on the Effects of Environmental Factors and Pathogen Development: Infection Processes

<table>
<thead>
<tr>
<th>Pathogen (Crop)</th>
<th>Infection/Incubation Period (ip)</th>
<th>Latent Period</th>
<th>Source</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Temperature</td>
<td>Wetness</td>
<td>Temperature</td>
</tr>
<tr>
<td>Septoria apiicola</td>
<td>Optimum 25°C</td>
<td>min 10 h</td>
<td>16 day @ 15°C</td>
</tr>
<tr>
<td>(celery)</td>
<td>Range at least 10–30°C</td>
<td></td>
<td>13 day @ 20°C</td>
</tr>
<tr>
<td>Septoria glycines</td>
<td>Optimum 25°C</td>
<td>min 6 h</td>
<td>Mathiea and Kushalappa (1993)</td>
</tr>
<tr>
<td>(soybean)</td>
<td>Range at least 15–39°C</td>
<td></td>
<td>Schuh and Adamowicz (1993)</td>
</tr>
<tr>
<td>Uromyces vicieae fabae</td>
<td>—</td>
<td>8–10 day @ 18°C</td>
<td>Sache and Zadoks (1995)</td>
</tr>
<tr>
<td>(field bean)</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

Note: See also Huber and Gillespie, 1992.

### Table 1b  Some Recent Studies on the Effects of Environmental Factors and Pathogen Development: Sporulation Processes

<table>
<thead>
<tr>
<th>Pathogen (host)</th>
<th>Temperature</th>
<th>Wetness</th>
<th>Light</th>
<th>Source</th>
</tr>
</thead>
<tbody>
<tr>
<td>lentis (lentil)</td>
<td>Maximum 30°C</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Botrytis cinerea</td>
<td>Optimum 17–18°C</td>
<td>3–7 days</td>
<td>—</td>
<td>Sosaalvarez et al. (1995)</td>
</tr>
<tr>
<td>(strawberry)</td>
<td>No sporulation @</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>30°C</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Cercospora carotae</td>
<td>Optimum 28°C</td>
<td>&gt;96% RH</td>
<td></td>
<td>Carisse et al. (1993)</td>
</tr>
<tr>
<td>(carrot)</td>
<td>Range at least 16–32°C</td>
<td></td>
<td>Enhanced by 12 h wetness</td>
<td></td>
</tr>
<tr>
<td>Oncobasidium theobromae</td>
<td>Maximum 26°C</td>
<td>Min 5 h for spore release</td>
<td>Alternating light and dark, release during dark</td>
<td>Dennis et al. (1992)</td>
</tr>
<tr>
<td>(cocoa)</td>
<td></td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Phaeoisariopsis personata</td>
<td>Optimum 28°C</td>
<td>&gt;94.5% RH</td>
<td></td>
<td>Butler et al. (1995)</td>
</tr>
<tr>
<td>(groundnut)</td>
<td>Range 10–33°C</td>
<td>Optimum 99–100%</td>
<td>Reduced by continuous wetness, but better with intermittent wetness</td>
<td></td>
</tr>
<tr>
<td>Phoma bakerana</td>
<td>Range about 15–30°C</td>
<td></td>
<td>Enhanced by near ultraviolet</td>
<td>Nebane and Ekpo (1992)</td>
</tr>
<tr>
<td>(culture)</td>
<td>No sporulation @ 10 or 35°C</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>(sunflower)</td>
<td>Range 4–39°C</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>
and some need the presence of free water (Table 1). Some spores will germinate at low relative humidities, for example, *Erysiphe graminis*, but even for this pathogen optimum germination occurs when the air is close to saturation (Polley and Clarkson, 1978). The presence of free water inhibits the germination of *Erysiphe graminis*, and is generally inhibitory to basidiospores or leads to the formation of secondary basidiospores (Gold and Mendgen, 1991). The duration of wetness needed to promote germination and infection is usually dependent on temperature (Table 1) and periods as short as 0.5 h (*Phytophthora cactorum* on strawberry, Grove et al., 1985) to greater than 100 h (*Diaporthe phaseolorum* on soybean, Damicone et al., 1987) have been reported. Huber and Gillespie (1992) list the wetness requirements for a number of foliar pathogens from studies done between 1985 and 1991. In some fungal pathogens which require free water for germination and infection, discontinuous wet periods will still initiate infection (Stuckey and Zadoks, 1989; Chakrabortry et al., 1990), and in recent years there has been an increasing interest in the effects of intermittent wetness on infection and sporulation processes (e.g., Shaw, 1991; Mridha and Wheeler, 1993; Schuh, 1993; Sah, 1994; Butler et al., 1994; Wadia and Butler, 1994b; Hong and Fitt, 1995; Vallaville-Pope et al., 1995; Vloutoglou et al., 1996). Intermittent wet periods can reduce the efficiency of infection; for example, fewer *Pyrenophora tritici-repentis* lesions developed on wheat plants exposed to different periods of dryness after an initial 6-h wetness than on plants exposed to continuous 24-h wetness (Sah, 1994). Butler et al. (1994) found that intermittent wetness enhanced infection efficiency of *Phaeoisariopsis personata* in groundnut (*Arachis hypogaea*) compared to continuous wetness. Intermittent wetness produced more germ-tubes per conidium and increased branching, which may have increased the chances of stomatal penetration (Wadia and Butler, 1994b). Interruption of wetness also enhanced infection of soybean by *Cercospora kikuchii*, but only when the relative humidity during the dry period was >95% (Schuh, 1993). There are suggestions that for some pathogens wetness periods may be partially cumulative (Stuckey and Zadoks, 1989; Wadia and Butler, 1994b). The significance of the interruption in wetness on infection processes is not known for most pathogens, and as wetness in crops is often discontinuous, especially when caused by rain, priority should be placed on studies in this area. Crop moisture status may also affect the development of disease after infection. Sah (1994) found that necrosis on wheat caused by *Pyrenophora tritici-repentis* was greater when the relative humidity during the incubation period (after a postinoculation wet period) was 95% than when it was 35 or 70%.

The first step in the spread of plant disease is the production of inoculum. For fungal pathogens inoculum consists of spores which can be dispersed in a number of ways (see below). In foliar pathogens spore production can be affected by temperature, moisture (atmospheric and surface), and light, each often interacting with the other (Rotem et al., 1978; Leach and Anderson, 1982). For soil pathogens, soil moisture and temperature are the most important environmental factors affecting sporulation (Lacey, 1986). Much work has been done on the effects of these environmental factors to sporulation *in vitro*, but there has been much less attention paid to their effects *in vivo* (Rotem et al., 1978; Huber and Gillespie, 1992) and there is concern that *in vitro* results may not be directly applicable to conditions in crops (Rotem et al., 1978; Lacey, 1986).

The effects of temperature interact with those of other environmental factors as well as those related to the host and saprophytic microflora (Rotem et al., 1978). The optimum temperature for spore production can be less than that for infection processes (Polley and Clarkson, 1978) and in some species spores may be formed at temperatures too low for lesion development (Rotem et al., 1978). Epidemiologically, the effects of temperature and moisture on sporulation are reflected by their effects on the latent period, the time between inoculation and sporulation (Table 1).

Moisture, whether humidity or free water, can be important in sporulation processes. Most leaf-infecting fungi require high relative humidities for sporulation (Lacey, 1986), although the powdery mildews, rusts, and some other pathogens sporulate over a wide range of relative humidities (Rotem et al., 1978). It is not always clear whether pathogens which sporulate under humid conditions prefer humidities close to saturation over the presence of free water (Rotem et al., 1978). Free water can inhibit sporulation in some species which require high humidities, for example, *Peronospora tabacina* and probably most powdery mildews (Lacey, 1986). Others (e.g., *Phytophthora infestans*, *Helminthosporium maydis*) sporulate equally well in the presence of free...
water or high humidity (Rotem et al., 1978). The effect of surface water varies with species and may depend in part on the type of water cover (drop size, film depth), duration of wetness, and other environmental or host factors (Lacey, 1986). For pathogens requiring surface wetness for sporulation the minimum periods can be as little as 2 to 3 h, but are generally much longer (Rotem et al., 1978) and are often longer than those needed for infection. Spore production tends to increase sigmoidally with an increasing wetness duration above the minimum needed for spore induction. For some pathogens (e.g., *Phyllosticta maydis*, *Ascochyta pisi*) the minimum wetness period may be several days, which is longer than that usually found in crops, suggesting that these fungi respond to intermittent wetness by “accumulating” wetness periods (Rotem et al., 1978). Some pathogens appear to benefit from dry interruptions: *Alternaria porri* f. sp. *solani* produced up to seven times more spores on potatoes under interrupted wetness than under continuous wetness in darkness (Bashi and Rotem, 1975); conidial production of *Phaeoisariopsis personata* on groundnut was more effective under intermittent than under continuous wetness (Butler et al., 1995). It is clear that the interactions between inoculum production and moisture are varied and complex, and that much work is still needed to quantify them for a large number of economically important pathogens.

There has been a substantial amount of work on the effect of light on inoculum production in plant pathogens (Rotem et al., 1978; Leach and Anderson, 1982), but there is little evidence that the direct effect of light on epidemic development is a substantial one once the pathogen is present in the crop (Manners, 1982). Most of the investigations of the effects of light have been made in vitro or in the laboratory (Leach and Anderson, 1982) and effects, particularly with shortwave (blue) or near-ultraviolet (NUV) radiation, have been demonstrated. For example, production of pycnidia in *Phoma bakeriana* in culture (Nebane and Ekpo, 1992), production of spores of *Botrytis cinerea* in culture and on leaves (Nicot et al., 1996), and the production of apothecia of *Sclerotinia sclerotiorum* (Mylchreest and Wheeler, 1987) can all be stimulated with NUV. Exposure to continuous light is inhibitory to spore production in many fungi (e.g., *Phytophthora infestans*, *Alternaria porri* f. sp. *solani*, *Peronospora destructor*), with shorter wavelengths appearing to be most effective (Rotem et al., 1978; Hildebrand and Sutton, 1984), while in others continuous light appears to have no effect (e.g., *Rhynchosporium secalis*, *Erysiphe cichoracearum*). *Ascochyta fabae* f. sp. *lentis* sporulated three to ten times more effectively in culture when exposed to continuous light (Kaiser, 1994) and daily sporulation rates of *Puccinia striiformis* on wheat increased with light intensity (McGregor and Manners, 1985). NUV can stimulate spore production in some species (e.g., *Leptosphaerulina trifolii*, *Botrytis cinerea*, *Alternaria solani*) (Leach and Anderson, 1982; Vakalounakis, 1991), while others (e.g., *Helminthosporium oryzae*, *Stemphylium botryosum*) sporulate under diurnal cycling of NUV and darkness (Leach and Anderson, 1982). Butler et al. (1995) found that *Pheoisariopsis personata* sporulated best on groundnut under a 12-h light/dark cycle. The importance of these effects on sporulation under field conditions are not clear. A better understanding of the effects of light on pathogen inoculum production could have significant implications on disease management, especially in protected crops where the surface wetness/temperature/climate in the crop is done routinely. For example, Vakalounakis (1991) showed that incidence of early blight of tomatoes, caused by *Alternaria solani*, could be halved when the crop was grown in greenhouses covered with a UV-absorbing vinyl film.

Spore dispersal and consequent disease spread is affected by both biological and physical processes (McCartney, 1994). Environmental factors dominate the three phases of dispersal: release, travel, and deposition. Wind obviously plays a major role in the spread of foliar diseases (Aylor, 1990), but rain, water availability, temperature, and light can also be important.

Spores and pollen can be removed from their host by simply being blown or shaken off (passive release), by an active release mechanism, or by being washed or splashed off. Many fungi have spore release mechanisms which can eject spores directly into the air (Ingold, 1971; Lacey, 1986). For example, ascospores of *Sclerotinia sclerotiorum*, can be ejected 2 to 3 cm from the apothecia (Harthill and Underhill, 1976). The mechanisms by which spores are ejected are generally not fully understood, although an adequate supply of water is needed (Gregory, 1973) and release is frequently triggered by changes in environmental factors such as humidity, temperature, or light.
ENVIRONMENTALLY SAFE APPROACHES TO CROP DISEASE CONTROL

In *Sclerotinia sclerotiorum* and *Pyrenopeziza brassicae*, ascospore release from turgid asci can occur in response to a decrease in relative humidity (McCartney and Lacey, 1990, 1992). Drying is also responsible for the release of conidia of *Peronospora tabacina*. The spores are held above the leaf surface on stalk-like structures which twist violently as they dry, throwing spores away from the leaf (Ingold, 1971).

Wind can remove spores by blowing them from surfaces or by shaking foliage or inflorescences. The aerodynamic or mechanical forces generated by wind must overcome the forces holding the particle to the surface (Aylor and Parlange, 1975). The wind speeds needed to remove spores are not known for many fungi, but, it seems likely that they can be relatively large (Grace, 1977). For example, Hammett and Manners (1974) found that conidia of *Erysiphe graminis*, which form chains above the leaf surface, are released in wind exceeding about 0.5 m s\(^{-1}\). Likewise conidia of *Helminthosporium maydis* were only removed when the wind speed exceeded about 5 m s\(^{-1}\) (Aylor, 1975). As the mean wind speeds within plant canopies are generally smaller than those needed to release spores, it seems likely that wind gusts may play an important role in spore removal (Aylor, 1978; Aylor et al., 1981). Spores can also be dislodged by shaking (Bainbridge and Legg, 1976), thus wind gusts may indirectly remove spores by moving the crop canopy. Although the wind conditions (speed and turbulence) needed to remove spores of most pathogens are not known, the gusty nature of wind means that it is unlikely that, for many pathogens, spore removal during the day is limited by the state of the wind.

Rain or spray irrigation can remove spores and pathogenic bacteria from crop surfaces (Fitt et al., 1989) by washing them off in runoff water or in splash droplets or by the shaking caused by the impact of rain drops. The spores of many plant pathogens are held in a mucilage which prevents dispersal by wind and can only be removed by water (in runoff or in splash droplets) when the mucilage is dissolved (Gregory, 1973; Fitt et al., 1989). Splash droplets can be thrown more than a meter from the point of the splash but most only travel a few centimeters (Fitt et al., 1989). However, splash droplets, particularly those smaller than about 100 \(\mu\)m in diameter, have the potential to be further dispersed by wind. The effectiveness of splash in removing spores depends on the size and velocity of the drop and on the orientation and mechanical properties of the surface, but the physical mechanisms are not well understood. Drop size plays an important role in the removal of spores (Gregory, 1973; Fitt and McCartney, 1986b; Fitt et al., 1989) and large drops are more effective than small one. The force of impact of the drops may determine the effectiveness of spore removal (Walklate, 1989). Thus, large slow-moving drops dripping from leaves may remove spores as effectively as small raindrops falling at their terminal velocity. Because of the importance of drop size, the potential for dispersal by splash depends partly on the size distribution of raindrops (Fitt et al., 1989; Walklate, 1989; Walklate et al., 1989), which in turn depends on the type of rainfall (Ulbricht, 1983). For example, Shaw and Royal (1993) found that *Septoria tritici* spores were only transported from the base to the top of a wheat canopy during heavy summer showers.

Wind conditions within and above crops are highly turbulent (McCartney and Fitt, 1985; Aylor, 1990). This causes individual spores to follow different paths and travel different distances, even if released from the same source under the same wind conditions. Therefore, as a spore plume is dispersed downwind of the source its concentration in the air decreases. This decrease in concentration is frequently referred to as a “concentration (or deposition) gradient” (Gregory, 1973). Deposition or concentration gradients tend to be steeper within crops than above them. Within crops the concentration is depleted not only by the expansion of the spore plume but also by deposition of spores on the crop and on the ground. Because spore-carrying water droplets are much larger than the spores they are deposited more effectively, making gradients for splash-dispersed spores steeper than for those dispersed by wind. Wind also transports spores up into the atmosphere where they have the potential to disperse over large distances. Fungal spores have been found at heights of between 500 and 1000 m in the atmosphere (Hirst et al., 1967). Dispersion gradients have been described by a number of different equations (McCartney and Fitt, 1985; Fitt et al., 1987). However, a negative exponential relationship can often be used to quantify gradients:

\[
C = C_0 \exp(-\gamma \cdot x)
\]  

(1)
where \( C \) is the concentration or deposition rate, \( x \) is the distance down wind, and \( C_0 \) and \( \gamma \) are constants. The coefficient determines the rate of decrease in spore concentration (or deposition) with distance. Although this relationship may not be strictly valid in some situations (Aylor, 1987), \( \gamma \) gives a convenient method for visualizing gradients as it can be expressed as a half distance, \( d_{1/2} = (0.693/\gamma) \), that is, the distance in which \( C \) decreases by half. For plant pathogen spores, \( d_{1/2} \) values measured within crops are usually in the order of a few meters, but when measured from the edges of the crops they tend to be larger (Fitt et al., 1987). For example, \( d_{1/2} \) for 20-μm particles (similar in size to mildew spores) released within a barley crop were in the range 0.3 to 1.8 m (McCartney, and Bainbridge, 1984), whereas values of about 9 m were found for Pyrenopeziza brassicae ascospores (McCartney, 1990). Unfortunately, little systematic work appears to have been done in relating dispersal gradients to wind and weather conditions. Atmospheric dispersal models have also been used to assess the effects of wind on spore dispersal (McCartney and Fitt, 1985; Fitt and McCartney, 1986a; Aylor, 1990; McCartney, 1994).

Deposition of spores and pollen onto surfaces can be thought of as being a combination of two mechanisms: gravitational settling and inertial impaction (Legg and Powell, 1979). The rate at which spores settle onto surfaces, \( S \), is proportional to the spore fall speed, \( V_f \), and the spore concentration above the surface, \( C \), \( (S = C \cdot V_f) \). \( V_f \) for most fungal spores generally lies in the range 0.1 to 3 cm s\(^{-1}\) (Gregory, 1973), but many spores and pollens have complex nonspherical shapes and usually fall more slowly than spherical ones of the same volume and density (McCartney et al., 1993). Deposition by impaction, \( I \), is proportional to \( C \) and wind speed, \( u \), \( (I = C \cdot u \cdot E) \). The constant of proportionality, \( E \), is known as the impaction efficiency and increases with spore size (\( V_f \)) and wind speed (\( u \)) but decreases with increasing width of the impaction surface (\( w \)) (Chamberlain, 1975; Aylor, 1982). Wind gusts can enhance the inertial impaction of spores (Aylor, 1978; Aylor et al., 1981), especially in crop canopies where mean wind speeds are small (McCartney and Bainbridge, 1987).

### III. THE CROP ENVIRONMENT

Leaf wetness, temperature, wind, and to a lesser extent radiation, are the most important microclimatic variables which influence the development of epidemics of most crop pathogens. This section will briefly consider each of these variables and how they may be measured or estimated for use in disease control systems.

#### A. Crop Architecture

Architecture plays an important role in the microclimate of vegetative stands as it exerts a considerable influence on where the exchange processes of heat mass and momentum take place. As most exchange processes take place at leaf surfaces, the vertical variation of foliage density is the most important crop characteristic of interest. In most agricultural crops the horizontal variation in leaf density can usually be considered of secondary importance and the crop can be considered to consist of one or more uniform layers. For example, cereal crops are often modeled as a single layer while crops like maize, sunflower, or many trees may be treated as having a single canopy layer above a stem layer. Because of its influence on exchange processes crop architecture strongly influences the profiles of temperature, wind, and water vapor.

#### B. Temperature

Excluding soil-borne pathogens, most pathogen/crop interactions take place either within or on leaves or stems. Thus, strictly, leaf temperatures should be estimated or measured, especially when results from controlled environment studies are being used to determine pathogen behavior. Leaves have a relatively low heat storage capacity and are subject to both radiative and convective heat loss, consequently, when they are not exposed to radiative heating or cooling (i.e., in shade or under overcast conditions) their surface and internal temperatures will be close to that of the...
surrounding air. However, the surface of a sunlit leaf is commonly 5 to 10°C warmer than the surrounding air, while the shade side is usually 1 to 3°C cooler than the sunlit side (Oke, 1978). At night under clear skies, leaves can cool to 5 to 10°C lower than the air temperature. The effects of radiative heating and cooling could be important for pathogens which infect leaves close to the tops of crop canopies (e.g., flag leaves in cereals) as they will be subjected to much larger fluctuations in temperature than air temperature measurements would indicate. The elevation or depression of temperature of an individual leaf can be estimated by considering its energy balance. Such calculations require measurement or estimates of a number of other micrometeorological variables such as net radiation, air temperature, water vapor pressure (for transpiring leaves), and wind speed, as well as knowledge of the physiological status of the leaf (Monteith and Unsworth, 1990).

Air temperature within crop canopies is not usually uniform, but varies with height depending on the architecture of the crop, time of day, and cloud cover. Details of the variation in temperature profile depend on the nature of the crop and local climatic conditions, but a few general observations may be made. At night under clear skies, long-wave radiative cooling can give rise to a temperature minimum near the top of the canopy so that temperature increases upward into the atmosphere and downward into the crop, although the variation may only be a few degrees. Under overcast skies temperature gradients both at night and during the day tend to be relatively uniform.

During the day the opposite happens, when the upper parts of the canopy may be subjected to a substantial radiation load. Then, an air temperature peak is often observed close to the top of the canopy. The difference between the maximum within-crop air temperature and that above the crop is usually about 2 to 3°C, although excess temperatures of 10°C or more are possible in cold climates when transpiration is low (Monteith and Unsworth, 1990). For many pathogens, using above-crop air temperature may be adequate for disease forecasting or modeling, especially when temperatures are close to the optimum. When temperatures are near the upper or lower limits of the pathogen's range the use of in-crop temperatures may be more appropriate. Micrometeorological models have been developed to predict within-crop air temperatures and other microclimatic variables for a number of crops (Table 2) and these could be incorporated into disease forecasting or warning models.

C. Wind

Wind can influence disease epidemic development either directly through inoculum dispersal, or indirectly by affecting other environmental factors such as crop temperatures and leaf wetness duration. The measurement of mean wind speeds above crops is generally fairly straightforward and anemometers compatible with most data acquisition systems are usually available. Wind speed and direction are nearly always included in automatic weather station specifications.

Above-crop mean wind speeds increase with height, depending on the nature of the crop (height, architecture, density) and the stability of the atmosphere (temperature profile) (McCartney and Pitt, 1985). For a neutrally stratified atmosphere, when buoyancy effects can be neglected, wind speed profiles over open terrain with uniform vegetation can often be described by the "logarithmic wind profile" (Monteith, 1973; Grace, 1977; Monteith and Unsworth, 1990):
Table 2  Examples of Microclimatic Models Used to Estimated Dew or Leaf Wetness Duration in Different Crops

<table>
<thead>
<tr>
<th>Source</th>
<th>Crop</th>
<th>Model</th>
<th>Remarks</th>
</tr>
</thead>
<tbody>
<tr>
<td>Monteith and Butler (1979)</td>
<td>Cocoa pods</td>
<td>Heat balance including thermal lag.</td>
<td>Overestimated duration of wetness.</td>
</tr>
<tr>
<td>Pedro and Gillespie (1982a)</td>
<td>Single leaf</td>
<td>Energy balance, micrometeorological data, exposed and shaded leaves</td>
<td>Dew duration within 30 min for exposed leaves and 60 min for shaded leaves.</td>
</tr>
<tr>
<td>Pedro and Gillespie (1982b)</td>
<td>Single leaf</td>
<td>As Pedro and Gillespie (1982a) but with weather station data</td>
<td>Dew duration within 1 h for exposed leaves and 1.5 h for shaded leaves.</td>
</tr>
<tr>
<td>Weiss et al. (1989)</td>
<td>Dry bean</td>
<td>CUPID (Norman, 1982)</td>
<td>Good agreement with sensor measurements and observations.</td>
</tr>
<tr>
<td>Huber and Itier (1990)</td>
<td>Field bean</td>
<td>Multilayer model with heat conduction</td>
<td>Good agreement with observations, especially near the base of the crop.</td>
</tr>
<tr>
<td>Jacobs et al. (1990)</td>
<td>Corn</td>
<td>Multilayer, Bowen ratio energy balance with soil diffusivity, dew deposition</td>
<td>Dew deposition range +10 to −30% of observed.</td>
</tr>
<tr>
<td>Zhang and Gillespie (1990)</td>
<td>Corn</td>
<td>Energy balance, single leaf, micrometeorological or weather station data</td>
<td>Good agreement for shaded leaves.</td>
</tr>
<tr>
<td>Weihang and Goudriaan (1991)</td>
<td>Short grass, rice</td>
<td>Multilayer energy balance based on Microweather (Goudriaan, 1977), daily weather data</td>
<td>Dew formation simulation, a simplified model works well for nighttime dew formation.</td>
</tr>
<tr>
<td>Lhomme and Jimenez (1992)</td>
<td>Banana, plantain</td>
<td>Pedro and Gillespie (1982b) applied to upper leaves, standard weather data</td>
<td>Predicted wetness duration to &lt;1 h on average.</td>
</tr>
<tr>
<td>Jacobs and Nieveen (1995)</td>
<td>Corn, barley</td>
<td>Multilayer, Bowen ratio energy balance, dew deposition</td>
<td>Good agreement on dew deposition for corn, duration underestimated by about 1 h. Not as good for barley due to effects of runoff and dripping.</td>
</tr>
<tr>
<td>Wittich (1995)</td>
<td>Apple orchard</td>
<td>Energy balance, single leaf, different boundary conditions.</td>
<td>Best agreement when temperature of canopy below leaf was assumed to be the same as that of the leaf, usually &lt;1 h difference</td>
</tr>
</tbody>
</table>

\[ u(z) = 0.4 \cdot u_* \cdot \ln \left( \frac{z-d}{z_0} \right) \]  

where \( u(z) \) is the mean wind speed at height \( z \) above the ground. In the equation the constant \( u_* \), the friction velocity, scales the wind speed; \( z_0 \), the roughness length, scales the height; and \( d \) is a datum level, less than the crop height, called the zero plane displacement. At height \( d \) Equation 2 predicts that \( u = 0 \), but the equation is not valid within the crop. For most crops \( z_0 \) is usually an order of magnitude smaller than the height of the vegetation, \( h \), and \( d \) is between 0.6 and 0.8 \( h \) (Monteith, 1973; Thom, 1975). Values of \( z_0 \) and \( d \) for a range of surfaces have been tabulated by
McCartney and Fitt (1985). Both $z_0$ and $d$ may show some dependence on wind speed if the canopy is distorted and smoothed as wind speed increases (Grace, 1977). On sunny days when there is convective activity (unstable temperature lapse rate) or in the evening when atmospheric mixing is suppressed (stable temperature lapse rate) the wind profile deviates from Equation 2 (Thom, 1975; Grace, 1977; McCartney and Fitt, 1985). The equation can be modified to account for the effects of atmospheric stability (Monteith, 1973; McCartney and Fitt, 1985), but as the modifications are not simple the reader should consult more specialized texts. The logarithmic wind profile is strictly only applicable to well-formed surface boundary layers, that is, large uniform areas. Wind profiles near obstructions such as hedges or near changes in terrain, for example woodland boundaries, may be more complex than suggested by Equation 2. However, the general form of the profile is usually similar and the equation can be used as a first approximation to estimate wind speeds at crop height from local synoptic measurements (usually made at 10 m).

The wind profiles within crops greatly depend on crop architecture, particularly the vertical distribution of foliage and the size, shape, and density of foliage elements. For crops where the foliage is relatively uniformly distributed with height, for example cereals, wind speed profiles can often be estimated using an equation suggested by Cionco et al. (1963):

$$u(z) = u(h) \cdot \exp \left[ \alpha \left( \frac{z}{h} - 1 \right) \right]$$

(3)

where $h$ is the crop height, and the attenuation coefficient $\alpha$ has a value between 0.3 and 3 depending upon crop type and leaf area density. Some values of $\alpha$ are given by McCartney and Fitt (1985). An alternative equation, but with a similar shape, was given by Landsberg and James (1971) and Thom (1975). As a first approximation, the wind speed within the crop can be taken as a fixed fraction of the speed at the top of the crop (i.e., $u(z) = \beta u(h)$); $\beta$ usually has a value between 0.1 and 0.5 (McCartney and Fitt, 1985). In both equations $u(h)$ can be estimated from Equation 2 with $z$ set equal to $h$. In crops with a “canopy and stem” structure (orchards) the wind speed profile may be “S”-shaped, with wind speeds in the “stem” layer greater than within the “canopy” layer (Grace, 1977; Oke, 1978).

D. Humidity

Relative humidity, or more strictly water vapor pressure, within the canopy can be significantly different from that of the air above the crop. The profile of vapor pressure is the consequence of the heat and water balance of the system. Understanding and modeling water vapor distribution and movement within crops has been the main concern of crop microclimatologist for many years (Monteith, 1975; Oke, 1978; Monteith and Unsworth, 1990). During the day the water vapor pressure is usually higher within the crop than above it, because the crop is transpiring, and it increases toward the soil surface. At night the differences between within- and above-crop water vapor pressures are smaller. Near the top of the canopy water vapor pressure may be less than above when dew is forming. In a transpiring crop the water vapor pressure close to the leaf surface, where the pathogen is growing, may be close to saturation.

Water vapor pressure is not normally measured directly but is calculated from relative humidity and temperature measurements. There are several techniques available to measure relative humidity including sensors compatible with most data acquisition systems (see, for example, Campbell and Madden, 1990). These include electronic (usually capacitive) sensors and wet and dry bulb psychrometers. Electronic sensors are simple to use but can suffer from calibration drift problems if they are used for long periods at high humidities. Dew-point meters give a more direct measurement of water vapor pressure, but are expensive and not usually suited for automatic use over long periods.

E. Wetness

Crop wetness duration is probably the most important environmental factor in crop disease epidemiology. Plants can become wetted directly by rain or spray irrigation or indirectly by dew
formation or guttation (exudation of water from foliage). Unlike the other critical environmental parameters important in host pathogen interactions, the duration of wetness on foliar elements (including fruits, pods, flowers, etc.) is very difficult to define physically because of the spatial inhomogeneity of wet and dry areas both at leaf and crop scale (Huber and Gillespie, 1992). Thus, making meaningful measurement or estimates of wetness duration is not simple. As far as a pathogen (spore, hyphal segment) on a leaf surface is concerned, it is either covered in liquid water (wet) or not (dry). Therefore, perhaps the best way to define “wetness” in relation to foliar disease is “the area or proportion of area containing free water”. Wetness duration could then be defined as the time a given proportion of the crop remained wet. Currently, none of the commonly used wetness sensors (see below) fulfil this standard and complex microclimatic models would be needed to estimate wet leaf area. Nevertheless, the measurement and estimation of wetness duration periods play essential roles in many crop disease warning and forecasting models. The ambiguousness in defining wetness duration complicates the translation of the results of laboratory or controlled environment experiments to the field. In laboratory studies leaves are usually completely dry or nearly 100% wet, therefore wetness duration is relatively easy to define. However, in the field naturally drying leaves will contain different amounts of water and some parts of the leaf will remain wet for longer periods than others, leading to a range of wetness durations for different foliar elements or areas of the crop. Huber and Itier (1990) found differences in leaf wetness duration after rain of up to 10 h in different parts of a bean canopy.

Unlike temperature and wind, it is very difficult to generalize about wetness duration in crops. Patterns of crop wetness are dependent on the complex interaction of crop and environmental factors. The method of wetting (rain, irrigation, dew, or guttation) determines both the amount of water on the crop and to some extent the pattern of leaf wetness: rainfall tends to produce a large range of water drop sizes giving a nonuniform cover on leaves (Royal and Butler, 1986), whereas dew tends to produce more uniform drop sizes and a more even cover. The nature of the foliar surface also plays an important role in determining the pattern of water on the leaf. Wettable leaves, such as tomato, will tend to have fairly uniform water films, while the water pattern on waxy leaves, oilseed brassicas for example, will consist of discrete drops. Wetness duration is a combination of the wetting period and the time taken to dry the crop. Wetting periods not only depend on the source of water but also on crop architecture as this controls the interception of rain and the pattern of dew formation on leaves. Evaporation of water from leaves depends on the radiation load, on the leaf, temperature, water vapor pressure, wind speed, leaf orientation, and water drop size and distribution on the leaf surface. Crop structure is also important in determining drying periods. Therefore, even under identical environmental conditions, wetness duration will differ for different crops or for the same crop at different growth stages.

1. **Rain (Irrigation)**

The length of time a crop remains wet partly depends on how much water it can hold on its foliage. Interception of rain by crops depends on rainfall duration and drop size distribution, wind speed, ground cover, crop structure, and foliage characteristics (Royal and Butler, 1986). Because of its importance in understanding the water balance of large catchments, the interception of rain by forests has been widely studied (Rutter, 1975; Huber and Gillespie, 1992). Agricultural crops, in contrast, have received relatively little attention (Clark, 1940; Appelmans et al., 1980; Butler and Huband, 1984). The type of rain determines both the rate of wetting and the efficiency with which the raindrops are captured. Small raindrops are captured efficiently but large drops will splash on leaves, redistributing the water, and may shake settled water from the plant (Fitt et al., 1989). Strong wind can also shake water from leaves, reducing the amount of water intercepted. Raindrop size distribution is generally related to rainfall intensity (Marshall and Palmer, 1948; Ulbricht, 1983; Bennett et al., 1984): light rain usually contains mostly small drops (<1 mm) but intense showers can have drops of up to about 5 mm in diameter (Willis and Tattelman, 1989). For low-intensity rainfall, crops which have complete ground cover will collect nearly all the rain which falls on them until the crop is saturated and water begins to run off the foliage (Royal and Butler, 1986). In heavy showers much of the rain falling on the crop may reach the ground as
through-fall or runoff. Raindrop size distributions are also important in the dispersal of inoculum by splash (Fitt et al., 1989). Studies by Appelmans et al. (1980) suggest that the water-holding capacity of a crop and the time taken to fully “wet” it may also be dependent on rainfall intensity. There appears to be little information available on the water-holding capacity of most crops, but it depends primarily on the ground cover (leaf area index), crop architecture (leaf orientation), and the nature of the leaves (Royal and Butler, 1985). Butler and Huband’s (1984) studies in wheat suggest that interception increases asymptotically with the rainfall amount until it reaches the water-holding capacity of the crop. Braden (1985, quoted in Friesland and Schrödter, 1988) has suggested the following relationship between rainfall interception \( I_c \), rainfall amount \( P \), leaf area index \( L \), and ground cover \( b \):

\[
I_c = a \cdot L \left[ 1 - \frac{1}{1 + P \cdot b/a \cdot L} \right]
\]

where \( a \) is the maximum water-holding capacity of the crop per unit leaf area index. Rainfall duration determines not only the amount of water held on a crop (until the maximum water-holding capacity is reached) but the distribution of water within the crop. Upper leaves will be wetted first, but after a prolonged rain the crop will become more or less uniformly wet. Mist or fog also produces uniform wetness.

Rain generally produces a large range of drop sizes on foliage, giving a relatively nonuniform cover (Brain and Butler, 1985). However, the surface characteristics of foliage also determine whether water forms films or discrete drops on leaves. Crops which hold water as discrete drops tend to have a larger water-holding capacity per unit leaf area than those which hold the water as films, although some of the leaf area may be “dry”. The size and pattern of water drops on leaves affect evaporation rates and hence wetness duration.

2. **Dew**

At night, radiative cooling can cause the temperature of plant surfaces to fall below that of the air. Dew is formed when the surface temperature falls below that of the dewpoint of the surrounding air. Water will continue to condense as long as the surface temperature is lower than the dewpoint of the air in contact with the leaves. The effective “drying” of the air by condensation means that for dew to continue to be formed there must be a transport of water vapor to the plant surface from the atmosphere and/or from the soil or other plants. When water is transferred from the atmosphere dew formation is known as **dewfall**, otherwise it is called **distillation** (Monteith, 1957, 1963). Dew formation is therefore a fairly complex process involving not only the energy balance of individual surfaces but mechanisms of water vapor transport. For dewfall to occur wind speeds need to be within a critical range, depending on crop canopy structure and the water vapor content of the air above the crop. If wind speeds are too low the water lost by condensation cannot be replaced by turbulent transport from the more humid air above and condensation stops. When wind speeds are too high the temperature of the surface is raised above the dewpoint by turbulent heat transfer and condensation stops (Oke, 1978). For a short grass, a minimum wind speed of about 0.5 m s\(^{-1}\) at 2 m above the ground is needed (Monteith, 1957). Dewfall and distillation probably occur simultaneously in most canopies (Garrat and Segal, 1988) and the relative contributions depend on a number of factors including soil moisture availability, canopy structure, and atmospheric moisture. Under humid atmospheric conditions dewfall dominates, but distillation becomes important under semiarid conditions or at low wind speeds if there is sufficient soil moisture. Calculations suggest that under favorable conditions distillation may account for up to 80% of dew formation (Monteith, 1963; Garrat and Segal, 1988) and therefore cannot be ignored when modeling dew formation in crop canopies. Maximum rates of dewfall for saturated air have been estimated to be between 0.06 and 0.07 mm h\(^{-1}\) (Monteith, 1973) but dew formation may reach 0.09 mm h\(^{-1}\) with contributions from distillation (Garrat and Segal, 1988). Measured values of total dew formation can reach 0.6 mm per night, but values of about 0.2 mm appear to be more typical (Garrat and Segal, 1988; Huber, 1992).
3. **Guttation**

Guttation is the exudation of water through special pores, called hydathodes, generally found on leaf margins. In grasses the hydathodes are usually at the tip of the leaf. When evaporation is low the exuded water forms drops supplied through the roots via the vascular system. Thus for guttation to occur the rate of water supply from the roots must exceed the loss by transpiration. Guttation is probably controlled by soil moisture and occurs at high moisture contents, and soil temperature may also play a regulatory role but only when soil moisture is not limiting (Hughes and Brimblecombe, 1994). In their study of short grass, Hughes and Brimblecombe (1994) found that guttation was responsible for about the same amount of water (0.1 mm per night) as dew. The average size of the guttation drops was much larger than drops formed by dew (1.49 mm compared to 0.2 mm), but they were formed mostly at the leaf tips. Guttation is probably more important in wetland crops such as a rice paddy where soil moisture is not a limitation, and when water temperature is high enough (optimum about 30°C) (Luo and Goudriann, 1997). The importance of guttation drops in disease processes is not clear, particularly since they tend to form along leaf margins.

4. **Measurement of Leaf Wetness Duration**

Methods to estimate or measure leaf wetness duration have been available for as long as its importance in crop disease has been recognized (Wallin, 1963, 1967) and there have been several recent reviews of methods and practices currently in use (Howard and Gillespie, 1985; Sutton et al., 1988; Campbell and Madden, 1990; Weiss, 1990; Huber and Gillespie, 1992). The ideal “wetness sensor” should measure both the duration and amount of water on the **surface of the crop** itself. This has rarely been achieved in practice, and simple-to-use relatively inexpensive sensors for general routine use which conform to this standard have still to be developed.

The amount of water deposited can be measured by weight, and this method has been used for both plant and artificial surfaces. For example, Leick (1932) used plates containing material which absorbed water and assessed dew deposition by weighing the plates before and after exposure. Mechanically recording balances have been used to automatically weigh water deposition on whole plants (Jennings and Monteith, 1954), detached plant shoots (Hirst, 1954), and on polystyrene test surfaces (Hirst, 1957). In recent years weighing methods have gone out of favor, but developments in sensor technology, especially in pressure sensors, make such methods worth reconsidering. Other indirect mechanical methods for sensing wetness have included using a water-soluble pencil to mark a rotating glass plate when it was wet (Taylor sensor) and measuring the change in length due to wetting of an absorbing material (Wallin, 1963).

Since the early 1970s, when electronic data acquisition became common, most wetness sensors have detected the presence of water by measuring the change in electrical impedance between two electrodes under wet and dry conditions. The simplest form of this type of sensor consists of an interlocking grid of electrodes on a nonconducting surface (Crossan, 1962); the surface can be painted with latex paint to enhance wettability (Davis and Hughes, 1970) and to match the color of the crop (Gillespie and Kidd, 1978). In a variation of this design McCartney and Lacey (1990) used a flat plate coated with electrically conducting paint with a “key” pattern inscribed on it; the narrow separation of the electrodes allowed the rapid detection of small droplets produced by dew. Synthetic sensors do not necessarily respond to wetting and drying in the same way as the crop, and wetness duration recorded by artificial sensors can be significantly different from that on the crop (Gillespie and Kidd, 1978; Huband and Butler, 1984; Potratz et al., 1994). To combat this, sensors have been designed to mimic the behavior of leaves; for example, “leaves” made from cotton cloth on a grid network of fine wires gave a better measure of leaf wetness on alfalfa than flat-plate painted sensors (Weiss and Lukens, 1981; Weiss and Hagen, 1983); and Gillespie and Duan (1987) used cylindrical sensors to mimic onion leaves.

The principle of measuring changes in resistance to detect surface water has also been applied directly to plant surfaces. In this type of sensor electrodes are attached directly to the leaf surface; for example, electrodes have been clipped onto leaves (Howard and Gillespie, 1985); Weiss et al.
(1989) used a grid network of independent, adjacent fine wires attached to dry bean leaves; Harold (1995) connected a circular array of fine wires to the capsules of linseed plants. McCartney and Lacey (1990) clipped oilseed rape debris between a frame consisting of two sets of electrodes in a study of *Pyrenopeziza brassicae* ascospore release and Fernandes et al. (1991) used needles inserted into wheat straw to measure water content when studying the maturation of ascospores of *Pyrenophora tritici-repentis*. Needles inserted through leaves have also been used to measure wetness in turfgrass canopies (Giesler et al., 1996). Such sensors are likely to give a more realistic measure of wetness duration than "artificial sensors": Giesler et al. (1996) found that wetness duration estimated by their sensor agreed within about 20 min of direct observation of the leaves. However, resistance sensors cannot easily be calibrated to give reliable estimates of the amount of water on the plant surface.

Indirect methods have been considered for monitoring liquid water on crop surfaces. Bunnenberg and Kühn (1977) used absorption of β-radiation to estimate dew on soil and Barthakur (1983) used this technique to measure the amount of water on individual leaves. The leaf was placed between a source (thallium-204) and a detector. The amount of radiation absorption could be directly related to the amount of water on the leaf surface. In laboratory and field tests the β-ray system gave consistently better estimates of actual wetness duration on leaves than electrical resistance sensors (Barthakur, 1985; Armstrong et al., 1993). Gillespie et al. (1990) have shown that radar backscatter measurements may have the potential to measure water deposition on a whole-crop basis. Both these approaches could have important research applications, but they are unlikely to have applications in routine disease management systems. Changes in the reflection characteristics of plant leaves or in the polarization of reflected light due to the presence of water may be potentially useful in determining crop wetness (A.F.G. Jacobs, personal communication). Microwave emissions from crops appear to be sensitive to the hydrological characteristics of the soil and crop canopy (Wigneron et al., 1996): 1.4-GHz emissions depend on soil moisture while 5-GHz emissions are sensitive to both soil moisture and crop water content. Remote sensing of microwave emissions may be potentially useful in monitoring crop wetness, at least for research purposes.

Direct monitoring of crop wetness has an important part to play in the development of disease control strategies, especially with the advent of on-farm computers and microprocessors. However, as most currently available sensors do not conform to an "ideal sensor", care must be taken with the interpretation of sensor output. This is particularly true for artificial sensors where the sensor response can be different from that of the crop. In such cases the sensor output should be "calibrated" to match the crop. Alternatively, the "disease model" should be matched to the output of specific sensors. There is a need to develop standards of wetness-sensing instruments of different types, and also to investigate and develop noncontact methods of wetness determination applicable to the whole crop or to those parts of the crop vulnerable to infection.

### F. Estimating Crop Microclimate

Direct measurement of microclimatic variables is obviously the best way to obtain the information needed for disease forecasting or warning schemes or for disease control decision strategies. Automatic weather stations are available for on-farm use, but it is unlikely that for the foreseeable future direct measurement of crop environments will be a practical option for most farmers. Thus, within-crop environmental factors need to be estimated from measurements or forecasts made at sites which may be remote from the field. In most cases the only meteorological information available is from a local synoptic weather station. Measurements at such sites are usually of a restricted number of variables and are made under standard exposure conditions, usually 1 m above short grass for temperature, humidity, and radiation or 10 m for wind speed. As noted above, the presence of a crop canopy can significantly alter the local microclimate and it may not be appropriate to use synoptic measurements in disease forecasting models which have been based on controlled environment studies or direct field measurements. Mathematical modeling of crop climates offers a method to estimate critical microclimate parameters from above-crop measurements, but the development of such models is not a trivial exercise (Huber and Gillespie, 1992).
Much of the research in agricultural meteorology is aimed at understanding the interactions between plants and their soil and aerial environments, but there have been relatively few attempts to simulate the complete microclimate of a crop (e.g., *Microweather*, Goudriaan, 1977; *CUPID*, Norman, 1982; Norman and Campbell, 1983; Sauer and Norman, 1995; *AMBETI*, Braden, 1995). In these models the crop is divided into a number of layers and the microclimate (leaf temperature, air temperature, relative humidity, leaf wetness) is simulated by solving the energy balance of each layer. The forcing variables are typically air temperature, solar radiation, vapor pressure (or relative humidity), wind speed, soil temperature, and soil moisture. The models usually operate on hourly or half-hourly time steps, therefore hourly values of input variables should be used, but these can be sometimes estimated from daily values (Norman, 1982). The models also require information on crop structure, usually in the form of leaf area index profiles, crop height, leaf angle distribution, and stomatal and boundary layer resistances. If this information is not available appropriate estimates can be made according to the crop (Norman, 1982). The accuracy of these models depends very much on the quality of the input information, but in the absence of direct measurements they can provide useful information.

In recent years there has been increasing interest in applying micrometeorological models to the estimation of leaf wetness duration (Huber and Gillespie, 1992). The models can use measured or estimated microclimatic inputs to simulate condensation and evaporation processes or can include these processes in complex microclimatic models (Table 2). Leaf wetness duration is calculated from evaporation rates of droplets or water films, and several models based on energy and mass balances have been devised (Butler, 1984; Brain and Butler, 1985; Barthakur, 1988; Butler, 1990; Zhang and Gillespie, 1990). The rate of evaporation depends upon the size and shape of the water drop or film, thus models must take into consideration the origin of the source of wetness. Leaf wetness duration models of various complexity have been developed for a number of crops (Table 2) and their accuracy has often been comparable with that of artificial wetness sensors (Huber and Gillespie, 1992). With the advent of personal computers the use of complex mathematical models for predicting crop microclimate is becoming a practical proposition for both regional and on-farm use.

The use of empirical models for estimating the duration of dew periods is an alternative to microclimatic modeling. Gleason et al. (1994) developed an empirical model which used relative humidity, air temperature, and wind speed to estimate occurrence and duration of dew. The model was based on data from one site and validated with hourly data from 13 weather stations in the north-central U.S. The model predicted the mean dew periods for the study to within 1 h and estimated the occurrence or absence of dew correctly 83.5% of the time. These results suggest that models of this type could provide dew period estimates over similar geographic areas where suitable weather station data are available.

### IV. ENVIRONMENT AND DISEASE CONTROL

Zadoks and Schein (1979) identified six general methods for control of crop diseases: avoidance of the pathogen, exclusion of the pathogen, eradication of the pathogen, protection of the plant, development of host resistance, and therapy applied to the diseased plant. Knowledge of the interactions between host, pathogen, and environment can assist in the development of control strategies in a number of these areas (avoidance, eradication, protection, and therapy) and is one of the cornerstones of “integrated plant disease management”. Disease control measures can also be grouped into “passive” measures (avoidance, exclusion, resistance) where no direct action is taken against the pathogen during the growth of the crop, and “active” measures (protection and therapy).

#### A. Passive Measures

Knowledge of the environmental conditions which are unsuitable for a disease to develop (e.g., lack of leaf wetness, too low or too high a temperature) can be used to devise “disease
avoidance” strategies. Regional or local areas where the climate is suitable for specific diseases can be identified and the growth of susceptible crops avoided (Payen, 1983; Weltzien, 1988; Rossi et al., 1995). Such analyses nearly always have to use synoptic climatological data for the region or area. This is unlikely to provide the microclimatic information needed to assess the impact of many crop diseases, especially those with specific leaf wetness requirements. Micrometeorological modeling can be used to provide in-crop climatic data (Huber and Gillespie, 1992). Scherm and Van Bruggen (1995) used Pedro and Gillespie’s (1982b) model to study the distribution of dew periods in different climatic regions of California. Microclimatic models are also being used to develop climatic zoning for disease in Europe (Löpmeier, 1994; Orlandini et al., 1994). Additionally, knowledge of the geographic and seasonal distribution of crop microclimatic factors may allow periods when the microclimate favors disease to be avoided by adjusting the sowing date.

Passive measures also include manipulation of the microclimate to discourage disease. This is generally more difficult for field crops than for protected crops. Manipulation of the light regime in greenhouses has been used to suppress disease spread (see above, Vakalounakis, 1991). In field crops, variation of agronomic practices (e.g., row spacing, sowing density, irrigation) could be used to manipulate microclimate. For example, Giesler et al. (1996) found that in tall fescue (Festuca arundinacea), increasing the canopy density, by using a different seed rate or cultivars increased leaf wetness and relative humidity, except in wet years. The severity of brown patch disease (Rhizoctonia solani) was greater in the high-density canopies, suggesting that reduction in canopy density could reduce disease incidence. Tompkins et al. (1993) found that the microclimate in no-till winter wheat crops could be affected by changing seed rate and row spacing. High seed rates and narrow row spacing produced canopy microclimates favorable to the development of disease caused by Septoria spp. The duration of leaf wetness was longer and the intensity of downy mildew (Bremia lactucae) was greater in lettuce crops under furrow irrigation than under subsurface drip irrigation (Scherm and Van Bruggen, 1995). Detailed knowledge of crop microclimate processes, including modeling, could be effective in suggesting appropriate changes in agronomic practice. Crop microclimate can also be manipulated by altering crop architecture. This could be achieved directly by mechanical means or by plant breeding. Removal of the leaves from around berry clusters in grapevine has been shown to alter the microclimate, particularly the wind speed, and to reduce the incidence and severity of Botrytis cinerea infection of the fruit (Gubler et al., 1987; English et al., 1989). This practice has since been adopted by wine growers in California. Micrometeorological models could be used to investigate the effects of crop architecture changes and suggest areas which could be exploited by crop breeders.

Mathematical modeling of disease epidemic development in crops has made significant advances over the last 20 years (Campbell and Madden, 1990). It is now feasible to combine disease progress, microclimate, and crop growth models to explore the potential for changing husbandry practices to minimize disease spread and development.

B. Active Measures

The majority of crop disease management measures involve taking action to either protect the crop from the pathogen or to kill or limit the growth of the pathogen on the crop. In both cases this usually involves applying the active ingredient (e.g., phytochemical, biological control agent) directly to the crop. As discussed in the Introduction to this chapter, environmental, social, and economic considerations prohibit the indiscriminate use of such measures. There is, therefore, a need to reduce the use of phytochemicals in agriculture, while maintaining an acceptable economic return for the farmer. This can be achieved in part avoiding the overuse of prophylactic measures and by taking action only when it is necessary to control the disease at an economically acceptable level. Knowledge of the relationships between pathogen, host, and environment can be used to build disease forecasting systems to aid in decisions on when or if to apply control measures. This obviously requires information on the effects of environmental factors on pathogen development.

Campbell and Madden (1990) define a disease forecaster as predictor of “the outbreak or increase in intensity of a disease based on information on the weather, crop or pathogen.” Disease forecasters usually predict one or more critical phases in epidemic development such as initial
Table 3  Some Recent Disease Forecasting/Warning Simulation Models

<table>
<thead>
<tr>
<th>Pathogen(s)</th>
<th>Crop</th>
<th>Source</th>
<th>Remarks</th>
</tr>
</thead>
<tbody>
<tr>
<td>Botrytis cinerea</td>
<td>Potato</td>
<td>Kaukoranta et al. (1993)</td>
<td>Device to monitor weather and calculate disease risk.</td>
</tr>
<tr>
<td>Peronospora destructor</td>
<td>Onion</td>
<td>Scherm et al. (1995)</td>
<td>Recommends spray applications.</td>
</tr>
<tr>
<td>Erysiphegraminisis f. sp. tritici</td>
<td>Winter wheat</td>
<td>Leptodotium elatus</td>
<td>Empirical model to predict first symptoms based on leaf wetness duration.</td>
</tr>
<tr>
<td>Peltaster tructicola</td>
<td>Winter wheat</td>
<td>Zygophiala jamaicensis</td>
<td>Empirical model which uses weather factors.</td>
</tr>
<tr>
<td>Stemphyllium vesicarium</td>
<td>Pear</td>
<td>Montesinos et al. (1995)</td>
<td>Regression model based on wetness duration and temperature.</td>
</tr>
<tr>
<td>Venturia inaequalis</td>
<td>Apple</td>
<td>Xu et al. (1995)</td>
<td>Disease simulation model based on weather factors.</td>
</tr>
</tbody>
</table>

infection, secondary infection, or disease development. Information on environmental factors is usually essential for the disease forecaster, although some only require biological information (Campbell and Madden, 1990). The nature of disease forecasting models depends partly on the source of the environmental information needed: regional synoptic measurements, local synoptic measurements, in-crop measurements, in-crop estimates or, more rarely, weather forecasts. For example, a forecaster based on detailed studies of pathogen processes would probably require measurements or estimates of in-crop micrometeorological variables, while one based on empirical field studies would most likely use local synoptic measurements or estimates. Thus, disease forecasters vary in complexity — from disease warning schemes which identify periods when weather conditions are suitable for infection or disease spread, to systems which predict disease development and potential effects on yield (see reviews cited below). An example of a “disease warning” forecasting system is that for the apple scab (Venturia inaequalis) which identifies periods when the environmental conditions favor infection (leaf wetness and temperature) (Mills and LaPlante, 1951). A microprocessor-based system which monitors weather variables and calculates “infection periods” has been commercially produced and has been validated at several locations (Jones et al., 1980; Campbell and Madden, 1990). In the Netherlands a complex computer-based disease and pest management system, EPIPRE (EPIdemics PREvention) (Zadoks et al., 1984) was developed to aid pest and disease control in winter wheat. This system was field based but used a central computer to simulate disease development of a number of pathogens based on individual field observations. The system used crop and pathogen information as well as weather data.

Many different disease forecasting systems have been produced over the last 50 years and there have been several excellent reviews on their development and use (Campbell and Madden, 1990; Fry and Fohner, 1985; Jones, 1986; Penypacker and Stevenson, 1982; Waggoner, 1984). The development of disease forecasting systems is discussed in detail by Madden and Ellis (1988). Lack of space precludes an attempt to review current and previously used disease forecasters in this chapter (some recent models are listed in Table 3). Instead, there follows an outline of three examples of disease forecasting systems which illustrate the use of environmental information in disease management.

**BLIGHTCAST** (Krause et al., 1975) late blight of potato (Phytophthora infestans) — This was a synthesis of the Hyre and the Wallin fungicide scheduling systems developed for the northeastern and midwestern U.S. **BLIGHTCAST** was based on predicting periods when infection
is likely. Spray recommendations were issued in response to two different weather triggers: after 10 consecutive days when the cumulative rainfall exceeded 30 mm and the 5-day average temperature did not exceed 25.5°C; or after accumulation of 18 to 20 “severity units” (each day was scored on a 0 to 4 “severity” scale according to temperature and hours with relative humidity >90%). **BLIGHTCAST** was a computer programme which calculated the weather triggers from temperature, relative humidity, and rainfall measurements. Initially, **BLIGHTCAST** was run on a computer at Pennsylvania State University and offered as a free service. Farmers collected weather information on a recording hygrothermograph. Each week they telephoned a summary of this information to the university and received **BLIGHTCAST**’s recommendations. The free system was widely used by farmers, but a subsequent subscription service was not successful. A battery-operated microcomputer-based weather data logger and forecasting unit was developed (“Blightcaster”, Campbell Scientific, Inc., Utah) for in-field use (MacKenzie, 1981) This equipment recorded the necessary weather information and displayed **BLIGHTCAST**’s fungicide recommendations. A number of growers successfully used these units, but they were not widely accepted. The experience with **BLIGHTCAST** illustrates one of the problems of using a disease forecaster. Although the program successfully predicted infection and its use generally reduced the number of fungicide applications, the direct financial saving to the farmer was relatively small compared to other production costs. Thus many farmers were not willing to risk using **BLIGHTCAST**, especially if it involved additional costs. However, as noted in the introduction, other pressures may force a reduction in the use of phytochemicals, making disease forecasters of this type more attractive to the grower.

**TOM-CAST** (Pitblado, 1992) early blight (**Alternaria solani**) on tomato — The forecaster for tomato early blight (**FAST**) developed by Madden et al. (1978) was modified for use in Southern Ontario, Canada. Like **BLIGHTCAST**, **TOM-CAST** identifies periods when environmental conditions are favorable for disease development and recommends a fungicide application schedule. Disease severity values (0, no risk — 4, high risk) were assigned to each day according to periods of wetness and temperature during the forecast periods. The first spray was applied when at least 35 disease severity values had been accumulated. Subsequent applications were recommended after the accumulation of at least 20 disease severity values. **TOM-CAST** required direct measurement of wetness duration in the field, which was possible using wetness sensors and data collecting systems such as those used with **BLIGHTCAST**. However, to encourage wider use of the forecaster Gillespie et al. (1993) examined methods of using synoptic weather data in place of direct wetness measurements. They concluded that with the appropriate calibrations, **TOM-CAST** could be used regionally by using locally observed daily maximum and minimum temperatures and dewpoint measurements made at two regional weather stations. This type of approach could provide a practical compromise between direct on-farm monitoring of crop microclimate (accurate but expensive) and general disease forecasts for whole regions (cheap but less accurate). Disease forecasters which use regionally estimated weather information could be operated by agricultural consultants serving a group of farmers in the same area.

**PLASMO** (Rosa et al., 1993, 1995) downy mildew (**Plasmopara viticola**) on grapevine — This forecaster has been developed to assist growers in Northern Italy reduce the number of chemical treatments needed to control downy mildew on grapevines. **PLASMO** is a sophisticated computer programme which simulates the critical phases in the pathogen’s life cycle (inoculation, incubation, and sporulation) using rate factors determined by environmental parameters (temperature, relative humidity, rainfall, and leaf wetness duration). The programme has been designed to be “user friendly” and is run interactively. The programme simulates infection development and allows the grower to decide whether or not spray treatment is needed. There is also an option to input the required weather data from automatic weather stations situated within the vineyard. Conventional control of this disease is done by application of sprays at regular 7- to 10-day intervals. In field trials in Tuscany, using weather data measured in the vineyard, spraying according to **PLASMO**’s recommendations reduced the number of treatments by 25 to 35% while attaining the same degree of disease control. Forecasters of this type are at the “top end of the market” and currently are probably only commercially suited to high-value crops. However, the computer revolution of recent years has produced personal computers capable of running extremely
sophisticated models. Simulation-based forecasters could easily be run on-farm or by agricultural consultant companies operating local environmental monitoring and disease prediction networks tailored to the farmers' needs.

Disease forecasting may not always improve the management of some diseases, for example, if the pathogen is always present and climatic conditions are always favorable then control measures will always be needed and forecasts may not be beneficial (Fry and Fohner, 1985). But, for diseases whose important occurrence is sporadic, disease forecasting should provide significant benefits (Fry and Fohner, 1985; Campbell and Madden, 1990). Currently, the use of disease forecasting as a tool in disease management is the exception rather than the rule. However, the need to reduce the use of phytochemicals should increase the adoption of rational decision support systems. Thus, disease forecasting incorporating environmental inputs will have an even more important role to play in crop protection programs and much effort is being devoted to developing new models for different crops (Table 3).

V. CONCLUSIONS

This chapter has highlighted the importance of environmental factors in the development of epidemics in crops and indicated how knowledge of the interactions of pathogen, crop, and environment can be of use in disease management. There are many areas which we have had to omit. For instance, the environment plays an equally important role in the epidemiology of soil-borne pathogens. Virus diseases of crops are also influenced by environmental factors, particularly though their effects on vector behavior. Knowledge of the effects of environmental factors on these pathogens is also needed to understand and control the diseases they cause. Disease can be affected by the growth of the crop and its physiological status; both are influenced by environmental factors. The effectiveness of crop spraying depends largely on the weather. Thus an understanding of the interactions between microclimate and spray droplets can help to improve the effectiveness of chemical control measures by better targeting and reduction of spray drift (see Matthews and Hislop, 1994, for a recent review of crop spraying techniques).

The environmental, social, and political pressure to reduce phytochemical use will probably continue to increase toward the millennium and beyond. It is clear that an understanding of the interactions between pathogens, crops, and environment can lead to more effective control of many crop diseases. However, for many important diseases these relationships largely remain unknown. There is, therefore, still much work to be done, and plant pathologists and environmental scientists will need to continue to work together to provide this information. Nevertheless, the potential for the development and widespread implementation of rational disease management strategies based on a sound understanding of the principles of disease processes has never been better: computing is becoming less expensive, more powerful, and more widely accepted; new methods of information handling such as decision support systems and expert systems are being developed; new methods for monitoring and estimating environmental variables are being developed. It is to be hoped that continued joint research will allow these factors to come together to produce environment- and farmer-friendly crop protection systems.

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