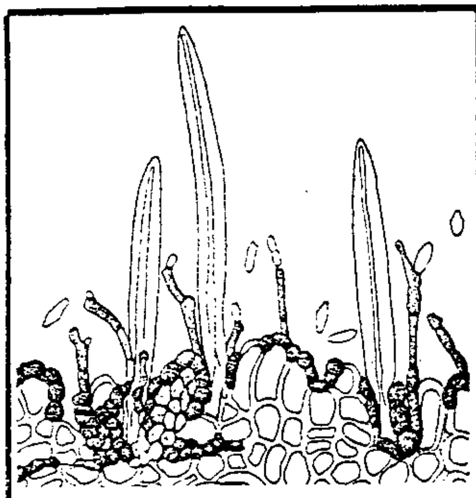
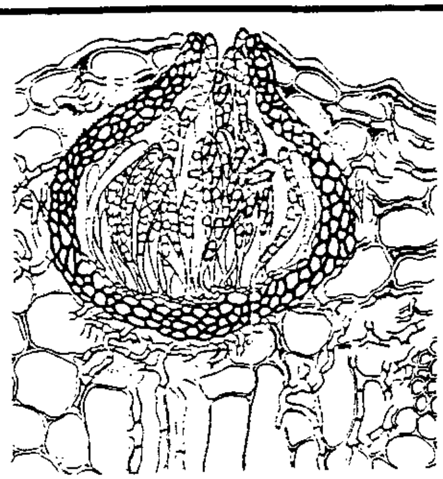
Volume XI Number 2


peach scab - *Cladosporium carpophilum*

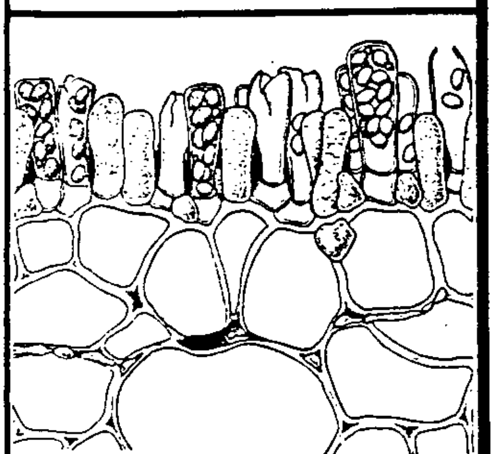
PLANT DIAGNOSTICIAN'S QUARTERLY

June 1990


apple scab - *Venturia inaequalis*

Features

Guidelines for Making Data Slides
Diagnosing Herbicide Injury on Crops
Charge Systems in Clinics
Fruit Blotch of Watermelon


peach leaf curl - *Taphrina deformans*

illustrations by Elsa O. Horn

Plant Diagnostician's Quarterly (PDQ) is a nonprofit publication which serves plant pathologists in extension, regulatory and industrial clinical laboratories, private consultants, and other interested persons. PDQ is published four times a year,: in March, June September, and December. Yearly subscription fees are:

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Volume XI, Number 2
June, 1990

PLANT DIAGNOSTICIAN'S QUARTERLY

FROM THE EDITOR

Hello again,

Well. A move to Indiana just prior to the growing season, a new job, a very wet spring, and samples up to the armpits have made for an exceedingly busy spring for your Faithful Editor. Hence this issue is a tad late.

Once more the APS meetings are rapidly closing in. I encourage you, if you are going, to attend the Diagnostics Committee meeting and the Diagnostics Committee Social. If you have not purchased your social tickets in advance, they will be available at the meetings. (All who wrote to me in response to the PDQ reader survey will have a chance to inundate me in person with all your ideas for a better PDQ.) I'll see you there!

The fact sheets that appear in this issue are courtesy of Mal Shurtleff; the originals have black and white photographs.

The TSWV enclosure can be ordered from Anne B. Sindermann, Maryland Department of Agriculture, 50 Harry S Truman Parkway, Annapolis, MD 21401.

As always I remain

Your Faithful Editor,

Melodie Putnam

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Cover by Ethel Dutky

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DIFFUSION

DNA relatedness of xanthomonad strains. D.C. Hildebrand and M. N. Schroth of the Univ. of California, Berkeley; and N. J. Palleroni, NY Univ. Med. School, have used S1 DNA:DNA hybridization to examine the relatedness of 23 *X. campestris* pathovars and *X. fragariae*. The taxonomy of this genus has, like other plant pathogenic bacteria genera, been a source of disagreement for years: Is the ability to attack a particular host a valid taxonomic criterion? The answer, according to this study is no: "...xanthomonads which attack members of the same host family usually show only a distant genetic relationship." Look for more taxonomic revisions of *X. campestris* in the future. Journal of Applied Bacteriology 1990, **68**:263-269.

Unusual or interesting records of plant pathogenic Oomycetes. G. Hall of the CAB Int. Mycological Inst. in Kew, UK, has provided a list of pathogens recovered from various plants: From the ornamental shrubs *Hebe gauntlettii, H. elliptica x pimelioides*, and *H. hulkeana* *Peronospora grisea* was isolated. *P. fragariae* was recovered from wild growing *Potentilla recta*, suggesting this pathogen may be able to attack commercial species of this genus. *Peronospora hariotii* was found growing on *Buddleia globosa* and *B. davidii* cv. Pink Delight, two other species of ornamental shrubs. *Pythium tracheiphilum* was isolated from cultivated lettuce. All of the above were found in England. Records from other countries include *Phytophthora erythroseptica* causing a leaf spot on *Cymbidium* sp. (Perth, Australia); *P. palmivora* from roots of *Zizyphus jujuba* (Taiwan); and *Plasmopara halstedii* from leaves of *Arctotis* (Auckland, New Zealand). Plant Pathology 1989, **38**:604-611.

Control of grey mold with anti-desiccants. Plastic polymers used as anti-transpirants were found by N. Ayish, *et al*. (The Volcani Center, Bet Dagan, Israel) to reduce the amount of grey mold on various plants under artificial and green house conditions. Wilt Pruf, Biofilm, Colfix, Vapor Gard, and Safe Pack were tested on cucumber, bean, tomato, pepper, cut rose, and geraniums using either detached leaves or whole plants. Plant material was inoculated with benzimidizole and dicarboximide resistant *Botrytis cinerea* cultures. Green house experiments were performed with cucumber plants. In both experiments inhibition of grey mold was compared to control obtained using diethofencarb plus carbendazim. All polymers tested "significantly reduced grey mould on detached leaves...." In the green house, the polymers significantly reduced disease of senescent female fruits of cucumber. The authors concluded that "This study has demonstrated that film-forming polymers may serve as an alternative means to protect crops against grey mould...." Plant Pathology 1990, **39**:249-254.

Mesoconidia of *Fusarium*. Heads up *Fusarium* fans! Now, in addition to microconidia and macroconidia there are mesoconidia! *Fusarium* taxonomy, never a dull subject, just became even more interesting. I. G. Pascoe (Plant Res. Inst., Victoria, Australia) has identified a morphologically and developmentally distinct conidium which as been "ignored or scarcely recognized" previously (one wonders why?). These conidia are "produced singly and holoblastically in the aerial mycelium" from sympodial conidiogenous cells. Mesoconidia and mesoconidiophores are described for *F. avenaceum, F. chenopodium, F. chamydosporum, F. pallidoroseum, F. sporotrichioides*, and *F. subglutinans*. Mycotaxon 1990, **37**:121-160.

TENTATIVE AGENDA
DIAGNOSTICS COMMITTEE
APS, GRAND RAPIDS, MI
AUGUST 5, 1990

1-Recognition of Committee membership for 1990/91 and thanks to those rotating off of the Committee

2-Nomination of Vice-Chair (to serve as chair in 1992)

3-Reports on Workshops for improvement of diagnostic skills
  a. Rhizoctonia Workshop (August 3rd at MSU)
  b. Rapid diagnostic assays for plant pathogens
  c. Ideas for future workshops-e.g. a Pythium workshop for the 1991 annual meeting; a Fusarium workshop

4-Diagnostics Committee Social - Aug 6 from 5-7 pm

5-Diagnostic Manual Progress Report

6-Diagnostic Lab Roster

7-Update on Diagnostic Reference Pathologist Listing

8-'Spotlight on Diagnosis' report

9-PDQ report

* * * * *

If you would like to add items to this agenda, please contact:

Margery Daughtrey
Diagnostic Committee Chairman
L. I. Horticultural Research Laboratory
39 Sound Avenue
Riverhead, NY 11901

ANNOUNCING THE DIAGNOSTIC COMMITTEE SOCIAL (plus slides!) AT THE 1990 APS MEETING

August 6, 1990

Since diagnosticians are sociable and have an insatiable curiosity about plant disease identification, we would like to offer the opportunity to satisfy both instincts at the forthcoming APS meeting in Grand Rapids. Anyone interested in plant disease diagnosis is welcome to attend the social: the more the merrier!

We will plan to socialize freely from 5-7 pm. In the corners of the room there will be Karamate projectors with slides of photogenic diseases, which will serve as conversation pieces.

Please plan now to bring with you five of your favorite slides, showing some of the interesting diseases that you have encountered in recent years---and perhaps a few slides of problems that you never correctly identified. See if you can stump the other experts in the room. Slides of symptoms on all types of plants will be welcomed.

Since others in the group will quite likely be interested in obtaining copies of some of these slides, it would be best if you would make a copy of each slide in advance, so that requested copies could be make from the slides you bring to the meeting (without requiring you to go without your only copy for several weeks during the copying/distribution process).

See you (and your slides) there!

Guidelines for Preparing 35 mm Transparencies that are Legible when Projected

P. W. Reeser

Certainly there *are* worse things than unreadable data slides at a seminar, but it *is* frustrating to have to squint at a slide that has been ill prepared. It is little consolation when the speaker assures you that the only important information on the slide is that contained on lines 3 and 55.

If you have ever resorted to the use of a reduction photocopier to get an abstract in that little blue box on the APS abstract submission form, you are already aware of the capabilities of modem print technologies. However, as the purpose of a presentation is to deliver meaningful information to an audience, we would do well to prepare our slides so that the information is clear, concise, and legible to the most distant viewer in the room. My intent here is to present a collection of easily followed guidelines which should assist you in this endeavor.

There are some excellent references available which can help you prepare data slides which are meaningful and legible when projected. Singha and Gartin make brief mention of these in their paper on improving the effectiveness of graduate seminars. In addition, Eastman Kodak has published extensive information on this topic, and much of what I present here is a synopsis of their work.

Rule No. 1

At a minimum, the information on your chart or graph should be legible at a distance no less than 8 times the height of the original.

For example, place an 8 1/2 x 11 inch page of typewritten text approximately 7 ft from your eyes (11 inches tall x multiplier of 8 = 88 inches or about 7 ft). If you have trouble reading this, then your audience will also have little chance of reading the text, especially since viewing conditions are usually less than optimal ("Is this in focus back there?").

Rule No. 2.

If you are using 10 or 12 characters per inch (cpi) print for titles, labels, text, or tables, the area in which you work should be limited to 3 x 4 1/2 inches. Oriented horizontally, this will allow about 40-48 characters across, with a 1/4 inch border all around. Use Rule No. 1 to verify that this is legible from a distance of 24 inches (3 inches x 8 [multiplier] = 24 inch viewing distance).

Rule No. 3

In general, character height should be no less than 1/50 the height of the information area, but a more conservative 1/25 might be better to accommodate those sitting in the back of a large meeting room. Measure the lower case characters, excluding those with 'tails,' such as q,y,p,g.

For example, the text you are reading right now uses lower case characters which are 1/16 inch tall. Using the 1/25 standard, the height of the information area should be 1/16 inch x 25 = 1 1/2 inches! By the same reasoning, the 1/50 standard would allow an information area 3 inches in height. Remember that the 35 mm slide format is a 2:3 height:width ratio, so dimensions of the information areas we just calculated would be 1 1/2 x 2 1/4 inches and 3 x 4 1/2 inches, respectively. Figures 1, 2 and 3 demonstrate that you can fit quite a bit of information in these small areas.

In applying this rule be conservative. Audio-visual facilities in meeting rooms vary and you usually cannot predict what the projected image size or maximum viewing distance will be. Be brutal and cut out all unnecessary information, or split the information over two or more slides.

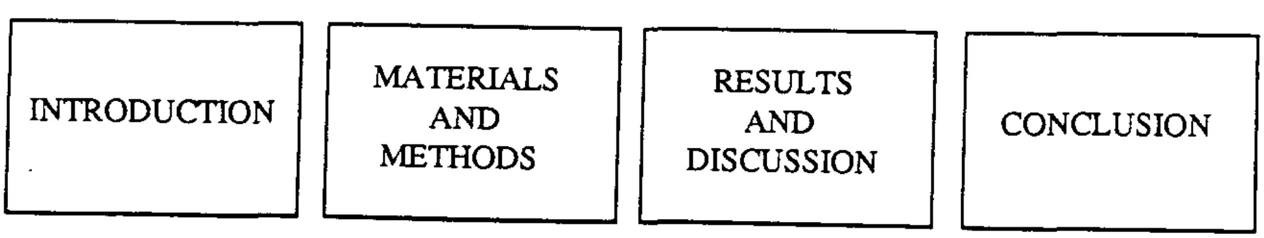

Figure 1. This is a 1 x 1 1/2 inch size template for titles or other short statements.

METHOD

1. Collect fresh sample
2. Refrigerate at -80° C for 3 hr
3. Grind to powder
4. Add 30 ml 95% EtOH

1. Plant pathology is a dying field
2. Keep one foot in the furrow
3. That is an abomination up with which we shall not put.
4. Nobody likes a sore loser.

Figure 2. An example of the 2 x 3 inch size for brief lists or messages. See text under Rule No. 4.

Table 2. Effect of various atoxigenic strains of Fusarium roseum on zearalenone contamination of corn seed by a toxigenic strain.

Atoxigenic strain	Zearalenone (µg/g)
None	65.12
22	38.24
13	21.79
11	14.03
85	8.62
50	5.22
33	0.89

Figure 3. An example of the amount of information that can be inserted into a 3 x 4 1/2 inch space.

Rule No. 4.

Standardize the format and size of artwork, text, charts, and graphs. This will save both cost and headaches in photographing the work, and will make for a more consistent and professional looking presentation.

For example, dimensions of the work area should be in a 2:3 ratio, the same as a frame of 35 mm film. All work should be oriented horizontally to avoid images being clipped above or below (or both) when projected. Use the same background for all slides.

I have seen people make title slides of single lines of text with varying numbers of characters per line, then photograph them all to fill the width of a 35 mm slide frame. This method results in a series of slides of various character sizes for related information as shown in Figure 4 (see next page). Instead, make a series of templates of various sizes for various types of material. For a typewriter or printer that produces 10 or 12 cpi, template sizes might range from 1 x 1 1/2 inches to 3 x 4 1/2 inches. Use the smaller size for single words or short phrase slides such as in Figure 1. Use 2 x 3 inches for brief lists or short take-home messages as in Figure 2. The 3 x 4 1/2 inch size template should allow plenty of room for data tables, graphs, charts, etc., as in Figure 3. Finally, if you are printing a full page from a computer generated

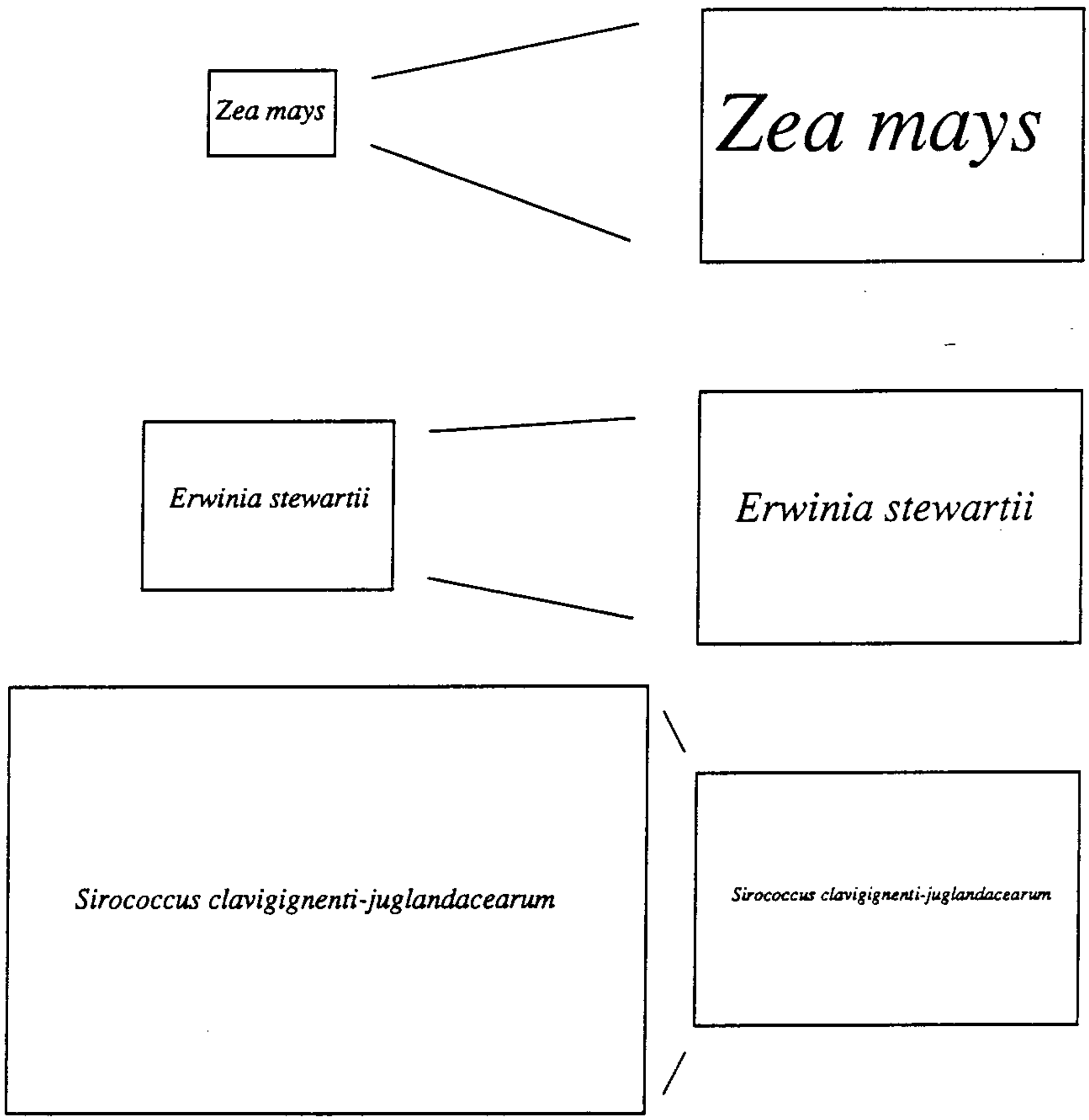

Figure 4. An example of poor planning. The boxes on the left have been sized for the amount of information, not taking into consideration how the slides will look when projected, as demonstrated on the right.

graphics program, fit the chart or graph in a 6 x 9 inch area (oriented horizontally) but be sure that character heights are not less than 1/8 inch.

Rule No. 5

Photograph all print material full frame otherwise all your efforts at templates, size standards, and proper dimensioning will be for naught. Full frame means that the image of the object you are photographing fills the viewfinder on the camera, and will likewise fill the frame on the slide. If you lack close focusing capabilities in you camera system, use an enlarging photocopier to increase the image size first.

Rule No. 6

All the above rules apply also to computer generated graphics that are photographed off the screen, with a few corollaries.

a. Position yourself at a distance from the monitor at least 8 times the height of the image you are producing to assess legibility. Be sure to use the font size recommendations included with the software.

b. Avoid the temptation to cram too much on a single chart or graph.

c. Colorful palette choices are nice, but remember that certain colors are incompatible and will render otherwise adequate artwork illegible (e. g. red or purple lines on a dark blue background, yellow on white, etc.).

If you want more information or more detailed explanations of some of the guidelines presented here, consult the Eastman Kodak literature. In the meantime, try applying some of these guidelines to your current work and see if the quality of your presentations is not improved.

References

Singha, S. and Gartin, S. A. 1988. Improving the Effectiveness of Graduate Seminars. HortScience 23:243-245.

Encyclopedia of Practical Photography. Vol. 12. Edited by Eastman Kodak and Amphoto. Garden City, NY: American Photographic Book Publishing Co. PP. 2064-2070 & 2250-2264.

DIAGNOSING HERBICIDE INJURY

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Dept. Botany & Plant Pathology
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When diagnosing herbicide injury consider all possible causes, including environmental and mechanical factors, as well as disease and insects. Observe the condition of the plant. Also, look for the effectiveness of the herbicide on weeds in the treated area. Herbicide injury will usually occur in definite patterns within a field. Look for:

1. drift patterns across a field
2. overlapped rates at the end of rows
3. uniformly injured strips caused by application equipment
4. differential injury across soil types (injury on light soils such as sandy knolls and/or lack of weed control on heavy soils such as in low spots in the field).

Primary Sites of Action and Injury of Some Common Herbicides

Most herbicides can be grouped according to their mode of action (the type of injury symptoms they express). By knowing a few characteristics of each group of herbicides, the injury caused in a particular field can be identified through a process of elimination. The specific herbicide may not be readily determined, but all herbicides that do not produce the characteristic injury symptoms can be eliminated. This leaves a much smaller group of herbicides to consider. Many new trade names of herbicides on the market today are simply prepackaged mixtures of 2 or more herbicides (i.e. Bicep, a mixture of Dual and atrazine). Several of these mixtures combine herbicides from different mode-of-action groups. Using the example above, Bicep is composed of Dual, a cell growth inhibitor, and atrazine, a photosynthetic inhibitor. While injury to a given crop could be caused by each representative herbicide of the mixture giving a variety of symptoms, it is most often noted that one class of herbicidal injury will be prominent. By checking records of the fields in question or by determining what was applied in adjacent fields, the herbicide which caused the injury can usually be identified.

1. Photosynthetic Inhibitors

Action: These herbicides are most often applied to the soil and are absorbed by plant roots. The herbicide moves with the flow of water into the foliage by the xylem system (systemic) but does not translocate back down into the roots by the phloem system. When photosynthetic inhibitor herbicides are used postemergence, their action is contact and requires thorough wetting of the foliage, usually with the aid of an adjuvant. Injury occurs on leaves and above ground stems. No damage is caused to the root system. Plants must germinate and turn green before they die.

Injury: Plants turn yellow then die back from the bottom to the top. Leaves turn yellow between the veins then begin dying from the tip toward the base and from the outer edges toward the center. Leaves will fall off the plant, leaving only a stem with an apical bud.

Specific Herbicides: Atrazine, Bladex, Sinbar, Lorox, Princep, Sencor/Lexone

Premixes: Bicep, Extrazine II, Marksman, Preview, Salute, Laddock, Lariat, Lorox Plus, Prozine, Rhino, Sutazine, Turbo. NOTE: The premixes listed for each mode-of-action group contain at least one representative herbicide from that group. The other herbicides in the premix may be from another group. Therefore, if symptoms do not fit the particular herbicide from that group, then refer to characteristics of other groups.

2. Cell Growth Inhibitors

Action: Herbicides work on the germinating seedlings and stop growth of the roots and/or shoots before they emerge.

Injury: Symptoms may be expressed in the roots and/or the shoots, but mostly in one or the other. Those herbicides that affect the root system usually do not cause any visible damage to the above ground system other than stunting or discoloration of foliage, while those that affect the shoot system usually cause no visible damage to the below ground system.

Root injury symptoms: Injury may appear as root pruning and the inhibition of secondary roots. Roots may be swelled or club-shaped. The underground portion of the stem will be thickened and shortened. Stems of soybean plants may become callus and brittle at the soil surface.

Specific Herbicides: Treflan, Surflan, Balan, Sonalan, Prowl

Premixes: Salute, Squadron, Commence, Tri-Scept, Prozine, Pursuit Plus

Shoot injury symptoms: Corn shoots appear twisted and leaves tightly rolled. Stems can become ruptured, with new growth protruding out of the ruptured tissue. Soybean leaves are dark green, crinkled, and/or leaf tips flattened. Leaves may fail to unfold from buds. Root damage may also occur, but not very often.

Specific Herbicides: Lasso, Dual, Eptam, Sutan, Vernam, Genep, Genate, Eradicane,

Premixes: Bicep, Rhino, Sutazine, Turbo, Lariat

3. Growth Regulators

Action: Growth regulator herbicides can be taken up from the soil by the root system. However, most growth regulator herbicides are used as postemergence treatments, thus cause more damage to the shoot system than to the already established root system. When growth regulator herbicides are used as pre-plant treatments in reduced or no-tillage systems, root damage can occur.

Injury: Soil absorbed herbicides can cause both root and shoot injury. Usually Banvel injury to soybean shoots is more severe than 2,4-D or 2,4-DB at equal rates. Both 2,4-D and Banvel cause plants to turn darker green than normal. In corn, the brace roots will fuse together and the shoot will "buggy whip" (onion leaf).

Root injury symptoms: A proliferation or clusters of short secondary roots are visible along the tap root of soybeans. Growth of secondary roots (brace roots) on corn is inhibited and usually the brace roots are fused together into a leaf-like structure. With Alanap, roots may turn upward as if growing out of ground.

Specific Herbicides: 2,4-D, Banvel, Amiben, Alanap

Premixes: Rescue

Shoot injury systems: 2,4-D and 2,4-DB cause bending and twisting of soybean stems. 2,4-D causes leaf strapping (feathering) of soybean leaves and brittle stems of corn. Banvel additionally causes new growth of soybean leaves to cup upward. Dalapon will give many of the same symptoms as both cell growth inhibitors and growth regulators. It will cause the plant tissue to become dark green and, in soybeans, the leaf damage will look similar to Banvel injury.

Specific Herbicides: 2,4-D, 2,4-DB, Banvel, Dalapon

Premixes: Rescue

4. <u>Cell Membrane Disruptors</u>

Action: This herbicide class consists mostly of postemergence contact herbicides, although some (i.e.Modown, Goal) can be applied preemergence. Thorough coverage of foliage is needed for complete weed control. Toxicity increases with high temperatures and direct sunlight. Incomplete coverage or spray drift can cause spots of dead tissue which may be confused with plant diseases.

Injury: Injury symptoms include desiccation of leaf tissue (leaf burn) caused by disruption of cell membranes (paraquat, Blazer, Cobra, Reflex). The soil applied herbicide of this class, Goal, destroys the membranes of germinating seeds. Applications of Cobra to soybeans may cause crinkling of new growth similar to Lasso or Dual injury, besides the characteristic leaf speckling or burning symptoms.

Specific Herbicides: Gramoxone Super, Basagran, Blazer/Tackle, Cobra, Reflex, Goal (NOTE: Basagran is actually a photosynthetic inhibitor, however herbicidal action occurs so quickly that it is classified under this mode of action group)

5. <u>Growing Point Disintegrators (Post Grass Materials)</u>

Action: This herbicide class is active on only grass species, while broadleaf plants are tolerant to these herbicides. Although some of these herbicides have shown minimum soil activity, the major activity is from postemergence applications. Most of the herbicides have both annual and perennial grass activity, however sensitivity will vary with species. Movement of the herbicide within the plant occurs in both the xylem and phloem.

Injury: Root and shoot growth ceases very rapidly and plants begin to turn color (usually red to purple). In about 7 to 14 days the growing points decay. The outward appearance of the plant will indicate the grass is still alive, but removing the top leaves from the whorl will show that the growing point is dead and rotten. Cutting through each node will reveal that the meristem tissue is dead and disintegrating. In perennial grasses the same decay will be found in the nodes of the underground stems (rhizomes).

Specific Herbicides: Poast, Fusilade, Option, Assure

6. <u>Amino Acid Inhibitors (Slow Death)</u>

Action: The herbicides in this group are effective on both broadleaf and grass weed species. Roundup is nonselective on most all species, Scepter is selective in soybeans and alfalfa, and Harmony is selective in wheat and barley. The herbicides prevent the production of essential amino acids, which cause the plants to slowly starve to death.

Injury: The affected plants stop growing and stay green for several days. Roundup and related compounds may show a small amount of bleaching of the foliage of grasses and sedges around the new growth. Most plants die slowly and turn a uniform harvest death color (golden in grasses and flat green in broadleaves). The meristematic tissue (nodes) do not deteriorate like those treated with the postemergence grass herbicides. Scepter injury symptoms on soybeans may include shortened internodes and possibly a reduced root system. With Classic herbicide, temporary yellowing and/or retardation of growth may occur on soybeans under certain conditions. Injury to corn from Scepter and Classic carryover may include one or all of the following injury symptoms: stunted plant, interveinal chlorosis, purpling, red-purple midrib of lower leaves, reduced root system including bottle-brush appearance of lateral roots. NOTE: Both Scepter and Classic inhibit the same enzyme within the plant, therefore, these herbicides will display the same injury symptoms.

Specific Herbicides: Roundup, Arsenal, Scepter, Glean, Classic, Oust, Harmony, Pursuit, Pinnacle

Premixes: Squadron, Tri-Scept, Preview, Lorox Plus, Pursuit Plus

7. <u>Pigment Inhibitors</u>

Action: These herbicides are mostly applied as preplant or preemergence treatments. Amino Triazole is used postemergence for control of annual and perennial weeds, especially poison ivy. These herbicides inhibit carotenoid synthesis, but do not affect preexisting carotenoids. Therefore, new growth turns white while growth prior to treatment does not. Carotenoids protect chlorophyll from light (photo-oxidation), therefore, destruction of chlorophyll occurs, since chlorophyll is not protected by the carotenoid pigments. The herbicides used in row crops can persist long enough to affect rotational crops the following year after application.

Injury: The primary injury to crops is the bleached white appearance of leaves. In soybeans this appearance may be interveinal. In some cases the affected areas will be highlighted with a pink to red margin.

Specific Herbicides: Amino Triazole, Sonar, Zorial, Command

Premixes: Commence

A Partial Review of Charge Systems in Plant Diagnostic Clinics

Compiled by:
Gail Ruhl
Purdue University

The question of charging for diagnostic services has been of growing concern due to the limited operating budgets of most plant clinics. Some clinics feel that fees for services may be necessary to offset operational costs. Some clinics already charge for nematode assays, while some charge for all samples submitted. Some clinics charge additional fees for testing services such as dsRNA analysis, serological assays, mycotoxin testing, and the use of specialized culture media.

An opinion survey on charging was mailed to PDQ subscribers in all 50 states several years ago. I recently updated the survey for extension based clinics and felt it might contain information of interest to PDQ readers. **If your state is not represented in this review, or if you find errors in the charges listed please send me the charge information pertaining to your clinic, and we will amend this listing in the next issue of PDQ.**

Respondents to Charge Survey

Clinic Category	Number of persons responding	Number. of facilities represented	Number that charge*
Extension (Univ. Labs.)	45	43	12 (18)**
State Regulatory Labs	10	10	2
Industry Labs	7	7	5
Research Stations (Canada)	2	2	0

*Charge only for disease diagnosis
**Charge for disease diagnosis, nematode assays, endophyte analysis, etc...

Charge Policies for Extension-University Supported Diagnostic Laboratories (EDL) (Contacted in 1990)

University	Charges	
Arkansas	$5	•For any sample not sent thru agents
	$15	•Virus analysis
	$10-$25	•For commercial samples (depends on work)
Cornell	$5	•Noncommercial-no culture
	$10	•Noncommercial-culture
	$10	•Commercial-culture
	$10	•Nematodes

Cornell (tentative proposed fee system for test basis - Oct. 1990-91)	$20	•All insect, disease & weed diagnosis (culture + visual)
	$40	•All virus I.D. (ELISA/Host Range)
	$40	•Nematode I.D.
Georgia	$5	•Aflatoxin
Guelph	$8	•Visual diagnosis
	$32/hr	•For extensive work (maximum $64-2hrs) charged in 15 min. increments.
Hawaii	$5	•General disease diagnosis/bioassay for virus or fungi
	$10	•Disease analysis for bacteria/pathogens
	$20	•Electron microscopy for virus I.D.
	$5	•Nematode I.D. to genus
Illinois	$5	•Regular diagnosis
	$10	•Pinewood nematode
	$10	•SCN I.D.
	$20	•Other nematodes
	$25	•SCN race determination
W. Kentucky	$5	•SCN I.D.
	$20	•SCN race I.D.
	$20	•Endophyte analysis (Regulatory Service Division in Lexington)
Kentucky	$5	•SCN I.D.
	$20	•SCN race determination
Kansas	$10	•Fescue endophyte analysis
Minnesota	$15	•Routine disease diagnosis
	$10	•Nematode egg count
	$8	•Larval count
	$50	•Race I.D.
Missouri	$0	•No general charge
	$0	•Free bioassay - SCN
	$25	•Samples sent to AGDIA
	$10	•Egg counts (Nematode lab is separate)
	$25	•Race I.D
	$15	•Complete profile
	$20	•Species I.D.
Mississippi	$5	•Nematode I.D. for chemical companies & out of state samples
New Mexico	$0	•No charge for disease diagnosis
Nebraska	$0	•No charge for disease diagnosis

North Dakota	$4	•Visual diagnosis for samples sent thru extension agents
	$8	•Visual diagnosis for samples sent directly (not thru agents)
	$15	•Visual-out-of-state
	$10	•Dutch Elm Disease
	$19	•Cultured diagnosis for county agents
	$23	•Cultured diagnosis for in-state samples not sent thru agents
	$30	•Cultured diagnosis for out-of-state samples
Oklahoma	$5	•Disease diagnosis/nematode I.D.
Ohio	$5	•General diagnosis/for extension agents
	$12	•General diagnosis for commercial growers, homeowners who do not go thru agents & walk-ins
	$15	•Lawn/turf samples
	$20	•ELISA
	$30	•dsRNA analysis
	$25	•Fatty acid analysis
	$15	•Nematode I.D.
Purdue	$15	•Nematode I.D.
Tennessee	$0	•No charge if submitted by extension
	$0	•No charge if submitted by others - visual
	$10	•Submitted by others - incubated, cultured, etc.
	$20	•Submitted by out-of-state
	$5	•Nematode I.D. from soil
	$15	•Nematode I.D. from soil (out-of-state)
	$15	•SCN race determination
	$25	•SCN race determination (out-of-state)
Texas	$0	•No charge if submitted thru county agent
	$10	•Homeowner & commercial samples submitted directly
		•Variable rates virus testing
Wisconsin	$4	•Disease diagnosis thru a county agent
	$5	•Disease diagnosis is for all others
	$20	•Potato ring rot
	$15	•Aphanomyces in soil
	$22	•All nematode I.D.

Who Handles the Billing & Money

Secretary 5
Secretary & Business Office.. 5
Business Office 3
Prepaid forms & Secretary.... 1 (Cornell)
Clinic Director.................. 1 (Tennessee)
Unknown 2

What Type of Account is Set-Up?

Revolving 5
Dedicated Diagnostic Lab Acct..... 3
Univ. Assigned Acct................ 3
Extension Acct........................ 3
4-H Fund.............................. 1
Unknown.............................. 2

Benefits From Charging

1. Decent samples
2. Defray cost (does not totally fund the clinic)
3. Fees support summer help
4. Fees help buy equipment, supplies, books
5. Fees help with diagnostician's travel to national meetings and workshops
6. Automatic phone billing (MN)
7. Deterrent of nuisance samples
8. Positive expression of the need for a clinic

Problems With Charging

1. Bad public relations
2. 'Red Tape'
3. Hassle to bill and receive payments
4. More complaints about 'poor' service
5. May lose extension support funds and then will not have enough money to run the clinic
6. Collecting $ from other extension specialists and walk-ins
7. Fees do not cover operating expenses
8. Not enough income to be worth the hassle
9. Need to advertise to inform public of the service
10. If the client checks the sample in with the secretary then client cannot discuss the sample with the specialist.
11. Refunding money
12. Time consuming
13. Do not have control over the money brought in
14. Different fees are cumbersome
15. Complicated systems should be simplified-have a sheet describing fee systems
16. Payments that do not come with samples
17. Lack of cooperation from county extension agents and other univ. personnel!
18. One needs a computer for letter generation with a notice for payment which automatically appears at the end of the letter.
19. Need a flat rate and then a high hourly rate for time consuming samples

BACTERIAL FRUIT BLOTCH OF WATERMELON

Donald L. Hopkins

University of Florida, CFREC-Leesburg

In the 1989 season, a bacterial fruit rot disease of watermelon was observed for the first time in commercial fields in the U.S.A. A disease with similar symptoms was observed by Crall and Schenck in 1967 and 1968 on the University of Florida research center farm in Leesburg, but it has not occurred there since 1968. This disease occurred in the Mariana Islands in 1987 and had been observed three years earlier in Australia, where it was referred to as 'fruit blotch'.

Last year fruit blotch was found in many states in the East including Florida, South Carolina, North Carolina, Maryland, and Indiana. Fruit blotch occurred in a few widely scattered fields in these states. However, the disease was devastating in some fields with 50-80% of the fruit rendered unmarketable. Florida and South Carolina growers were hardest hit by this disease. Although there can be earlier symptoms, many of the Florida growers that suffered 50-80% losses at harvest had observed no symptoms in their fields 10-14 days prior to harvest.

The causal bacterium is a nonfluorescent pseudomonad that is similar to a bacterium that has been reported to cause a disease of watermelon seedlings, *Pseudomonas pseudoalcaligenes* subsp. *citrulli*. However, the taxonomic classification of this bacterium is probably incorrect, based on DNA-rRNA hybridization studies.

Disease diagnosis

Symptoms on the fruit begin as small, greasy-looking, water-soaked areas of a few millimeters diameter that enlarge to several centimeters diameter with irregular margins. Lesions originate on the upper surface of the fruit and, at first, do not extend into the flesh of the melon, but are primarily surface lesions. As the lesions age, the melon surface often cracks and bacterial ooze can be seen on the fruit. Fruit decay follows in many of the melons and can be a problem in post-harvest shipments. Symptoms are also produced on watermelon seedlings and consist of water-soaking of the lower surface of cotyledons and leaves, followed by necrotic lesions frequently with chlorotic halos. These foliar symptoms may be inconspicuous in the field.

With a microscope, bacterial flows can be seen from small pieces of rind tissue from the melon with fruit blotch. To isolate the bacterium, surface disinfest the rind with alcohol and cut a small piece of rind tissue from the margin of the rind blotches. Crush the piece of tissue in a small amount of water (1 ml) and streak onto the surface of nutrient agar or King's medium B agar. Incubate the plates at room temperature or in an incubator at 25-30C. Bacterial colonies should grow in 24-48 hrs. King's medium B is valuable because you can distinguish any fluorescent pseudomonads that might also occur. Pure cultures of the fruit blotch bacterium are usually obtained with this technique. Colonies on these media are white and transparent. The fruit blotch bacterium produces a hypersensitive reaction in tobacco.

While the symptoms along with isolation of a bacterium that is nonfluorescent on King's medium B and positive for hypersensitivity on tobacco may be diagnostic for watermelon fruit blotch, confirmation can be obtained by inoculating watermelon seedlings. Seedlings in the first or second true-leaf stage can be inoculated by misting the leaves and cotyledons with inoculum (10^6 colony forming units per ml) or by rubbing cotyledons with an inoculum-saturated cotton gauze pad. Stem and hypocotyl inoculations can also be done by injecting 0.1 ml of inoculum with a syringe. Inoculated plants should be incubated at 100% relative humidity for 48 hrs. Leaf and cotyledon symptoms are as described above and usually appear 3-4 days after inoculation. Stem inoculation results in water-soaking followed by stem collapse.

References

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2. Schaad, N. W., G. Sowell, Jr., R. W. Goth, R. R. Colwell, and R. E. Webb. 1978. Pseudomonas pseudoalcaligenes subsp. citrulli subsp. nov. Int. J. Syst. Bacteriol. 28:117-125.

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Cooperative Extension Service

report on
PLANT DISEASES

DEPARTMENT OF PLANT PATHOLOGY
UNIVERSITY OF ILLINOIS AT URBANA-CHAMPAIGN

No. 1108
(4/86)

NEMATODE PARASITES OF TURFGRASS

T.A. Melton, R.B. Malek, and M.C. Shurtleff

Parasitic nematodes feed on all species of turfgrasses throughout the world and may be limiting factors in the production of high-quality turf in Illinois. Although the damaging effects from nematodes are not as well established for turfgrasses as they are for many other crops, several species of nematodes are known to cause serious damage to bentgrass and bluegrass, especially when the turf is also stressed by other factors. Other species may feed on turfgrasses without causing noticeable damage. Often nematodes are overlooked when turfgrass problems are being diagnosed because (1) they are translucent and microscopic and virtually all live in the soil; (2) they generally reduce plant vigor but only cause severe symptoms during times of stress; and (3) most diagnoses are based on plant shoots rather than roots, soil, or both. Recognizing nematode problems and understanding the relationships of these parasites to turfgrasses are essential for growing high-quality turf.

Figure 1. Nematode-infested turfgrass showing typical chlorosis and lack of vigor in untreated strips and foreground compared with healthy turfgrass in the darker, nematicide-treated strips. (Photo by G.C. Smart)

Plant-parasitic nematodes are microscopic, translucent roundworms, generally 0.5 to 3 mm (1/50 to 1/10 inch) in length and 30 to 100 μm (1/1000 to 1/250 inch) in diameter as adults. All are vermiform (worm-like) at least during the early stages of their life cycles. Females of root-knot *(Meloidogyne)* and cyst *(Punctodera)* nematodes grow to be pear- and lemon-shaped, respectively, at maturity. Most nematodes have well-developed digestive and reproductive systems. The digestive system from anterior to posterior is composed of the oral opening, stylet, esophagus, intestine, and anal (female) or cloacal (male) opening.

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Possession of a stylet separates plant-parasitic from nonparasitic forms, although many stylet-bearing species feed only on lower plants, such as the fungi associated with turfgrass roots. The stylet is a hollow, spear-like structure in the head region that is used to probe and penetrate cells, egest digestive enzymes into plant tissues, and ingest cell contents.

Figure 2. Turfgrass severely affected by nematodes. Note the thinning and dying in small to large irregular areas. (Photo by L.T. Lucas)

SYMPTOMS AND DAMAGE

Above-ground symptoms of nematode injury may include a slow green-up in the spring, chlorosis and dieback of the grass blades, sensitivity to stresses, reduction in vigor, stunting, and gradual thinning of the turf (Figures 1 and 2). Symptoms usually occur in gradually enlarging circular to irregular areas because of the clustered distribution of nematodes (Figure 2). Damage is most obvious during periods of moisture and temperature stress. Severely affected plants may wilt during the heat of midday and recover at night. These symptoms may be indicative but are *not* diagnostic of nematode problems. Nematode damage varies considerably with environmental conditions and is easily confused with insect injury, other diseases, nutrient and moisture stresses, pesticide injury, thick thatch, compaction, or other turfgrass problems. Nematode-infested roots commonly are shallow, coarse (lack feeder roots), bushy, or stubby (Figure 3). Suspect a nematode problem if the turf does not respond normally to applications of fertilizer, water, and fungicide or to increased aeration in compacted or heavy soils.

Figure 3. Roots damaged by stunt nematodes. Note that lower lateral roots are sparse, short, and shriveled, resulting in a shallow, coarse root system. (Photo by George Powell)

Symptoms of root injury (Table 1) vary depending on the species and number of nematodes and the other stresses that are involved. Root-knot (*Meloidogyne*, see *Report on Plant Diseases* No. 1101) and cyst (*Punctodera*) nematodes alter the grass physiology by egesting specific enzymes that induce giant cell formation within the root at the feeding site. Giant cells act as sinks by "attracting" energy-rich plant metabolites, which are consumed by the nematode. The abnormal cells disrupt moisture and nutrient transport within the plant. Some nematodes inhibit root growth. For example, stubby-root nematodes (*Trichodorus*) feed at

Table 1. Nematode Genera Associated with Turfgrass Problems in the Midwest, Their Importance in Illinois, and Their Types of Root Damage

Genus name (common name)	Importance*	Root symptoms
Helicotylenchus (Spiral)	1-II-B	Roots shriveled, short and discolored; cortical tissue sloughs off
Tylenchorhynchus (Stunt)	1-II-B	Roots shriveled; short and sparse
Pratylenchus (Lesion)	2-I-B	Small, brown to black spots to large girdling lesions; severe root rot
Hoplolaimus (Lance)	2-I-B	Slight swellings and brown lesions; cortical tissue sloughs off
Xiphinema (Dagger)	2-I-B	Roots sparse and discolored; lack of feeder roots
Criconemoides (Ring)	2-II-C	Tiny lesions; roots discolored
Longidorus (Needle)	3-I-A	Short, stubby roots; small root-tip swellings
Meloidogyne (Root-knot)	3-I-A	General swellings and galls; roots distorted
Paratylenchus (Pin)	3-II-C	Roots shriveled; short and sparse
Trichodorus (Stubby-root)	3-III-A	Large brown lesions, mostly near tips; short, stubby roots with slightly swollen tips
Punctodera (Cyst)	4-II-A	Tiny, swollen, white females and brown cysts on roots; roots distorted and discolored
Belonolaimus (Sting)	4-III-A	Large, girdling lesions; short, stubby roots with knobby tips

*Arabic numeral indicates how commonly the genus occurs in Illinois: 1 (very common); 2 (common); 3 (uncommon); and 4 (not recorded but may occur rarely in some parts of the state). Roman numeral indicates soil preference: I (all soils), II (mainly heavy and amended soils); and III (mainly or only sandy soils). Capital letter indicates potential for damage: A (very damaging); B (moderately damaging); and C (damaging only at high populations).

root tips. A combination of nematode enzymes and mechanical damage is thought to reduce cell multiplication, resulting in a "stubby-root" appearance; swelling of the devitalized root tips is common. Other nematodes cause dark necrotic lesions (dead areas) in roots. Usually lesions are produced by nematodes that are actually entering, feeding, and multiplying within the roots. Some of the most obvious lesions are caused by lesion nematodes (*Pratylenchus*, see *Report on Plant Diseases* No. 1103). Often lesions are enlarged by fungi and bacteria that invade and multiply in the wounds. Regardless of the type of root symptom, all nematodes reduce the size of the root system, and the damage is reflected in symptoms that are expressed above ground.

Nematodes have been shown to increase the severity of other diseases in certain plants. Disease complexes often produce synergistic effects that devastate the hosts. Soil fungi, such as species of *Fusarium*, *Phialophora*, *Leptosphaeria*, *Rhizoctonia*, and *Pythium*, are common pathogens of turfgrasses and are likely to form disease complexes with nematodes in turf. Nematode-fungus interactions result from mechanical damage to roots and physiological changes that lower disease resistance in plants. The fungus may also affect the nematode. For example, species of *Fusarium* may either increase or decrease reproduction of certain nematodes.

GENERALIZED LIFE CYCLES

Nematodes develop from eggs through four juvenile stages into adults (Figure 4). In most species, the nematode hatches from the egg as a second-stage rather than as a first-stage juvenile. The juvenile worm moves in a wave-like motion through the water that surrounds soil particles, searching for a suitable host plant on which to feed. Molting takes place between each life stage as the nematode increases in size. Most nematodes may feed at any stage once they hatch from the egg. In some species, adults mate and females lay fertilized eggs. In others, males are either unknown or very rare and are not needed for egg production. Nematodes usually complete their life cycles in about one month if they are within their optimum soil temperature range of 20-30°C (68 to 86°F). However, some species may take up to a year to complete the cycle.

Nematodes feed ecto-, endo-, or semiendoparasitically (Figure 4). Ectoparasites, such as stunt nematodes (*Tylenchorhynchus*), remain outside of roots but push their stylets into roots in order to feed. However, most ectoparasites are migratory and may move from one place to another to feed. A few, such as ring nematodes (*Criconemoides*), are sedentary and rarely move once they begin feeding. Endoparasitic nematodes enter roots entirely in order to feed. Migratory endoparasites, such as lesion nematodes (*Pratylenchus*), feed and move freely within roots. Sedentary endoparasites, such as root-knot nematodes (*Meloidogyne*), never relocate after feeding sites are established. Semiendoparasites may feed externally or internally during parts of their life cycles or, more commonly, with their bodies partially embedded in roots. In any case, nematodes rarely travel more than one foot per year in the soil. One uncommon form, the seed-gall nematode (*Anguina agrostis*), does not attack roots but feeds internally in flower heads, principally of bentgrass, where it causes formation of tiny galls instead of seeds. For more information on root-knot, lesion, or ectoparasitic nematodes, see *Report on Plant Diseases* No. 1101, 1103, or 1106.

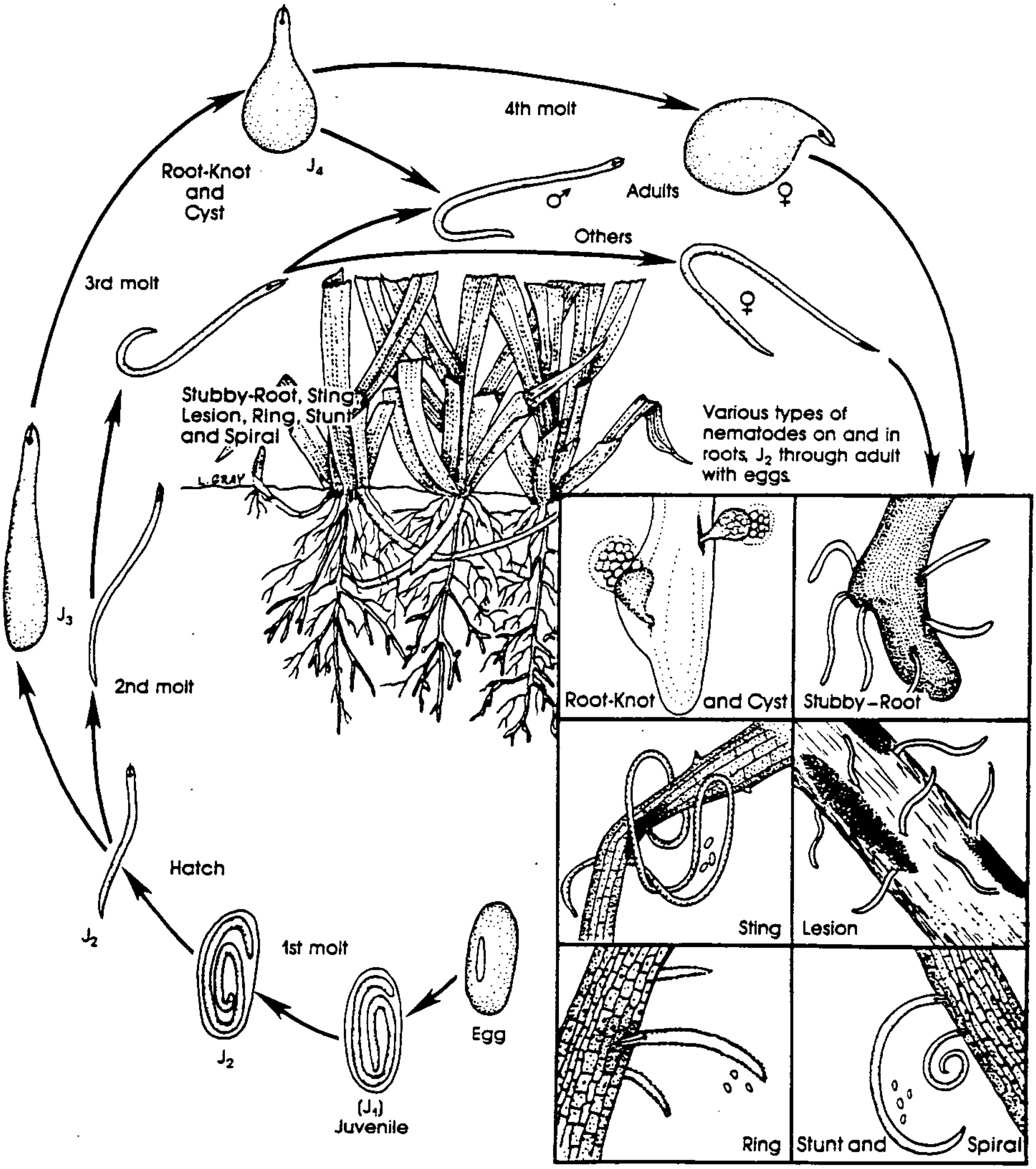

Figure 4. Generalized life cycles of some common turfgrass nematodes in the Midwest.

POPULATION DYNAMICS

Nematode populations fluctuate both seasonally and spatially. Changes vary according to the nematode species. Knowledge of the shifts in population sizes is necessary for properly diagnosing and managing nematode problems on turfgrasses. Populations of most species increase both during and immediately after periods that are favorable to turfgrass growth. Populations often peak in late spring and again in midfall. Although population levels generally increase when environmental conditions are favorable, above-ground symptoms may not appear until later when the turfgrass is under some stress.

Nematode spatial distribution depends upon the nematode involved, root growth habit of the grass species, soil characteristics, and relative health of the root system. Populations of most nematode species tend to cluster in small areas, which accounts for the frequent patchiness of the damage. Usually, nematodes are concentrated in the top 10 cm (4 inches) of turfgrass soil. Within a severely damaged area, the highest numbers of nematodes are found at the margins rather than in the center of the area because the healthier roots that have not been heavily fed upon provide a better food source. Nematodes move very slowly through the soil but are spread easily with infested soil, water, and plant material, primarily the result of humankind.

DAMAGE THRESHOLDS

A *damage threshold* is either the number of nematodes in a given volume of soil or the weight of roots that will cause detectable plant damage. Damage thresholds are extremely complex and difficult to establish: they vary with the form of nematode, type of soil, season of the year, species, growth stage and vigor of the plant, and cultural practices. Moreover, nematodes almost always exist in communities of numerous species. One, several, or all of the species present may be contributing to the damage. Population thresholds for damage to turfgrasses are not well defined and are based largely upon personal experience rather than research.

General thresholds for damage to turfgrasses differ widely among the genera and sometimes the species of nematodes (Table 1). For example, stubby-root (*Trichodorus*) and sting (*Belonolaimus*) nematodes can cause severe damage at relatively low population levels, whereas ring (*Criconemoides*) and pin (*Paratylenchus*) nematodes cause no detectable damage until populations increase to very high levels. Spiral (*Helicotylenchus*) and stunt (*Tylenchorhynchus*) nematodes, although damaging only at high levels, are the two most widespread genera, usually are the most abundant forms, and cause the most frequent problems on turfgrasses in the Midwest.

Other factors may increase or decrease the damage threshold for each nematode form. Usually, thresholds are lower in sandy soils than in heavy-textured soils that are more fertile and moisture-retentive. Additionally, turfgrasses are more tolerant of a given population level during the favorable growth periods in spring and fall than during summer when grass is exposed to numerous physical stresses. Also, shallow-rooted and nonrhizomatous grass species tend to be less tolerant than deeper-rooted and rhizomatous species. Seedlings are more sensitive than well-established plants. Furthermore, threshold levels are considerably higher on grass that is well fertilized and watered than on grass that is under nutrient or moisture stress. Close mowing places severe physiological stress on turfgrass, thus substantially lowering threshold levels.

DIAGNOSIS OF NEMATODE PROBLEMS

Soil Sampling

The principal concerns with turfgrasses are appearance, uniformity, and vigor. The collective symptoms of nematode injury usually produce a nonuniform appearance, exhibiting areas of both unthrifty and relatively healthy grass. Thus, a diagnosis of turf nematode problems requires paired sampling, which consists of both a sample from a suspect area and one from a "healthy" area. This procedure allows for the comparison between nematode populations of damaged and undamaged areas for both proper diagnosis and in order to provide data that helps to refine damage thresholds.

A nematode problem can be diagnosed only by a qualified nematologist in a laboratory equipped for nematode analyses. Although obtaining a "good sample" may be time-consuming, difficult, and appear costly, it is the most reliable method of diagnosis. The best time to sample is about one month after the grass resumes growth and greens up in the spring, or in midautumn. All samples should be taken from a depth equal to that of the root system, or about 10 cm (4 inches).

Sampling patterns depend on the size of the area to be sampled and the symptoms that have been observed. When only a gradual decline over an area is noticed, subsamples should be taken randomly in a zig-zag pattern throughout the area. A sample should consist of a minimum of twelve subsamples for an area that is up to an acre in size. A larger area should be subdivided into equal sectors, each being sampled separately. If more severe symptoms occur in patches, subsamples should be taken just inside the perimeter of the patches. Six or more subsamples must be obtained to submit at least one pint of soil. Additionally, a sample from an affected area should be accompanied by a sample from an adjacent, unaffected area. The best sampling tool for turf is a 2.5 cm (1-inch) diameter soil sampling tube. Subsamples also may be taken with a cup digger or narrow-bladed trowel: a vertical section of the soil from each subsample should be saved for the composite sample.

Subsamples should be mixed together and placed in a sturdy plastic bag. Samples must be either shipped immediately or stored in a refrigerator until they can be submitted for analysis. Furthermore, samples should always be kept out of direct sunlight and never be exposed to high temperatures. Also, they must be accompanied by a completed Nematode Sample Form, which is available from your county Extension office, or by a description of the problem and a thorough history of soil fertility, irrigation, and pesticide practices. Samples must be labeled separately and should be submitted to the Plant Clinic, St. Mary's Road, Urbana, Illinois 61801. (See *Report on Plant Diseases* No. 1100 for more information on sampling soil for nematode analysis.)

Nematicide Strip Tests

Infinite combinations of nematode species and environmental conditions exist which may affect the amount of damage to turfgrasses. Thus, a nematode analysis alone may not confirm that these parasites are the primary cause of poor growth. For an additional means of confirmation, growers should conduct a nematicide strip test, using a registered nematicide (see **CONTROL PRACTICES** and *Illinois Extension Circular* No. 1076).. The response of treated plants, when compared with that of untreated plants (Figure 1), frequently confirms that nematodes are the causal agents and thus justify the expense of more extensive nematicide applications. Strip tests also may be used for a preliminary diagnosis prior to soil sampling.

The strip test involves chemical treatment of a limited amount of turf and should be conducted within the problem area. A nonfumigant nematicide (most are also insecticides) should be applied in a minimum of three 1 by 2-meter (about 3 by 6-foot) strips accompanied by a similar number of untreated strips (Figure 1). Both treated and untreated strips must be alternated because of the wide natural variation in soil nematode populations across an area of turfgrass. To determine the efficacy of the nematicide application and to correlate plant response with nematode control, either the grower, the county Extension adviser, or a professional consultant should take a soil sample from each treated and untreated strip, 5 to 6 weeks after application, and submit the labeled samples to the Plant Clinic for a follow-up nematode analysis. The sampling procedure is the same as that described above.

CONTROL PRACTICES

There are three basic methods for controlling turfgrass diseases caused by nematodes.

1. *Reduce the initial nematode population.* This procedure is applied prior to planting. Tactics include ensuring that sprigs, stolons, plugs, soil, and equipment are free of parasitic nematodes: sprigs and stolons can be freed of nematodes by hot water treatment (49 to 52°C, or 120 to 125°F, for 45 minutes). Plugs should be from grass grown in heat-treated or fumigated soil. Soil mixes for golf greens and tees can be steamed or treated with methyl bromide or a similar fumigant biocide. Grading and planting equipment should be thoroughly washed or steamed before use.

   An extensive area of unthrifty turfgrass with a history of nematode or nematode-fungus problems can be treated with a fumigant nematicide prior to re-establishment of the turf. Fumigant nematicides include 1-3D (Telone II), metham-sodium (Vapam), and MITC (Vorlex). Soil fumigation is expensive and requires specialized equipment but usually is highly effective in overcoming these problems. Soil fumigation cannot be used where roots of ornamental trees and shrubs are present in the soil.

   Soil fumigation is best done in late summer or early autumn when soil temperatures are high enough for fumigant action and a new stand of grass can be established before winter or early spring. The area should be scalped of old turf, and the soil should be prepared for a seedbed condition to a depth of at least 15 cm (6 inches) prior to treatment. Soil temperatures at 10 cm (4 inches) must be 15 to 30°C (59 to 86°F) for at least a week after treatment. Fumigants are injected 10 to 15 cm (4 to 6 inches) beneath the soil surface, and the surface is partially sealed with a roller or drag bar. A post-fumigation waiting period of 3 to 4 weeks is required before seeding or sodding to allow the chemical to leave the soil without injuring new plants. The soil should be aerated several days before replanting if soil temperatures drop below 15°C (59°F) during the waiting period. *Fumigants are extremely hazardous chemicals, and all precautions for their safe use must be followed closely. Humans and animals must be kept off treated areas during the waiting period.* For more information on fumigation and other methods of soil disinfestation, see *Illinois Extension Circular* No. 1213.

2. *Reduce the nematode population increase.* Chemical control of nematodes on established turfgrasses can be accomplished with nonvolatile nematicides formulated as either granular materials or liquids (Figure 1). However, nematicides should be applied on a broad scale *only* when the results from a laboratory analysis have shown that nematodes are part of the observed problem or when treated areas in a strip test return to normal. Nonvolatile nematicides can be applied from midspring to midautumn, but the soil temperature at 10 cm (4 inches) must be at least 15°C (59°F) to ensure that a high percentage of the nematodes are active and are not in the resistant egg stage. Grass will respond most rapidly to May or June treatment, when nematodes are feeding most intensively and before damage and summer stress become severe.

   Ethoprop (Mocap), fensulfothion (Dasanit), and phenamiphos (Nemacur) are registered for use on established turf. These nematicides work best in light-textured, sandy soils and are less effective in soils that are high in silt, clay, and organic matter. They must be watered in with 1.2 to 2.5 cm (1/2 to 1 inch) of water *immediately* after application to avoid severe burning of the grass and to bring the chemicals into contact with the nematodes. *All nematicides are extremely toxic and must be applied only by trained personnel who are licensed to handle restricted use pesticides. Always read label directions and precautions in their entirety before opening the container.*

   The most effective and practical means of controlling nematode diseases is through the use of resistant turfgrass cultivars. However, little research has been done in this area, and no cultivars that are now available are known to be nematode resistant.

3. *Increase the turfgrass tolerance to nematode parasitism.* This procedure utilizes cultural practices to reduce nematode damage. A deep, healthy root system achieved through good turfgrass management can withstand a much higher population of nematodes than a shallow, poorly developed one. The most important steps are using clean equipment and sterile soil amendments, keeping the thatch layer under 1.2 cm (1/2 inch), aerating compacted soils, and proper and timely application of fertilizers and irrigation. While underfertilizing decreases tolerance, overfertilizing can result in an increase in leaf and crown diseases caused by fungi, as well as in nematode-fungus disease complexes. Although frequent, light watering, especially at midday, may be necessary to minimize wilting when nematode populations are high, brief irrigation must be interspersed with deep, heavy watering to prevent roots from becoming concentrated near the soil surface.

Malcolm C. Shurtleff

M.C. Shurtleff, Extension Plant Pathologist

Cooperative Extension Service

report on

PLANT DISEASES

DEPARTMENT OF PLANT PATHOLOGY
UNIVERSITY OF ILLINOIS AT URBANA-CHAMPAIGN

RPD No. 641
(Revised 8/89)

DECLINE AND DIEBACK OF TREES AND SHRUBS

M.C. Shurtleff

"Decline" is a general term describing the gradual reduction of growth and vigor in a plant. "Dieback" refers to the progressive death of twigs and branches which generally starts at the tips (Figure 1). Trees and shrubs affected by the decline and dieback syndrome may survive indefinitely, or they may die within a year or two after the symptoms are first noticed. Symptoms may continue to develop even when corrective practices such as proper watering, fertilization, and pruning are applied.

Fig. 1. Dieback of large elm as a result of a thick fill of clay soil.

Decline and dieback may be caused by many factors (Figures 2 and 3) and is usually progressive over several years. Trees and shrubs of all ages may be affected, although this disease complex is usually associated with plants that have attained some size and maturity.

SYMPTOMS

Symptoms of decline and dieback are often subtle, slow in developing, and usually uniform throughout the crown. A tree or shrub in the dieback stage, however, may have localized symptoms such as apparently healthy twigs and branches adjacent to dead or dying twigs and branches. Dieback usually begins in the top of a plant and progresses downward, but it may start on the lower branches, especially with conifers.

General symptoms of decline and dieback may include pale green or yellow leaves, delayed spring flush of growth, scorching of the leaf margins, small leaves, reduced twig and stem growth, early leaf drop, premature fall coloration, and, as the disease complex worsens, thinning of foliage in the crown, dieback of twigs and branches, and production of suckers on branches and trunk (Figure 1).

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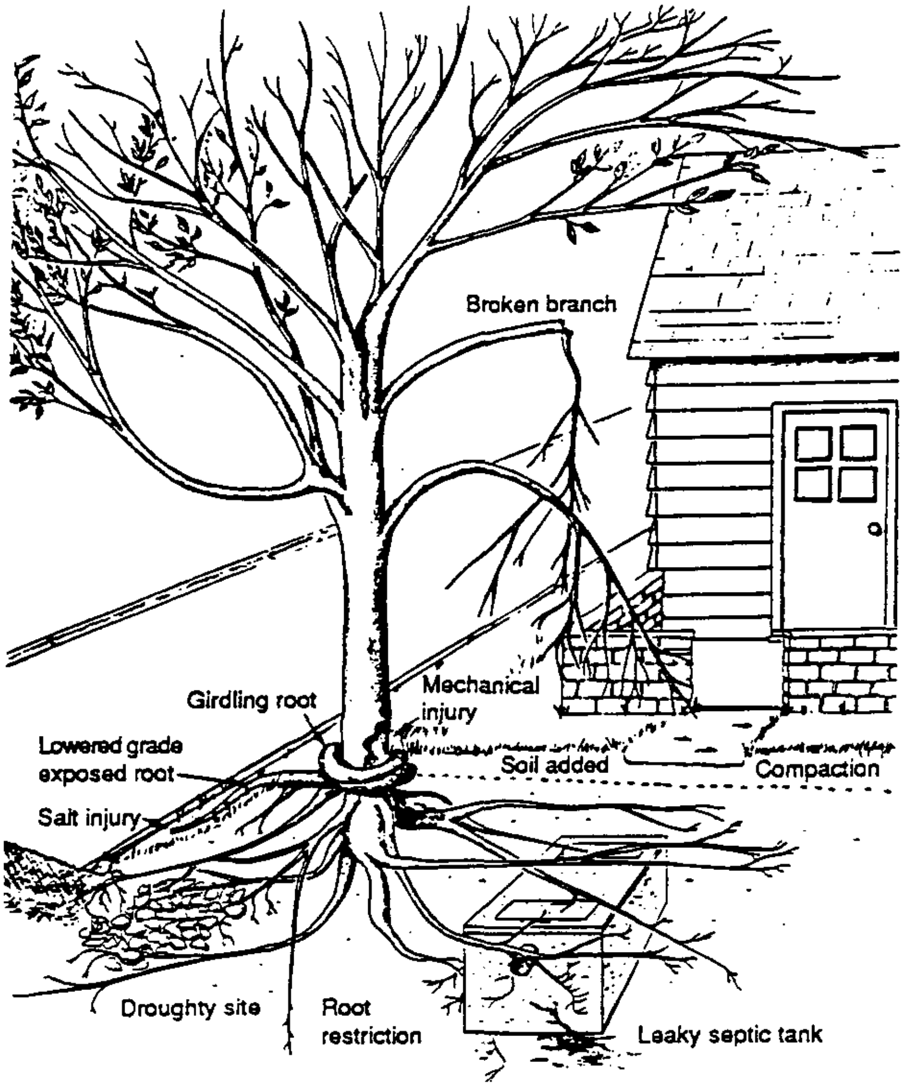

Fig. 2. Site factors that can lead to tree decline and dieback (Purdue University photo from a drawing by Rebecca J. Goetz).

Leaf scorch, a yellow to brown discoloration of the leaf margins and tip, is commonly a part of the decline and dieback syndrome, but scorch may result from many factors including a lack of adequate soil moisture, which results in less water reaching the leaf tips and margins. For more details on leaf scorch, read Report on Plant Diseases (RPD) No. 620.

Abnormally large seed crops are sometimes associated with decline. An occasional heavy seed crop, however, may be a normal response to certain weather conditions and may not be detrimental. In some tree species, heavy seed production occurs normally every few years; often the year before a plant dies.

Premature fall coloration, delayed spring flush, decrease in twig growth, and early leaf drop are typical symptoms of maple, oak, ash, honeylocust, birch and sweetgum decline and dieback, and the conditions usually become progressively worse each year with the leaves becoming smaller in size and fewer in number.

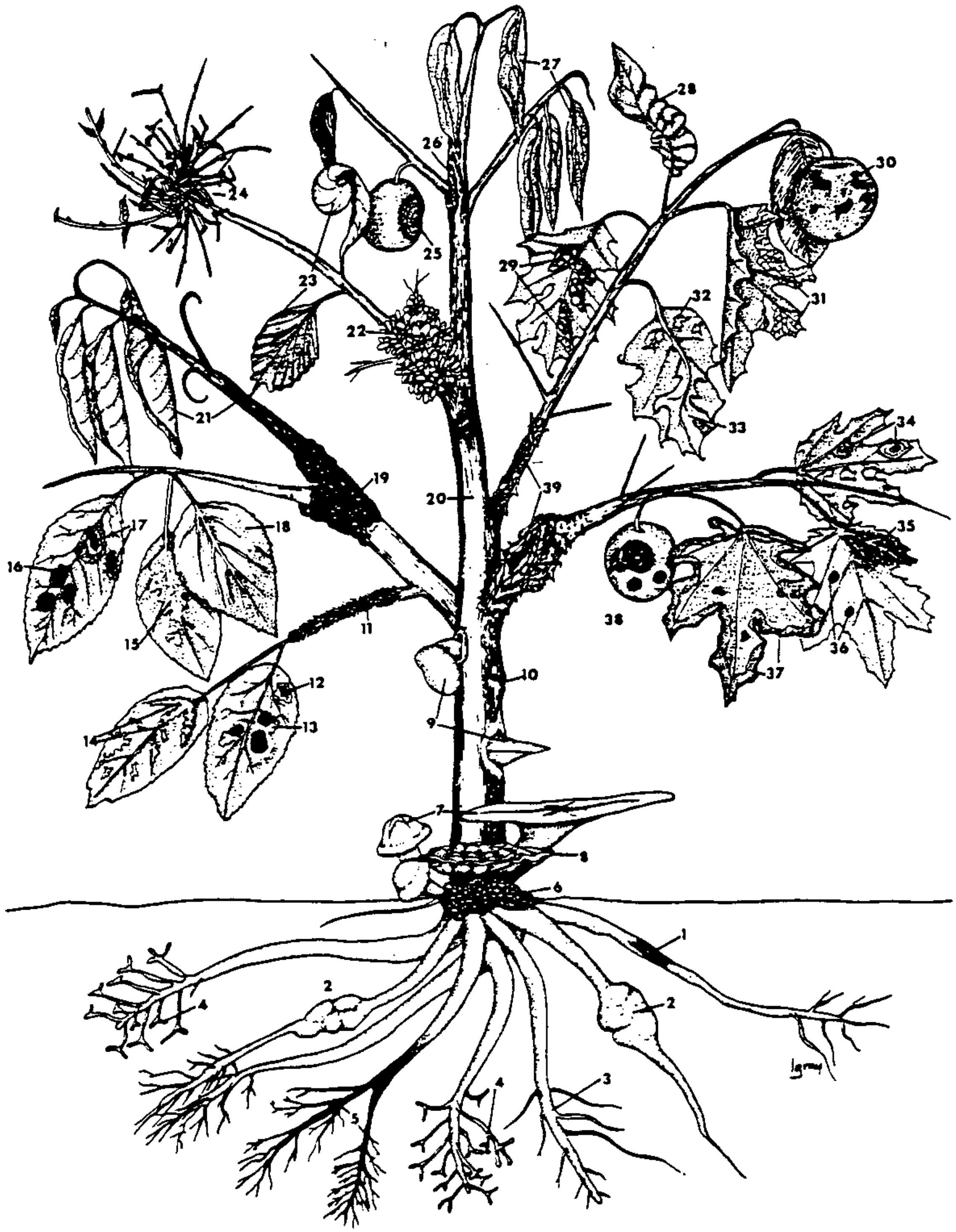

Fig. 3. Many of the diseases on this "sick" tree could result in decline and dieback. All 39 diseases would not occur on the same tree. The diseases are: *1*, Root-lesion nematode; *2*, root-knot nematode; *3*, root pruning by nematodes; *4*, stubby-root nematode injury; *5*, root rot; *6*, crown gall; *7*, fruiting bodies of Armillaria root rot fungus; *8*, fruiting body of Ganoderma wood and root rot fungus; *9*, fruiting bodies of Fomes wood rot fungi; *10*, trunk canker; *11*, cedar-quince rust on hawthorn; *12*, cedar-hawthorn rust; *13* cedar-apple rust; *14*, mosaic; *15*, downy mildew; *16*, apple scab; *17*, leaf spot; *18*, powdery mildew; *19*, black knot of plum and cherry; *20*, wetwood (slime flux); *21*, fire blight; *22*, American mistletoe; *23*, 2,4-D injury; *24*, witches' broom; *25*, fruit rot (apple); *26*, overwintering canker of fire blight; *27*, wilt; *28*, leaf curl or blister of peach, cherry or plum; *29*, leaf blister (oak); *30*, sooty blotch and flyspeck of apple; *31*, leaf blotch; *32*, shothole; *33*, anthracnose; *34*, ringspot; *35*, sooty mold; *36*, tar spot; *37*, leaf scorch; *38*, apple scab on fruit, *39*, twig and branch canker. Drawing by Lenore Gray.

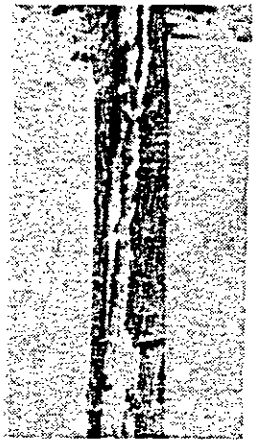

Fig. 4. Red maple killed by a rapid drop in temperature following a period of mild weather in mid-winter.

Fig. 5. A deep cut exposed and severed major roots on this oak and now the soil is eroding. The tree will probably soon show symptoms of decline and dieback or even die (USDA photo).

CAUSES

Trees and shrubs are long-lived and over a period of years are subject to attack by a variety of insects and diseases (Figure 3), extremely high or low temperatures--especially harmful is a rapid drop in temperature following a period of mild weather in the fall or spring--(Figure 4), great fluctuations in soil moisture during long-term weather cycles, mechanical damage to roots from construction (Figure 5) or livestock, and numerous other environmental effects (Figure 2).

Fig. 6. Root strangulation caused by improper planting.

Fig. 7. 2,4-D injury to redbud leaves.

These stress factors alone or in various combinations can reduce leaf and shoot growth (that is, initiate decline) and lead to dieback, although decline and dieback rarely result from a single stress factor. The combination of human impact on the local environment and natural climatic changes provides a multiplying effect that is more serious than any single factor. Usually a tree or shrub is first injured or damaged by disease (Figure 3), insect attack, or adverse soil or air environmental conditions (Figure 2). The damaged or weakened plant is then subject to attack by one or more secondary organisms or agents. For example, trees and shrubs weakened by drought or neglect are more susceptible to attack by borer-type insects and canker diseases than healthy, vigorous plants. Severe defoliation by leaf-eating insects, diseases, herbicides, hail, or wind at critical times of plant development also may initiate decline and dieback. If a defoliated plant develops new leaves late in the year, and if these leaves are, in turn, killed by an early frost, the plant will be low in food reserves and more subject to winter injury. Weakened trees also are subject to invasion by various fungi. Armillaria root rot (see RPD 602), for example, commonly attacks and kills the roots of weakened trees. Many other fungi attack the lower trunk and buttress roots of weakened trees.

Of the many stress factors that can initiate the beginning of decline and dieback, those that weaken or damage the root system are perhaps the most threatening. In most instances of decline and dieback the deterioration of the root system or the blockage of normal root functions occurs before any symptoms are visible in the crown. The root system is especially vulnerable to changes in the soil environment. Soil compaction, changes in the soil drainage pattern, excessive soil moisture (from rain or poor drainage) or lack of water (from prolonged drought), the removal or addition of soil over the root system (Figures 1 and 5), soil compaction, and chemical injury from excess deicing salt, pesticide (Figure 7), or fertilizer all can weaken the root system of trees and shrubs. An excess or deficiency of water, in particular, can lead to permanent root damage. Ash, birch, honeylocust, maple, oak, and sweetgum trees appear to be particularly sensitive to an excess or deficiency of water.

Trees and shrubs planted improperly or in unfavorable locations will also be stressed by poor root growth and development. Planting trees and shrubs too deeply or incorrectly (Figure 6) or in sites with poor drainage, mineral deficiencies or imbalances, a soil reaction (pH) that is too alkaline, poor soil type, or soil compaction should be avoided. Paved sidewalks, driveways, streets, building foundations, patios, septic tanks, and other obstructions can greatly restrict the growing space for proper root development. If a balance between the crown and root system cannot be maintained, the tree or shrub will be weakened, and decline and dieback may develop a few years after transplanting.

Because so many factors can cause decline and dieback, the primary causes are listed below in the approximate order of general frequency:

1. Poor soil structure and drainage (important when the soil is predominantly clay)

2. Herbicide injury to foliage, roots, or other parts (Figures 3 and 7)

3. Poor transplanting procedure and lack of proper maintenance after transplanting (Figure 6)

4. Construction damage--cutting and removal of roots (Figure 5)

5. Significant damage to trunk or major limbs (mechanical injury from lawn mowers, vandalism, vehicles, squirrels and other rodents, livestock, etc.) (Figure 2)

6. Repeated defoliation by insects or diseases, especially such leaf disorders as scorch (see RPD 620), anthracnose (RPD 621), rust (RPD 605 and 802), and leaf spot or needle blight (RPD 600, 610, 624, 637, and 800) (Figure 3)

7. An extended drought in combination with high temperatures and strong southerly winds

8. Vascular diseases--such as Verticillium wilt (see RPD 1010), oak wilt (RPD 618), or Dutch elm disease (RPD 647) (Figure 3)

9. Soil nutrient deficiencies

10 Insect borer injury to the trunk or branches.

11. Canker disease--(see RPD 604, 606, 626, 636, 801, 809, 812, and 814) (Figure 3)

12. Excessive soil moisture

13. Extremely low winter temperatures or a rapid change in temperatures (Figure 4)

14. Poorly formed or girdling roots (Figures 2 and 6)

15. Soil compaction from vehicles or heavy construction equipment

16. Fungal root and trunk decays such as Armillaria root rot (see RPD 602) (Figure 3)

17. Lightning injury

18. Soil fill or removal (Figures 1 and 5)

19. Bacterial wetwood and slime flux (RPD 656)

IDENTIFICATION

The exact cause or causes of decline and dieback needs to be identified so that corrective steps may be taken. Accurate diagnosis is often difficult however, especially on older trees. Usually an on-site examination of the diseased tree is required to assess the influence of the environment and to inspect for foliage, branch, trunk, and root problems. Laboratory examination of diseased leaf, twig, or branch specimens may confirm that an infectious disease problem exists (Figure 3). A careful examination of the roots, trunk, and soil conditions can reveal some basic causes for decline. In some cases, a precise diagnosis can be made only by a combination of field and laboratory examinations. It is very important to consider <u>both</u> the site and the past care given the plant. The following steps, as well as the answers to the questions posed, may help to determine the underlying cause or causes of the decline and dieback.

1. Determine the case history of the plant and general area: Has severe and repeated defoliation by insects, disease, or another cause occurred in recent years? Has severe drought or other adverse weather factors affected the plant in recent years? Has the soil been saturated or flooded for extended periods? has there been construction work near the tree in recent years causing trunk or root damage, soil compaction, or soil deterioration? Has there been soil or root removal? Has there been soil fill? (If unknown, observe whether the normal trunk flare is visible at the soil line. If not, determine the depth to the buttress roots.) Has the water table in the area changed? The use of a soil profile tube is essential in making many of the observations concerning soil problems.

2. Examine nearby vegetation: Is there evidence of injury to surrounding trees, flowers, shrubs, vegetables, fruits, turfgrass, or weeds that would suggest general environmental (Figure 2) or toxic symptoms? Is the tree or shrub's root system subject to salt accumulation from winter ice control along nearby sidewalks or streets? Is there a toxic sewage disposal field or gas line near the root system that may be leaking?

3. Consider chemical treatments to or near the tree or shrub: What is the history of pesticide use, particularly herbicides or "weed and feed" combinations? Was a soil sterilant or biocide used in a nearby gravel driveway?

4. Examine leaves for foliar diseases and insects.

5. Eliminate the possibility of a vascular disease, that is, oak wilt, Dutch elm disease, Verticillium wilt, or mimosa wilt, by considering the pattern of symptom development and by examining for internal sapwood discoloration.

6. Have a professional arborist determine the year(s) or period(s) of tree stress by examining the amount of twig growth and the width of growth rings in the wood. Also have the arborist check the pattern of annual stem elongation to determine if and when growth has slowed or stopped. (The arborist will examine the growth of annual rings over the last several years with an increment hammer or borer.)

7. Examine branches and trunk for extensive cankers that may be the cause of damage or that may be associated with an environmental or other stress.

8. Examine trunks and buttress roots for evidence of injury, for example, a sunscald, fire, mower, frost crack, or lightning injury. Look for loose bark (tap the bark and exposed roots and listen for a telltale hollow sound). Check for mushrooms or conks of wood and root decay fungi (see RPD 642). Fungal fruiting structures are most common in spring and fall following periods of wet weather.

9. Carefully excavate the buttress roots for evidence of fungal decay, poorly formed roots, girdling roots or twine, and similar problems.

CONTROL

Once the symptoms of decline or dieback are evident, it is difficult to stop or reverse the progress of disease. The key to control is <u>early detection and prevention</u>. The following measures will aid in maintaining the health of trees and shrubs.

1. Match the tree or shrub to the site. A common mistake is to select trees and shrubs that grow to a large size and to plant them where the roots are confined, such as between the sidewalk and street or close to a building. Many shade trees have very specific site requirements and grow poorly if these requirements are not met. Common mistakes are planting pin oak and sweetgum trees where the soil reaction is neutral or alkaline (pH 6.5 or above); planting white pines, peaches, cherries, plums, roses, yews (*Taxus*), and white oaks in poorly drained soil; and planting pines, spruces, Douglas-fir, or other evergreens that may reach a mature height of 100 feet or more and a crown width of 50 feet on both sides of a sidewalk on a narrow city lot. In these cases the trees eventually

will become crowded and decline because of competition for sunlight and soil moisture. Maintain wide spacings between trees and shrubs based on their size at maturity.

2. Maintain plant health. Plant properly in a deep, fertile, well-drained soil. Cut and remove all tying material and the container, and spread the roots into a natural position. Fertilize every year or two in early spring or late fall (after leaf drop) following a soil test report and the suggestions of either Extension horticulturists at the University of Illinois at Urbana-Champaign or the Illinois Natural History Survey Circular 52, Fertilizing and Watering Trees. Using a lawn sprinkler, water heavily with the equivalent of an inch or more of rainfall (soil moist 12 inches deep) at about 10- to 14-day intervals during drought periods. Watering is beneficial during relatively dry autumns to insure that the roots have adequate moisture during the winter dormant period. A two- to three-inch mulch of wood chips will benefit most trees and shrubs, as will the elimination of all grass competition to tree and shrub roots, although this may not be practical in many situations.

3. Avoid changes in the growing site. Any change in the growing site of a tree or shrub may cause decline. A delicate balance exists between a plant's root system and its environment. Any change in drainage, any damage to the roots from trenching or construction, or any other site change almost always results in root damage and decline. This process is usually irreversible, and prevention is the key to control. Soil fill will induce drought, reduce the exchange of gases to the roots, and lead to invasion by root decay fungi. Removal of four inches of topsoil can destroy 50 to 75 percent of the essential feeder roots and provide injuries that increase the chances of root or trunk decay and infection by Verticillium wilt or other diseases. If fill must be added, be aware that the tree may suffer and may die within 1 to 10 years depending on the depth and type of fill and the kind of tree if proper precautions are not taken. Trunk wells are worthless and should be avoided.

4. If soil compaction is a problem, apply two to three inches of wood chips and eliminate foot and vehicle traffic over the tree root area.

5. Avoid wounding the trunk and roots whenever possible. Avoid pruning that opens the crown excessively and increases trunk and scaffold limb exposure to summer or winter sunscald and frost crack. Wrap young, thin-barked trees to reduce these problems. Prune crossing branches and double leaders when the branches or leaders are still small. The resulting wounds will heal much more rapidly and completely than larger wounds from major branch removal. If branch, trunk, or root wounds occur, promptly and properly repair them. Protect the base of young trees from lawnmower damage by placing a ring of black plastic tubing several inches away from the base.

6. If decline or dieback symptoms appear, and no specific cause can be determined, try fertilizing and watering (see No. 2 above). Judicious pruning to remove all dead, dying, diseased, and crowded or rubbing branches may also reduce the stress on the root system and encourage renewed vigor. To avoid spreading disease-causing organisms, disinfect all tools by dipping or swabbing them with 70 percent rubbing (or wood) alcohol before using them on another plant.

7. If trees and shrubs have been defoliated recently by disease(s) or insects, take extra precautions to prevent repeated defoliation. Most established trees and

shrubs will tolerate one defoliation. Repeated defoliation during the same or succeeding years may result in fatal decline. Where practical, control foliar-eating insects following the suggestions of Extension entomologists at the University of Illinois at Urbana-Champaign. If disease is the problem, check suggestions by Extension plant pathologists at the same institution. Insect and disease control suggestions can also be obtained at your county Extension office.

8. Treat trees in decline. If trees and shrubs are in the early stages of decline or dieback, follow the suggestions outlined above for routine watering, fertilization, and pruning. You may also wish to call a competent, licensed arborist to check for such things as girdling roots, an unfavorable soil pH, and damage by borer-type insects and to treat the plant(s) where needed. Pruning may be desirable and necessary to remove dead, dying, and diseased wood, to reduce the crown size and put it into balance with a weakened or reduced root system, and to promote new growth. With proper care and management, plants can often be maintained, and the rate of decline reduced or further problems prevented.

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Cooperative Extension Service

PLANT DISEASES

DEPARTMENT OF PLANT PATHOLOGY
UNIVERSITY OF ILLINOIS AT URBANA-CHAMPAIGN

RPD No. 925
(Revised 6/89)

POWDERY MILDEW OF CUCURBITS

M.C. Shurtleff, R.E. Wagner, D.M. Eastburn

Powdery mildew, caused primarily by the fungus *Erysiphe cichoracearum*, may attack all vine crops or cucurbits. The vegetable crops most commonly affected are cucumber, gourds, muskmelon (cantaloupe), pumpkin, and squash. At times watermelon fruit, citron, vegetable marrow, West Indian gherkin, and other seldom-grown vegetables may become infected.

Other strains of the causal fungus infect about 300 other plants. Fortunately, the fungus strain that attacks cucurbits does not attack other crops.

The strain of *Erysiphe cichoracearum* that attacks cucurbits is further subdivided into physiological races. Plant breeders have successfully bred powdery mildew-resistant muskmelons only to have new races of the fungus develop that are capable of producing disease on these new varieties. The battle to "keep ahead" of this fungus is an ever-continuing one.

Powdery mildew in Illinois is most common and destructive near harvest in commercial pumpkin and squash fields and on greenhouse-grown cucumbers. The greatest loss occurs when day temperatures and humidity are relatively high, nights are cool, air circulation is poor, and moisture occurs as heavy dews instead of moderate to heavy rains.

Yield losses may approach 10 to 20 percent where control measures are not practiced.

Fig. 1. Powdery mildew on a cucumber leaf in the greenhouse (Courtesy R. C. Rowe).

SYMPTOMS

Powdery mildew appears first in midsummer on the leaves as small, circular, talcum-like spots that later expand and may merge. A superficial, powdery, white to dirty-gray fungus growth later covers part or all of the upper leaf surface, petioles, and

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young stems (Figure 1). The powdery to mealy growth may also appear on the underleaf surface and uncommonly on the fruit. Leaves 16 to 23 days after unfolding are more susceptible to infection than younger or older leaves. Severely infected leaves gradually turn yellow, then wither, die, and finally become dry and brittle. Vines are weakened and generally yellowish. Premature killing of the foliage results in fruit of poor quality and unfit for processing. Such fruit may be malformed, sunburned, may ripen prematurely, have poor flavor and texture, and low-soluble solid levels. Under favorable conditions the causal fungus may reproduce so rapidly that an entire field may appear white within a week to ten days.

DISEASE CYCLE

During warm, humid weather the powdery mildew fungus may produce over 2 million microscopic spores (conidia) on a square inch of leaf surface within a week or ten days (Figure 2). These spores, which are borne in chains, may be blown many miles in mild, moist air to infect vine crops. Thrips and other insects and farm equipment may disseminate the spores locally from leaf to leaf and from plant to plant.

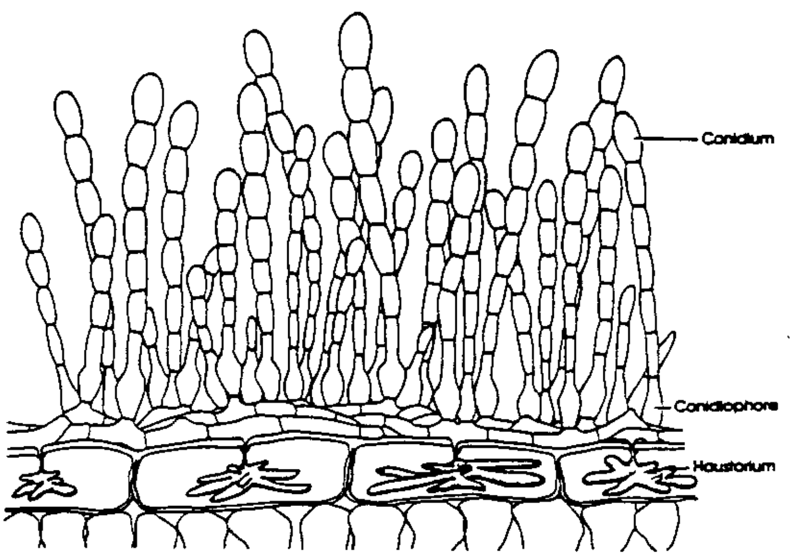

Fig. 2. *Erysiphe cichoracearum* as would be seen under a high-power microscope. The conidiophores give rise to chains of barrel-shaped conidia. The fungus gets its food by fingerlike haustoria in the epidermal cells (drawing by Lenore Gray).

The normal period between infection and the appearance of symptoms is from 3 to 7 days, or longer if weather conditions are unfavorable for the fungus. Infection and production of conidia occur most readily at about 81°F (27°C). The minimum and maximum temperatures are 50° and 90°F (10° and 32°C), respectively. The powdery mildew fungus is influenced greatly by plant age, humidity, and temperature. The fungus can reproduce under quite dry conditions with infections taking place with a relative humidity as low as 46 percent. The incidence of infection increases as the humidity rises to 90 percent or more, but does not occur when the leaf surface is wet.

In Illinois, the causal fungus is presumed to overwinter in crop and weed refuse. More important sources of infection may be spores produced on greenhouse vine crops or on cucurbits and perennial or other hosts grown in the field in frost-free areas of the far south during the winter. The spores are believed to be blown northward during the spring and early summer.

CONTROL

The principal control measures are the use of fungicides and the growing of resistant varieties.

1. Apply a recommended fungicide starting when powdery mildew is first seen, often on the shaded undersurface of crown leaves. Repeat the spray application in 7 to 14 days. Several sprays may be needed. Thorough coverage of the foliage is essential. Spray programs for vine crops are given in Report on Plant Diseases No. 900, Controlling Diseases in the Home Vegetable Garden, and University of Illinois Cooperative Extension Service Circular 1184, Disease Management Guide for Commercial Vegetable Growers (revised annually). Follow the manufacturer's directions regarding amounts to use, the interval between the last spray and harvest, and compatibility between fungicides and insecticides.

2. Where practical and possible, plant tolerant or resistant varieties of vine crops recommended for growing in Illinois. For more information refer to Illinois Circular 1174, Vegetable Varieties for Commercial Growers (revised annually), Illinois Circular 1150, Vegetable Gardening for Illinois, and the latest Illinois Vegetable Research Report. Also consult current seed catalogs and trade publications. A list of seed companies and distributors is included in Horticulture Facts VC-10-80, Sources of Vegetable Seeds.

3. Keep plantings free of weeds as long as is practical. Refer to Illinois Circular 907, Weed Management Guide for Commercial Vegetable Growers (revised annually), for current recommendations.

4. Seed treatments and crop rotation have no effect on powdery mildew infection.

For information on how and where to obtain the publications mentioned above, contact your local county Extension adviser.

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