RUTGERS
COOPERATIVE
EXTENSION

NEW JERSEY AGRICULTURAL EXPERIMENT STATION

PLANT DIAGNOSTIC LABORATORY

AND

NEMATODE DETECTION SERVICE

1992 ANNUAL REPORT

THE STATE UNIVERSITY OF NEW JERSEY
RUTGERS

PLANT DIAGNOSTIC LABORATORY AND NEMATODE DETECTION SERVICE
1992 ANNUAL REPORT

Dr. Karen Kackley-Dutt, Laboratory Coordinator
Mr. Richard Buckley, Diagnostician and Nematologist
Dr. Ann Brooks Gould, Faculty Coordinator

INTRODUCTION

The mission of the Rutgers Plant Diagnostic Laboratory and Nematode Detection Service (RPDL-NDS), a service of the New Jersey Agricultural Experiment Station (NJAES), is to provide the citizens of New Jersey with accurate and timely diagnoses of plant problems. These goals are achieved in cooperation with Rutgers Cooperative Extension (RCE) and research faculty at Cook College/NJAES. Since its establishment in April of 1991, the Plant Diagnostic Laboratory has examined over 1,100 samples submitted for plant problem diagnosis or nematode analysis. The laboratory has become an integral part of Rutgers Cooperative Extension and Cook College/NJAES programs by providing diagnostic and educational services and by assisting with research. This report summarizes the activities of the RPDL-NDS during the calendar year 1992, the laboratory's first full year of operation and the first half-year of operation for the nematode service.

HISTORY

The Rutgers Plant Diagnostic Laboratory was established in 1991 with an internal loan and is projected to be self-supporting within five years of establishment. The laboratory was established by the dedicated efforts of RCE faculty members Dr. Ann Brooks Gould and Dr. Bruce B. Clarke, Specialists in Plant Pathology, Dr. Zane Helsel, Director of Extension, formerly Chairperson of the Agricultural and Resource Management Specialists Department, and Dr. Karen Giroux, past Assistant Director of NJAES. Without their vision and persistence, this program would not exist.

On April 1, 1991 a Laboratory Coordinator was hired, on a consultant basis, to renovate laboratory space and order equipment. The laboratory is temporarily located in Building 6020, Old Dudley Road, on the Cook College Campus. This space belongs to the Department of Plant Pathology, who paid for renovations to the facility. We acknowledge the Department's generosity and thank them for their monetary support. The completion of the new Plant Science Building (Foran Hall), projected for 1994, necessitates the demolition of the current facility; therefore, the laboratory must be moved to a permanent location.

The Rutgers Plant Diagnostic Laboratory began accepting samples on June 26, 1991. At that time, the majority of equipment and supplies were in place. A full-time diagnostician (program associate) was hired September 1, 1991, and the Laboratory Coordinator was hired on a permanent basis on November 1, 1991.

The 1992 calendar year represents the first full year of operation of the Rutgers Plant Diagnostic Laboratory. On July 1, 1992, the laboratory assumed the responsibilities of the Nematode Detection Service, a service previously rendered by Dr. Jack Springer, Specialist in Plant Pathology, Rutgers Research and Development Center, Upper Deerfield, NJ. Subsequently, the laboratory has changed its name to the Rutgers Plant Diagnostic Laboratory and Nematode Detection Service (RPDL-NDS).

STAFF AND COOPERATORS

Karen Kackley-Dutt is the Coordinator of the RPDL-NDS. Dr. Kackley-Dutt came to Rutgers from Monsanto Agricultural Company where she worked in Product Development with plant protection compounds for turf and ornamentals. Dr. Kackley-Dutt received her Ph.D. in turfgrass pathology from the University of Maryland in 1989. Her M.S. and B.S. degrees are in ornamental horticulture, and she has over six years of experience in teaching horticulture at the University level. Dr. Kackley-Dutt worked as a diagnostician at the University of Maryland Plant Diagnostic Laboratory for five summers and as a Horticulture Consultant for the University of Maryland Cooperative Extension Service in Baltimore County for two years.

Richard J. Buckley is a Diagnostician and Nematologist at the RPDL-NDS. Mr. Buckley received his M.S. in turfgrass pathology from Rutgers University in 1991. He has a B.S. in Entomology and Plant Pathology from the University of Delaware. Mr. Buckley has work experience in diagnostics, soil testing, and field research. He has also received special training in nematode detection and identification and is responsible for the operation of the Nematode Detection Service. Together, Dr. Kackley-Dutt and Mr. Buckley are responsible for sample diagnoses and the day-to-day operation of the laboratory.

The laboratory benefits from the assistance of faculty in the Departments of Entomology, Plant Pathology, Crop Science, and Horticulture. Special thanks are extended to Dr. Louis Vasvary of the Entomology Department for all his help and encouragement. He is responsible for the majority of the insect diagnoses, and his assistance has been invaluable.

In the Plant Pathology Department, Dr. Ann Brooks Gould (laboratory Faculty Coordinator) and Dr. Bruce Clarke have devoted hundreds of hours to laboratory business from the inception of the diagnostic laboratory concept through its eventual set-up and operation. Additional faculty and staff in this department

who have provided substantial assistance during 1992 include: Dr. Donald Kobayashi, phytobacteriology; Dr. Steve Johnston, vegetable pathology; Dr. Jack Springer, fruit pathology and nematology; Dr. Brad Hillman, virology; Dr. T. A. Chen Plant Pathology, Chair, for administrative assistance; and Glenn Tappen, Mark Peacos, and Pradip Majumdar for general assistance.

We would like to thank Dr. John Meade of Crop Science for assistance in herbicide injury and weed identifications and Dr. George Wulster of Horticulture for assistance with horticultural crop problems. Our sincere gratitude goes to Ms. Ethel M. Dutky of the University of Maryland Plant Diagnostic Laboratory. Her advice and assistance has been instrumental in the set-up and operation of the RPDL-NDS. Additional thanks go to Dr. Steve Nameth of the Ohio State University Plant Diagnostic Laboratory and Ms. Margery Daughtrey of the Long Island Horticultural Research Laboratory.

LABORATORY POLICY

The RPDL-NDS receives samples from a varied clientele. According to clinic policy, samples for diagnosis from residential clients may be submitted only after they have been screened by appropriate county faculty or staff. If a sample requires more than a cursory diagnosis, it may be submitted, along with the appropriate payment, to the laboratory for evaluation. The county office provides the appropriate form, including instructions for proper sample selection and submission. Samples from professional clientele may be handled as above or may be submitted directly to the laboratory.

Detailed records are kept on all samples. A written response including the sample diagnosis, management and control recommendations, and other pertinent information is mailed or sent by FAX to the client. Additionally, the client is billed if payment does not accompany the sample. Copies are forwarded to appropriate county faculty and extension specialists for their records. Commercial growers are contacted by telephone or FAX to help them avoid delay in the treatment of pest problems.

OPERATIONS

Diagnostics

From January 1 through December 31, 1992, the RPDL-NDS examined 676 specimens submitted for diagnosis or identification (Table 1) and assayed 113 soil samples for nematodes (Table 2). As expected, the majority of samples were submitted during the summer months and diminished in the fall and winter. This represents a substantial increase in samples from 1991, when the laboratory was in operation for only six months and did not process nematode samples.

For comparison purposes, a listing of 1991 and 1992 sample submissions from the University of Maryland Plant Diagnostic Laboratory is included in Table 1. From an agricultural perspective, New Jersey and Maryland are quite similar. Both states have similar demographics (a mix of major urban centers with surrounding suburban and rural areas), geographies, and agricultural crops. The University of Maryland Plant Diagnostic Laboratory has been in operation since 1979 and should serve as a predictive model for future sample submission to the RPDL-NDS. The University of Maryland Plant Diagnostic Laboratory does not assay soils for nematodes because the University has a separate Nematology Laboratory; hence, the Rutgers Nematode Detection Service data are presented in a separate table (Table 2).

Table 1. RPDL-NDS plant sample submissions by month - Rutgers University vs. the University of Maryland, 1991 and 1992.

Month	Rutgers (1991)	Maryland (1991)	Rutgers (1992)	Maryland (1992)
January		19	11	19
February		33	8	32
March		56	23	63
April		75	52	71
May		140	78	109
June[1]	6	156	95	136
July	107	147	117	94
August	104	132	80	147
September	59	113	103	125
October	45	85	56	59
November	25	36	38	32
December	25	13	15	13
Total:	371	1005	676	900

[1]Note that there were only three working days in June, hence the small number of samples.

In both 1991 and 1992, sample submissions to the RPDL-NDS followed a pattern similar to that of the University of Maryland laboratory (Table 1); however, total submissions to Rutgers were less. This is due to several reasons including: 1) the Maryland laboratory is established and well-known to the growers of the State, while the Rutgers laboratory is relatively new; and 2) the Maryland laboratory does not charge for samples submitted through a county agent, whereas the Rutgers laboratory charges to process samples. We expect Rutgers sample numbers will increase significantly as we continue to advertise laboratory services and as more growers become aware of our services. It should be noted however, that the University of Maryland laboratory experienced a decline in samples from 1991 to 1992. This is a trend that the University of Maryland laboratory has noted over a period of four years. The Laboratory Coordinator at Maryland attributes the decline in sample numbers to a reduction in Cooperative Extension field faculty.

During its first six months of operation, the Nematode Detection Service at the Rutgers laboratory processed 113 soil samples for nematode assays. Prior to July 1, 1992, this service was rendered by Dr. Jack Springer at the Upper Deerfield Station. After July 1, 1992, Dr. Springer continued to process samples submitted by county extension faculty free of charge, but will not continue this practice in 1993. In addition, he continued to process samples received from commercial growers but informed them that all future samples must be submitted to the RPDL-NDS. It is expected that the number of samples submitted to the Nematode Detection Service will increase dramatically in 1993.

Table 2. RPDL-NDS nematode sample submissions by month - 1992.

Month	Samples
July	26
August	2
September	40
October	42
November	3
December	0
Total:	113

Of the specimens submitted to the RPDL-NDS for diagnosis or identification, 63% were from commercial growers, 30% were from residential clientele, and 7%

were submitted from research faculty at Rutgers University (Table 3). Of the samples submitted to the Nematode Detection Service, 74% were from commercial growers, 24% were from research faculty at Rutgers University, and 2% were received from residential clientele. We expect that the number of nematode samples submitted from residential clients will remain low since this clientele is not familiar with these pests. While samples from research programs represent a relatively small percentage of the total number of plant and soil samples received, they are an extremely important component. Research samples allow the diagnosticians to cooperate with University faculty on problems often of great importance to the State of New Jersey. The problems associated with these samples are challenging and occasionally lead to the diagnosis of a new disease.

Table 3. RPDL-NDS sample submissions by origin - 1992.

Sample Origin	Number of Plant Samples	Percent of Total	Number of Nematode Samples	Percent of Total
Commercial Growers	428	63%	84	74%
Residential	202	30%	2	2%
Research Programs (Rutgers University)	46	7%	27	24%
Total:	676	100%	113	100%

A sample submission form and the appropriate payment accompanied the majority of samples received from residential clientele. Most commercial samples were accompanied by a submission form; however, the majority of these submissions did not include payment. In most cases, commercial growers preferred to be sent a bill. A number of samples (see Table 7) were examined free of charge. Laboratory policy allows Rutgers employees and government agencies to submit a small number of samples at no cost for educational development and government service. Some of the research samples were paid for by transfer of funds.

The vast majority of samples submitted for diagnosis (87%) were either turfgrass or ornamental plants (Table 4). This may be due to the fact that turfgrass and ornamentals represent the largest agricultural commodities in New Jersey. The wide variety of turf and ornamental species grown under diverse conditions results in a large number of problems not readily identifiable by growers or county faculty. In addition, pest diagnosis and identification for commercial growers of other crops are still handled by Extension Specialists in other parts of the State at no charge. It is hoped that, in the future, more of the commercial growers will submit samples to the RPDL-NDS.

Table 4. RPDL-NDS sample submissions by crop category - 1992.

Crop	Number of Plant Samples	Percent of Total	Number of Nematode Samples	Percent of Total
Turf	232	34%	49	43%
Ornamentals	360	53%	2	2%
Other Crops	42	7%	62	55%
Identification	42	6%		
Total:	676	100%	113	100%

Samples were submitted to the RPDL-NDS from all of the counties in New Jersey (Table 5). The majority of samples were submitted from the counties in closest proximity to the laboratory. Many citizens in central New Jersey contact Rutgers University directly for help with their plant-related problems and are referred to the laboratory. This distribution may also be influenced by the agricultural nature of the individual counties. Most of the counties with a high number of submissions are densely populated. The major commodities in these counties are frequently turf and ornamentals in residential landscapes. As mentioned above, problems on these crops are difficult to diagnose and are subsequently submitted to the laboratory. This county profile also identifies the county faculty who are familiar with the RPDL-NDS and utilize its services.

Approximately 8% of the samples submitted for diagnosis to the laboratory were from out-of-state (Table 5). Nearly all of these samples were turf. Because of his national reputation, many golf course superintendents around the country submit samples to Dr. Bruce Clarke, who has often forwarded these samples to the Diagnostic Laboratory. Because there are very few laboratories in the country that diagnose turfgrass diseases, these superintendents have continued to submit samples to the RPDL-NDS. The charge for out-of-state samples is substantially higher to help defray the cost of in-state samples.

Of the plant specimens submitted to the RPDL-NDS for diagnosis or identification, 47% were associated with biotic disease-causing agents (Table 6). Injury to 10% of the samples was caused by insects and related arthropods, and 37% were associated with abiotic injuries and stresses (e.g., nutrient deficiencies, poor cultural practices, poor soil conditions, etc.). Another 6% included plant, insect, and substance identification. This breakdown of samples is typical of those received by other diagnostic laboratories in the United States.

Table 5. RPDL-NDS sample submissions by county - 1992.

In-State	Number of Samples 1991	Number of Plant Samples 1992	Number of Nematode Samples 1992
Atlantic	9	20	0
Bergen	34	70	0
Burlington	16	38	0
Camden	8	14	0
Cape May	7	8	5
Cumberland	0	9	0
Essex	3	14	22
Gloucester	7	38	27
Hudson	0	9	0
Hunterdon	11	14	1
Mercer	26	32	1
Middlesex	50	75	0
Monmouth	24	65	1
Morris	16	24	0
Ocean	18	41	1
Passaic	3	21	1
Salem	1	2	0
Somerset	27	37	0
Sussex	7	15	1
Union	11	16	0
Warren	14	14	0
Research Samples (Rutgers University)	10	46	27
New Jersey Total:	302	622	87
Out-of-State	69	54	26
Total:	371	676	113

Table 6. RPDL-NDS plant sample submissions by diagnosis - 1992.		
Diagnosis	**Number of Samples**	**Percent of Total**
Disease (biotic)	319	47%
Insect	66	10%
Identification	42	6%
Other	249	37%
Total:	676	100%

Educational Opportunities

Extension lectures and presentations. In addition to providing diagnostic services, the staff of the RPDL-NDS provides educational services to Cook College/NJAES, Rutgers Cooperative Extension, and other agencies. Many of these educational activities generated additional income for the laboratory.

In 1992, the staff of the Plant Diagnostic Laboratory participated in a number of short courses offered by the Office of Continuing Professional Education. During the spring session, Mr. Buckley assisted Dr. Phil Halisky in the teaching of the Turf Diseases section of the Rutgers Professional Golf Turf Management School. He assumed full responsibility for this section of the school, which constitutes one two-hour lecture per week for ten weeks, during the fall session. Mr. Buckley will continue to teach this section of the course in the coming years. Dr. Kackley-Dutt presented two lectures in the Professional Turfgrass and Landscape Management Short Course and one lecture in the Greenhouse Crop Production Short Course. The income generated by these speaking engagements was $1,600.

Dr. Kackley-Dutt participated in three other educational activities in 1992 that generated further income for the laboratory. She presented lectures at the 1992 Advanced Landscape Management IPM Short Course at the University of Maryland; the Delaware and Maryland Ornamental and Turf Workshop at the University of Delaware; and the Plant Pathology and Mycology special course presented for employees of American Cyanamid. The income from these teaching activities totaled $3,300. Other educational services provided by the staff of the RPDL-NDS, for which the laboratory received no compensation, included lectures presented at the New Jersey Turf Expo 92, the 25th Annual Regional Grounds Maintenance Conference in Ocean City, and selected lectures in graduate level plant pathology courses.

On several occasions during 1992, the staff of the RPDL-NDS generated extra income for the laboratory by contracting labor to help with various research projects within Cook College. This contract labor brought in an additional $340 to the laboratory.

Extension publications. Dr. Kackley-Dutt co-authored four Extension Fact Sheets and four Extension Bulletins. The four fact sheets were all produced in association with Dr. Ann Brooks Gould and are titled: Effects of Ozone, Fluoride, and Sulfur Dioxide Pollution on Landscape Plants; Root and Crown Rots of Herbaceous Ornamentals in the Landscape - Diseases Caused by the Fungus *Rhizoctonia*; Root and Crown Rots of Herbaceous Ornamentals in the Landscape - Diseases Caused by the Fungus *Pythium*; and The Impact of De-icing Salt on Roadside Vegetation. Two of the Extension Bulletins were produced in association with Dr. Gould and are titled: Common Spring-Time Diseases of Woody Ornamentals in the Nursery; and Common Spring-Time Diseases of Woody Ornamentals in the Landscape. Two other bulletins, entitled: An Integrated Approach to Summer Patch Control in Turfgrass; and An Integrated Approach to Necrotic Ring Spot Control in Turf, were produced in association with Dr. Bruce B. Clarke.

During 1992, the RPDL-NDS contributed regularly to the Insect-Disease-Weed Newsletter. The laboratory staff wrote a brief article for each issue of the newsletter that is published weekly from March to September (24 issues) by Dr. Louis Vasvary of the Department of Entomology.

The laboratory staff serves on the Rutgers Cooperative Extension Home Horticulture Working Group and volunteered time at the New Jersey Flower and Garden Show at the Garden State Convention and Exhibit Center in February 1992.

Training and Advertising

In 1992, the staff of the laboratory developed a Plant Diagnostic Laboratory Policy and Procedures Manual. The purpose of this manual is to inform the staff of the county offices, and selected offices on campus, about the Plant Diagnostic Laboratory and how to properly access its services. The manuals are set up in a three-ring binder format with nine sections addressing such topics as how to collect and send a sample, how to contact the laboratory, etc. These manuals were distributed to each county during two half-day training sessions held in May for secretaries, program associates, and master gardeners from county offices. During these training sessions, the Policy and Procedures Manuals were distributed and explained in detail, a slide presentation on the Plant Diagnostic Laboratory was shown, and a tour of the laboratory was provided. In addition to the Policy and Procedures Manual, each county office received a supply of laboratory advertising brochures for distribution to the public. The training sessions were well-attended and enthusiastically received.

The RPDL-NDS developed a 15 minute slide presentation to help advertise laboratory services to various grower groups. Copies of this presentation are available on loan to anyone who wishes to advertise the laboratory's services. Numerous presentations of this program were made throughout 1992 by the staff of the Plant Diagnostic Laboratory, Extension Specialists, and County Faculty.

An advertising brochure was developed for general distribution at county offices, grower meetings, and other activities. This brochure briefly describes the services of the RPDL-NDS and how to access its services, and over 2,700 copies of this brochure have been distributed. A copy of this brochure is included with this report.

In the summer of 1992, the RPDL-NDS was visited by the editor of Northern Turf Management Magazine. She interviewed the laboratory staff and wrote a full-page feature article, with color photographs, about the laboratory that appeared in the August 1992 issue of the magazine. A copy of this article is included with this report.

Professional Improvement and Service

Mr. Buckley attended the Nematode Identification Course for Professional Consultants held at Clemson University, 12/28/92 to 1/7/93. This training greatly refined Mr. Buckley's skills in nematode detection and identification. This will allow more efficient processing of nematode samples, resulting in an increased sample capacity in 1993. Funding for this training was provided by Dr. Steve Johnston of the Rutgers Research and Development Center in Upper Deerfield, NJ. We wish to acknowledge his generosity and support.

The staff of the RPDL-NDS actively participated in several meetings of the American Phytopathological Society (APS). At the APS Potomac Division meetings in March 1992, the laboratory staff presented a research poster on a new disease in New Jersey, and another general information poster about the Rutgers Plant Diagnostic Laboratory. Mr. Buckley presented a research paper at the APS Northeast Division meetings in October. Dr. Kackley-Dutt served as the vice-chair of the Extension Committee for the APS Northeast Division in 1992, and will serve as Chair in 1993.

FUNDING

The Plant Diagnostic Laboratory is expected to be self-supporting within five years of its establishment. Funding for the laboratory is generated by charging clientele for diagnostic services and educational activities.

The 1992 fee schedule for diagnostic services and nematode assays was:

Residential Clients	\$20.00/sample
Commercial Growers	
Fine turf	\$50.00/sample
All others	\$20.00/sample
Out-of-State Growers	\$75.00/sample

This fee schedule represents a substantial fee increase from 1991 when the fee schedule was as follows:

Residential Clients	\$10.00/sample
Commercial Growers	
Extensive procedures not required	\$10.00/sample
Extensive procedures required	\$20.00/sample
Out-of-State Growers	\$30.00/sample

There was no reduction in sample numbers with the increase in diagnostic fees, and the increased fees greatly increased revenues. Over \$21,000 was generated from diagnostic services and nematode assays during the 12 months of 1992, whereas approximately \$5,700 was generated over six months in 1991. Compared to 1991, sample numbers doubled, and income increased nearly 370%.

Whereas the majority of samples received from residential clients were accompanied by payment, most commercial grower samples were not. In most cases, commercial growers preferred to be billed. Over 99% of the clients billed have remitted payment.

County faculty, Extension Specialists, and selected government agencies are allowed to submit a small number of samples "free of charge." These samples are to be used for educational development and government service. The Diagnostic Laboratory processed 129 of these "no charge" samples in 1992 (Table 7). These samples accounted for 19% of the samples processed. The value of these no charge requests was \$2,670.

Table 7. Plant Diagnostic Laboratory sample submissions - no charge requests.

Client Category	Number of Samples
RCE County Faculty/Program Associates	29
RCE Specialists	26
Rutgers Research Programs (not RCE)	19
Rutgers Non-Research Faculty/Staff	12
Direct Mail/Walk-ins	31
Other Government Agencies	6
Payment Returned-Sample Inadequate for Diagnosis	4
Resubmissions for Further Diagnosis	2
Total:	129

Income generated from all laboratory activities covered 100% of the non-salary expenses incurred in 1992, plus 24% of salaries, or 30% of the laboratory's total expenditures (including salaries and one-time costs for equipment). Salaries and benefits for the two full-time employees accounted for nearly 92% of laboratory expenses. For more detailed budget information see Appendix I.

FUTURE DIRECTIONS

As in the past, the top priority for 1993 will be to generate more income. To accomplish this, we will continue to advertise laboratory services to increase sample number. Continued cooperation with the Office of Continuing Professional Education and other educational activities are expected to generate additional funds.

Other priorities in 1993 include: developing additional educational materials in the form of bulletins and fact sheets in cooperation with extension faculty; focusing on ways to add and train labor for the laboratory during its busiest periods; finding and moving into suitable permanent facilities as soon as possible; and professional improvement (which includes participation in professional societies).

We are constantly evaluating the immediate and future needs of the State for additional services. Possibilities for additional services include assays for determining pest tolerance (Apple Scab, Brown Rot and European Red Mite) for the Fruit IPM program, and expanded insect identification services. In order to offer additional services, however, it will be necessary to increase staffing. It is hoped that the additional services will decrease the input costs per sample.

PLANT DISEASE HIGHLIGHTS

The occurrence and severity of plant diseases are strongly influenced by environmental conditions. The 1992 growing season was unusually cool and wet. Foliar diseases are strongly favored by these conditions. In addition, root injury is a common result of extremes in soil moisture. Symptoms of root injury include wilting, leaf scorch, branch dieback, premature fall color, premature defoliation, and decline. Root dysfunction also weakens plants by predisposing them to infection by a number of opportunistic organisms. Many of the plant diseases diagnosed in the laboratory in 1992 were favored by adverse environmental conditions.

Ornamentals

The majority of ornamental plants submitted to the laboratory were affected by abiotic agents. Due to poor planting depth, soil drainage, and site conditions, many Christmas tree specimens submitted were in a state of decline. Of the diseases that were caused by biotic agents, leaf spots, shade tree anthracnose, and ash rust were particularly prevalent. Root-infecting pathogens frequently detected this year on a variety of ornamental plants included *Phytophthora*, *Pythium*, *Fusarium*, and *Rhizoctonia*. The two insect problems most commonly diagnosed were spruce mites and scale.

Greenhouse diseases of note included Bacterial Blight of Geranium, Downy Mildew on snapdragon, Tomato Spotted Wilt on New Guinea impatiens, and Botrytis Blight on a wide variety of plants.

During the spring of 1992, two new and unusual disease problems caused by bacteria were detected in New Jersey nurseries. *Pseudomonas syringae* was found to cause excessive bleeding and dieback in recently pruned *Magnolia virginiana*. *Pseudomonas syringae* and *Xanthomonas campestris* were associated with a dieback and foliar blight of *Euonymus fortunei*. These diseases were investigated in cooperation with Dr. Donald Kobayashi of the Plant Pathology Department. Reports of these new diseases will be published in the scientific literature.

Turf

The unusually cool, wet weather was very conducive for cool-season diseases of turf. Pink Snow Mold was detected throughout the spring and early summer, and reappeared in September. Other commonly occurring cool-season diseases included Root Pythium, cool temperature Brown Patch, Necrotic Ring Spot, and Leaf Spot and Melting-out. Anthracnose was common on *Poa annua* that had been stressed by poor root development and excessive soil moisture. Annual bluegrass weevil was detected in a number of samples. Of particular note was the dramatic decrease in many of the hot weather diseases, such as Summer Patch.

Vegetables

Diseases of note in 1992 included Maize Dwarf Mosaic Virus in commercial sweet corn, Tomato Pith Necrosis in commercial tomatoes, and Cercospora Leaf Spot in Swiss chard.

APPENDIX I. RPDL-NDS BUDGET

Table 8. RPDL-NDS expenditures in 1992.	
Salaries & Benefits:[1]	$86,402.22
Supplies and Services:[2] (includes) Diagnostic supplies Printing/advertising References/publications Equipment maintenance Office supplies Photographic services	$ 4,837.41
Communications:[3] Telephone/FAX Postage	 $346.13 $668.52
Travel:[3] (includes) Travel to give paid talks Travel to professional meetings Travel for training	$1914.79
Total Expenditures:	$94,169.07

[1]From Account #89676.
[2]From Accounts #89676 and #89232.
[3]From Account #89232.

Table 9. RPDL-NDS income in 1992.	
Sample fees:	$20,460.00
Unpaid sample fees:	$625.00
Contract labor:	$340.00
Lecture fees:	$4,916.60
Faculty gifts for education of RPDL-NDS staff:	$1,798.58
Value of no-charge samples	<$2,670.00>
	$30,810.18
Actual Total Income:	$28,140.18

Table 10. RPDL-NDS estimated expenditures for 1993.	
Salaries and benefits[1]:	$80,000
Seasonal labor:	$3,600
General operating:	$7,500
Equipment required for Nematode Detection Service[2]: Refrigerator, sieves for elutriator, centrifuge and supplies, microscope accessories, pH meter and solubridge...............................$7,300	$0
Educational development and travel:	$2,000
New facility renovation?	?
Total Estimated Expenditures:	$93,100

1 Salary includes projected six weeks disability for Dr. Kackley-Dutt. Depending on circumstances, this estimate may be further reduced.

2 Equipment to be donated by plant pathology faculty.

Table 11. RPDL-NDS estimated income for 1993.		
Estimated number of plant samples:		
Turf (34%):	262 samples @ $50 each:	$13,100
	78 samples @ $75 each:	$5,850
All others (66%):	660 samples @ $20 each:	$13,200
	Estimated Plant Sample Income:	$32,150
Estimated number of nematode samples:		
Turf (43%):	98 samples @ $50 each:	$4,900
	48 samples @ $75 each:	$3,600
All others (57%):	194 samples @ $20 each:	$3,800
	Estimated Nematode Sample Income:	$12,380
	Estimated Total Sample Income:	$44,530
Professional services: Short courses, Extension presentations, etc.		$7,000
	Total Estimated Income:	$51,530

Table 12. Plant Diagnostic Laboratory Charges in Neighboring States.	
Connecticut (Ag Expt. Sta.): All salaries and operating expenses are covered. Types of samples handled include diseases, insects, nematodes and soils.	No charge for any sample.
Maryland (UMD): All salaries and operating expenses are covered by Cooperative Extension. Discussing implementing a charge of \$15-\$20 per sample.	No charge if submitted through county agent.
Massachusetts (UMass): There is no Plant Diagnostic Laboratory. All samples are handled by Specialists who charge growers.	\$25.00 No charge to county agents.
New York (Cornell): All salaries and operating expenses are covered by Cooperative Extension. General diagnosis: Nematode or virus assay: These fees are charged by both the Diagnostic Lab and by Specialists. There are no free samples; even county agents pay for services. Some county offices charge to look at samples (usually only \$2-\$3).	 \$25.00 \$40.00
Pennsylvania (Penn State): All salaries and operating expenses are covered by Cooperative Extension. Discussing implementing a charge for samples not submitted through county agent.	No charge if submitted through county agent.
Vermont (U of VT): All salaries and operating expenses are covered by Cooperative Extension.	\$15.00

RUTGERS COOPERATIVE EXTENSION

NEW JERSEY AGRICULTURAL EXPERIMENT STATION

Plant Disease Control

ROOT AND CROWN ROTS OF HERBACEOUS ORNAMENTALS IN THE LANDSCAPE

DISEASES CAUSED BY THE FUNGUS *RHIZOCTONIA*

Karen Kackley-Dutt, Ph.D.
Coordinator, Plant Diagnostic Laboratory

Ann Brooks Gould, Ph.D.
Extension Specialist in Plant Pathology

The fungus *Rhizoctonia* is a common pest capable of infecting a wide range of herbaceous ornamentals in the landscape. Plants infected with this fungus exhibit a variety of symptoms that include root rots, crown rots, and damping-off of seedlings. *Rhizoctonia* usually lives in the soil and is favored by warm, moist conditions. *Rhizoctonia* is found throughout the world and has been reported to cause disease in over 500 genera of plants. Some of the more common herbaceous ornamentals grown in New Jersey that may be attacked by this fungus are listed in Table I.

SYMPTOMS AND SIGNS

Plants infected with *Rhizoctonia* may grow poorly and wilt, even when sufficient water is present in the soil. A close examination of the plant may reveal the presence of brown to red-brown areas of dead tissue (lesions) on root and lower stem (crown) tissue near the soil surface. When conditions are favorable for disease development, these lesions quickly enlarge to form sunken cankers and large areas of rotted tissue. Severely infected plants die after root systems are killed or when the stems become girdled. Under conditions of high humidity, fluffy fungal threads (mycelium) may be seen on affected tissue.

Table I. Herbaceous ornamental plants commonly affected by *Rhizoctonia*.

ageratum	iris
ajuga	lily
alyssum	liriope
aster	lobelia
baby's breath	loosestrife
begonia	lupine
calla	marigold
candytuft	mint
carnation	morning glory
chrysanthemum	nasturtium
cockscomb	pansy
coleus	peony
columbine	petunia
cornflower	phlox
cosmos	poppy
daffodil	portulaca
dahlia	pot-marigold
daylily	primrose
delphinium	sweetpea
English ivy	salvia
foxglove	snapdragon
gerbera daisy	sunflower
geranium	tulip
gladiolus	vinca
hosta	violet
impatiens	zinnia

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DISEASE DEVELOPMENT

Rhizoctonia survives in the soil as resting structures (sclerotia) or as mycelium associated with organic matter or plant debris. When plants are placed into an infested soil, the fungus is stimulated by substances released from the plant roots. Conditions that favor the growth of *Rhizoctonia* are warm soil temperatures (70 to 90°F) and moderate soil moisture (65% soil saturation).

DIAGNOSIS

Rhizoctonia is one of a group of fungi that can cause similar root and crown rot symptoms on herbaceous ornamentals. Frequently, more than one of these fungi are associated with diseased plants, resulting in a "root rot complex." If fungicides are to be used effectively as part of a disease management strategy, the causal agent(s) must be identified. Since there is no simple way to distinguish between these fungi in the field, samples of infected plants should be submitted to a plant diagnostic laboratory for positive identification. Diagnosticians identify root and crown rot fungi by looking for fungal structures in plant tissues under a microscope and by growing these fungi on culture media. In addition, there are commercially available detection kits that use antibodies to detect *Rhizoctonia* in affected plant tissue.

DISEASE CONTROL

Once established, *Rhizoctonia* is very difficult to eradicate. Successful disease management requires an approach that utilizes preventive cultural practices. For best results:

- **Plant only in pathogen-free soil or new potting mix.** Commercially available soilless mixes are usually free of pathogens and do not require treatment before use. The addition of composted hardwood bark as an amendment has been shown to suppress disease development. It is advisable to heat-pasteurize mixes that contain soil before use. In landscape areas with a history of this disease, chemical fumigants should be applied before planting. These chemicals may be used by licensed pesticide applicators only.

- **Use only pathogen-free stock.** Inspect plants carefully and purchase only healthy, vigorous stock.

- **Maintain plant vigor.** Select sites that are most appropriate for vigorous plant growth. Maintain proper levels of soil nutrients, moisture, and soil pH. Avoid cultural practices that promote overly succulent growth such as heavy fertilization, over-crowding, and low light. Succulent plants and those under stress are more susceptible to disease.

- **Apply an appropriate fungicide when necessary.** Since no single fungicide will control all fungi, the selection of the proper fungicide depends upon an accurate diagnosis. Application of an inappropriate fungicide may encourage disease by removing beneficial microbes that compete with disease-causing agents. For this reason, and because root rot diseases frequently occur in complexes, it is sometimes advisable to apply a tank-mix of fungicides or to use a combination product.

When applying fungicides, be certain that the plant you intend to treat is on the label. **Always apply fungicides according to label directions.** Fungicides labeled for the control of Rhizoctonia root and crown rot on many herbaceous ornamentals include iprodione (Chipco 26019), PCNB (Terraclor and Turfcide), thiophanate-methyl (Cleary 3336, Domain, and Fungo Flo) and Banrot. For current recommendations, contact your local County Extension Office.

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Plant Disease Control

ROOT AND CROWN ROTS OF HERBACEOUS ORNAMENTALS IN THE LANDSCAPE

DISEASES CAUSED BY THE FUNGUS *PYTHIUM*

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Fungi in the genus *Pythium* are common pests capable of infecting a wide range of herbaceous ornamentals in the landscape. Plants infected with this fungus exhibit a variety of symptoms that include root rots, lower stem (crown) rots, and damping-off of seedlings. *Pythium* species live in the soil and belong to a group of fungi known as the "water molds." Water molds and the diseases they cause are favored by soil conditions that are wet and poorly-drained.

SUSCEPTIBLE PLANTS

Pythium species are common inhabitants of water and soil and are found throughout the world. This fungus can cause diseases on almost all types of flowers, vegetables, cereals, grasses, and on many woody plants. Under environmental conditions favorable for disease, virtually every plant species is vulnerable to attack.

SYMPTOMS

Plants infected with *Pythium* may grow poorly and wilt, even when sufficient water is present in the soil. A close examination of the plant may reveal the presence of brown areas of dead tissue (lesions) on root and crown tissue near the soil surface. When conditions are favorable for disease development, these lesions quickly enlarge to form sunken cankers and large areas of rotted tissue. Severely infected plants die after root systems are killed or when the stems become girdled. Under conditions of high humidity, fluffy fungal threads (mycelium) may be seen on affected tissue.

Germinating seeds and seedlings are extremely vulnerable to attack by *Pythium*. The fungus may cause total collapse and rot (a symptom known as damping-off) either before or immediately after the seedling emerges from the ground. As seedlings mature, they become more resistant to attack, and disease may be limited to crown and root tissue.

DISEASE DEVELOPMENT

Pythium can survive in the soil as resting structures known as oospores or as mycelium associated with dead plant material. When plants are placed into an infested soil, the fungus is stimulated by substances released from the plant roots. The growth of *Pythium* is favored by wet soil conditions. Disease is most severe when the soil is wet for prolonged periods of time or when the temperature is too high or too low for optimal plant growth. Plants that are stressed or that are overly succulent are more susceptible to attack. *Pythium* is also a problem in locations where the same crop is planted year after year.

DIAGNOSIS

Pythium is one of a group of fungi that can cause similar root and crown rot symptoms on herbaceous ornamentals. Frequently, more than one of these fungi are associated with diseased plants, resulting in a "root rot complex." If fungicides are to be used effectively as part of a disease management strategy, the causal agent(s) must be identified. Since there is no simple way to distinguish between these fungi in the field, samples of infected plants should be submitted to a plant diagnostic laboratory for positive identification. Diagnosticians identify root and crown rot fungi by looking for fungal structures in plant tissues under a microscope and by growing these fungi on culture media. In addition, there are commercially available detection kits that use antibodies to detect *Pythium* in affected plant tissue.

DISEASE CONTROL

Pythium is very difficult to eradicate once it has become established. Successful disease management requires an approach that utilizes preventive cultural practices. For best results:

- **Plant only in pathogen-free soil or new potting mix.** Commercially available soilless mixes are usually free of pathogens and do not require treatment before use. The addition of composted hardwood bark as an amendment has been shown to suppress disease development. It is advisable to heat-pasteurize mixes that contain soil before use. In landscape areas with a history of this disease, chemical fumigants should be applied before planting. These chemicals may be used by licensed pesticide applicators only.

- **Use only pathogen-free stock.** Inspect plants carefully and purchase only healthy, vigorous stock.

- **Maintain plant vigor.** Select planting sites that are most appropriate for vigorous growth. Maintain proper levels of soil nutrients, moisture, and soil pH. Avoid cultural practices that promote overly succulent growth such as heavy fertilization, over-crowding, and low light. Succulent plants and those under stress are more susceptible to disease.

- **Apply an appropriate fungicide when necessary.** Since no single fungicide will control all fungi, the selection of the proper fungicide depends upon an accurate diagnosis. Application of an inappropriate fungicide may encourage disease by removing beneficial microbes that compete with disease-causing agents. For this reason, and because root rot diseases frequently occur in complexes, it is sometimes advisable to apply a tank-mix of fungicides or to use a combination product.

To avoid harming vegetation, be certain that the plant that you intend to treat is on the fungicide label. **Always apply fungicides according to label directions.** Fungicides currently labeled for the control of *Pythium* on many ornamental plants include fosetyl-aluminum (Aliette), propamocarb-HCl (Banol), metalaxyl (Subdue), ethazole (Truban and Terrazole), and Banrot. For current recommendations, contact your local County Extension Office.

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Plant Disease Control

EFFECTS OF OZONE, FLUORIDE, AND SULFUR DIOXIDE POLLUTION ON LANDSCAPE PLANTS

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INTRODUCTION

Three pollutants, **OZONE**, **FLUORIDE**, and **SULFUR DIOXIDE** are the most common causes of air pollutant problems in the landscape. Prolonged or repeated exposure to these pollutants may result in foliar discoloration and decreased growth and vigor of vegetation. Within any plant group, there is often a wide variation in sensitivity. For instance, white pines vary greatly in their response to most common air pollutants.

DIAGNOSING POLLUTION INJURY

To diagnose pollution injury, compare the affected vegetation with other vegetation in the vicinity. Generally, more than one species of plant will be affected. Visually examine the plants and compare the symptoms with those described in the literature. In the cases of fluoride and sulfur dioxide, verify the existence of a pollution source in the area. Symptoms due to fluoride pollution are more prominent on the side of the plant facing the pollution source.

In general, deciduous species develop symptoms more rapidly than conifers, but they do not accumulate toxic levels of pollutants from year to year because affected leaves are shed. Although conifers develop symptoms more slowly, they can accumulate pollutants to lethal levels because affected needles may persist for several years. Ultimately, the type of injury that results from pollution depends on the pollutant, its dose (concentration x time), the time of year, the plant species involved, the genetic make-up of the vegetation, and the age of the foliage affected.

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FLUORIDE

Fluoride is a natural component of soil, rocks, and various minerals. Toxic quantities of fluoride may be released into the atmosphere when materials containing fluorides are heated or treated with acid during industrial processing. The primary sources of fluoride pollution in New Jersey are glass and brick manufacturing plants.

In deciduous species, tissue along leaf margins turn light green and appear water soaked when first exposed to fluoride. These areas later turn brown. A dark-brown band may appear where the toxin accumulates between the affected and green, inner leaf tissue. Eventually, the entire leaf turns brown. In conifers, fluoride injury is most evident on first-year needles. The tips of these needles turn reddish-brown from the tip toward the needle base. Older needles are rarely affected. Gladiolus, one of the most susceptible plants to fluoride, is often used as an "indicator plant" for fluoride pollution. Utilize fluoride tolerant species in areas with known fluoride toxicity problems.

SENSITIVITY TO FLUORIDE

FLUORIDE SENSITIVE PLANTS	FLUORIDE TOLERANT PLANTS
apricot	ailanthus
ash	birch
boxelder	cherry, flowering
douglas-fir	dogwood
gladiolus	elm, American
grape	hawthorn
larch	juniper
mahonia	linden, American
maple	mountain ash
oak	mulberry
peach	pear
pine, mugo	pyracantha
pine, Scotch	sassafras
pine, white	spirea
poplar	sweetgum
redbud	sycamore
rhododendron	virginia creeper
spruce, blue	willow
sumac	
tulip	
walnut, black	
yew	

SULFUR DIOXIDE

Sulfur dioxide is released into the atmosphere by the combustion of fossil fuels and by the smelting and refining of ores. The primary source of sulfur dioxide pollution in New Jersey is the burning of coal to generate electricity. Most damage to vegetation occurs in urban areas and in the vicinity of large power plants.

Sulfur dioxide enters leaves through natural openings in the plant called stomata. Plants are able to utilize small amounts of sulfur dioxide, but accumulations can cause injury and death. Acute injury occurs when plants are exposed to high levels of sulfur dioxide for a short time. In deciduous species exposed to sulfur dioxide, tissue between the leaf veins turns yellow, white, or brown. The veins, however, remain green. Unlike ozone, where injury appears only on the upper leaf surface, injury due to sulfur dioxide is evident on both the upper and lower surfaces of affected leaves. In conifers, a reddish-brown discoloration begins at the needle tip and progresses toward the needle base.

Chronic injury occurs when plants are exposed to low levels of sulfur dioxide for long periods of time. In most deciduous species, this type of injury is characterized by a general yellowing, or chlorosis, of the leaves. Older conifer needles turn yellow and are shed prematurely. Blackberry, raspberry, pumpkin, and squash are useful "indicator plants" for this pollutant.

SENSITIVITY TO SULFUR DIOXIDE

SULFUR DIOXIDE SENSITIVE PLANTS	SULFUR DIOXIDE TOLERANT PLANTS
apple	ash
ash	boxelder
aster	dogwood
birch	gum, black
catalpa	juniper
elm, American	maple
larch	spruce
mulberry	sycamore
pine, white	tuliptree
poplar	
spruce, blue	
violet	
zinnia	

OZONE

Ozone is a by-product of automobile and industrial combustion. Ozone is formed when nitrous oxides and hydrocarbons released from incomplete combustion undergo chemical reactions in the presence of sunlight. Injury to vegetation from ozone can occur at long distances from the hydrocarbon source. As a result, ozone injury is becoming more prevalent each year in rural as well as in urban areas.

In deciduous trees, ozone pollution results in a breakdown of chlorophyll causing small "flecks" on the upper leaf surface between the larger veins. These flecks range in color from white to orange-red. In conifers, yellow flecks (1/8 to 1/4 inch in diameter) frequently occur on affected needles. Yellow bands that girdle the needle may also form, causing the tip of the needle to turn brown and die. In general, herbaceous plants are more sensitive to ozone than are woody plants. White pine can serve as an "indicator plant" since most plants within this species are highly sensitive to this pollutant.

SENSITIVITY TO OZONE

OZONE SENSITIVE PLANTS		OZONE TOLERANT PLANTS
ailanthus	mulberry	birch, European white
alder	oak, white	boxwood
ash, green	petunia	douglas-fir
ash, white	pine, Austrian	locust, black
boxelder	pine, Scotch	maple
boxwood	pine, white	pine, Japanese black
carnation	poplar	pine, red
catalpa	privet	oak, red
chrysanthemum	snowberry	spruce, blue
crabapple	spirea	spruce, Norway
grape	sweetgum	walnut, black
honeylocust	sycamore	
larch	tuliptree	
lilac	willow, weeping	
linden	zelkova	
maple, silver		

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Plant Disease Control

THE IMPACT OF DE-ICING SALT ON ROADSIDE VEGETATION

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INTRODUCTION

Vegetation is exposed to salt from a variety of sources. These sources include sea water, salt-laden rain and snow, fertilizers, pesticides, sewage effluent, and de-icing products. In New Jersey, the demand for ice-free roadways and sidewalks has led to an increase in the use of de-icing salts. Runoff from treated pavement contains dissolved salts that can injure adjacent vegetation. In plants sensitive to excessive salt, affected foliage may appear "scorched" and drop prematurely. In severe cases, the death of twigs, branches, and sometimes the entire plant, may occur.

DEICING SALTS AND THEIR USES

Deicing salts make roadways, driveways, and sidewalks safer by melting snow and ice. In the Northeast, up to 1/2 inch of salt is applied to road surfaces each year. During the 1980s, de-icing salt was applied annually on the New Jersey Turnpike and the Garden State Parkway at the rate of 400 lbs per mile. De-icing salts are usually applied during snow storms before the snow can accumulate. These salts dissolve in water to form a brine that possesses a freezing point much lower than water. The brine melts ice and helps to prevent the formation of more ice as temperatures drop.

The two most commonly applied de-icing salts are sodium chloride and calcium chloride. Although calcium chloride is a better material for melting ice, sodium chloride (rock salt) is used most widely because it is relatively inexpensive and is easier to handle. To improve traction, de-icing salts are usually mixed with abrasives. These abrasives, which include sand, cinders, gravel, and sawdust, can accumulate along roadways and cause drainage problems.

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HOW SALT AFFECTS VEGETATION

Plants become injured by salt when roots and foliage come into contact with salt-laden water. Salted water percolates down through the soil and comes into contact with soil particles, soil microbes, and plant roots. Salt injures vegetation in four ways:

- **Salt increases water stress.** Salt accumulates on the surface of affected plant tissue. In the root zone, water molecules are held very tightly by salt ions. Roots must expend considerable energy to absorb water from a salt solution. Although sufficient water may be present in the soil, the plant may have difficulty absorbing it and a condition known as "physiological drought" occurs. In sensitive species, this may result in depressed growth and yield.

- **Salt affects soil quality.** The sodium ion component in salt becomes attached to soil particles and displaces soil elements such as potassium and phosphorus. As a result, soil density and compaction increases and drainage and aeration are reduced. Plant growth and vigor are poor under these conditions.

- **Salt affects mineral nutrition.** When the concentration of both the sodium and chloride components of salt in the root zone is excessive, plants preferentially absorb these ions instead of nutrients such as potassium and phosphorus. When this occurs, plants may suffer from potassium and phosphorus deficiency.

- **Salt accumulates within plants.** The chloride component of salt is absorbed by roots and foliage. Although this ion can accumulate in any plant part, it is usually concentrated in actively growing tissue. Plants repeatedly exposed to salt over long periods of time may accumulate chloride ions to toxic levels, resulting in leaf burn and twig die-back. The injury a plant sustains increases with an increase in foliar chloride levels. Foliage in direct contact with road salt sprayed by tires and wind becomes desiccated and may appear "burned."

Unlike animals, plants do not have mechanisms to excrete excess salt from tissues and can only "shed" salt in dead leaves and needles. Because conifers do not shed leaves on a yearly basis, they tend to suffer damage from accumulated salt more easily than do deciduous trees.

HOW PLANTS RESPOND TO EXCESSIVE SALT

Plant species vary in their tolerance to salt exposure (Table 1). Plants that are tolerant of salt grow as well in saline soils (soils high in salt) as they do under normal conditions. Salt tolerance is directly related to the concentration of chloride ions in the foliage. Many herbaceous plants such as grasses adapt fairly readily to high salt levels. Among woody plants, tolerance varies with the species. Plant species with waxy foliage are generally more tolerant of salt spray.

In salt-sensitive plants, exposure to salt often results in an unthrifty appearance and poor growth. Other symptoms of salt injury include stunted leaves, heavy seed loads, twig and branch die-back, leaf scorch, and premature leaf drop. In addition, plants stressed by excessive salt concentrations are more susceptible to biotic diseases and insect pests. The extent of injury a plant sustains in response to salt depends on:

- **The kind and amount of salt applied.** Although sodium chloride (rock salt) is less expensive and easier to handle than calcium chloride, it is also more damaging to vegetation.

- **The volume of fresh water applied.** In well-drained soils, salt is easily leached by water low in salt. Salts tend to accumulate, however, in poorly-drained soils, so the potential for damage to vegetation in these soils is high. High volumes of water, whether from rainfall or melting snow, will decrease the possibility of injury. Rainfall also washes salt from foliage surfaces.

- **The distance plants are situated from treated pavements.** Plants within the "spray zone" of moving vehicles are more likely to sustain salt injury. Injury is usually most evident on the side of the plant that faces the highway.

- **The direction of surface-water flow.** The channeling of drainage water away from susceptible plants will prevent salt from coming into contact with plant roots. If plants are situated up-slope or away from drainage areas, they are less likely to be affected.

- **The time of year salt is applied.** Salt applied in late winter and early spring is more likely to damage vegetation than is salt applied in early- to mid-winter. This is because there is less time for winter snow and precipitation to leach salt from the root zone before growth resumes in the spring. The depth and duration of soil freezing is also important. Dormant trees continue to absorb water and nutrients in unfrozen soils. Salted water can percolate through frozen soils, reaching active plant roots in unfrozen soil horizons.

MINIMIZING SALT INJURY

The best solution to the de-icing salt problem is to **prevent** contamination. Homeowners can use abrasives instead of salt when treating driveways and walkways. If vegetation is located in areas where salt spray occurs, barriers or screens can be erected to protect plants during the winter months. Anti-desiccants may also help prevent injury when applied to evergreen foliage along the coast or where de-icing salt will be used. County, state, and municipal officials can help prevent salt injury by carefully training equipment operators and frequently calibrating equipment.

Once soil becomes contaminated with salt, damage can be reduced by leaching the salt with fresh water as soon as possible after exposure. Under certain circumstances, incorporation of gypsum at the rate of 50 lb./1000 sq. ft. into the top six inches of soil at the drip-line of trees may also be helpful. Furthermore, foliage exposed to salt spray may be washed with salt-free water to remove deposited salt.

When landscaping, place trees and shrubs that are sensitive to salt as far as possible from problem areas. Select planting sites that are not subject to salt-contaminated waters, and place shallow diversion ditches between roadways and plantings. When vegetation must be placed near roadways, utilize salt-tolerant plants.

Table 1. Salt tolerance of common woody landscape plants.

SHRUBS	
Tolerant	**Sensitive**
Autumn elaeagnus (*Elaeagnus umbellata*) bayberry (*Myrica* spp.) honeysuckle (*Lonicera* spp.) Pfitzer juniper (*Juniperus chinensis* 'Pfitzerana') California privet (*Ligustrum ovalifolium*) rugosa rose (*Rosa rugosa*) yucca (*Yucca filamentosa*)	Japanese barberry (*Berberis thunbergii*) boxwood (*Buxus* spp.) winged euonymus (*Euonymus alata*) multiflora rose (*Rosa multiflora*) Van houtte spirea (*Spiraea x vanhouttei*) viburnum (*Viburnum* spp.)
DECIDUOUS TREES	
Tolerant	**Sensitive**
green ash (*Fraxinus pennsylvanicum*) boxelder (*Acer negundo*) black cherry (*Prunus serotina*) Siberian elm (*Ulmus pumila*) honeylocust (*Gleditsia triacanthos*) black locust (*Robinia pseudoacacia*) bur oak (*Quercus macrocarpa*) English oak (*Quercus robur*) red oak (*Quercus rubra*) white oak (*Quercus alba*) Russian olive (*Elaeagnus angustifolia*) white poplar (*Populus alba*) weeping willow (*Salix babylonica*)	beech (*Fagus* spp.) flowering dogwood (*Cornus* spp.) shagbark hickory (*Carya ovata*) ironwood (*Carpinus* spp.) American linden (*Tilia americana*) little-leaf linden (*Tilia cordata*) red maple (*Acer rubrum*) silver maple (*Acer saccharinum*) sugar maple (*Acer saccharum*) sycamore (*Platanus* spp.) black walnut (*Juglans nigra*)
EVERGREENS	
Tolerant	**Sensitive**
red cedar (*Juniperus virginiana*) Austrian pine (*Pinus nigra*) Japanese black pine (*Pinus thunbergiana*) pitch pine (*Pinus rigida*) white spruce (*Picea glauca*) yew (*Taxus* spp.)	balsam fir (*Abies balsamea*) Douglas-fir (*Pseudotsuga menziesii*) Canadian hemlock (*Tsuga canadensis*) eastern white pine (*Pinus strobus*) red pine (*Pinus resinosa*)

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Plant Disease Control

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Common Spring-Time Diseases of Woody Ornamentals in the Nursery

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Many of the disease problems encountered in the nursery occur in the spring. Spring-time diseases are more severe when plants are under stress, have suffered extensive winter damage, or when the weather is cool and rainy. The sections that follow briefly describe some common diseases that occur in the nursery in spring.

It is important to remember that trees and shrubs in poor health are more susceptible to disease. **Improving plant vigor** is the most important management tool for disease control in the nursery.

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Specific Spring-Time Diseases Caused By Fungi

Leaf Spots

Fungal leaf spot on sugar maple.
(Courtesy B. B. Clarke)

Leaf spots are very common and can occur on many species of ornamental plants. **Leaf spots** are caused by leaf inhabiting fungi that discolor and kill small, discrete regions of tissue between or on the leaf veins. Frequently, these spots have a light-colored center with a distinct dark-colored border. Individual spots may grow together to form larger **leaf blotches.** Most leaf spot fungi produce spores in dead leaf litter on the ground. Spores are splashed or carried by wind to developing leaf tissue at budbreak. The development of **leaf spots** is favored by abundant moisture and cooler temperatures. Severe spotting can occur when moisture remains on leaf surfaces for long periods of time. Fungicides are effective only if they are present on leaf surfaces at the time the fungi are producing spores. Fungicides applied after **leaf spots** are visible are ineffective. Most damage caused by the fungi that cause **leaf spots** is merely cosmetic.

Proper Management

Improve plant vigor and reduce inoculum by removing leaf litter. Irrigate in the early morning hours and avoid overhead watering to prevent excessive moisture from remaining on foliage.

Chemical Control

Apply chlorothalonil, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl according to label recommendations.

Anthracnose

Anthracnose lesions on veins of sycamore leaves.

Anthracnose is a common disease of many shade tree species, particularly sycamore, ash, oak, maple, and walnut. Diseased leaves appear "scorched" along veins and leaf margins. Twigs and branches may die back if infection is severe or if the tree is in poor health. Leaves infected with **anthracnose** are often shed. As with leaf spot diseases, **anthracnose** is more severe when moisture remains on leaf surfaces for long periods of time. Since **anthracnose** does not usually cause serious damage to healthy trees, application of fungicides is recommended only when it is necessary to keep trees as blemish-free as possible.

Proper Management

Improve plant vigor, prune dead branches, avoid planting highly sensitive plants, and remove leaf litter to reduce inoculum. Irrigate in the early morning hours and avoid overhead watering to prevent excessive moisture from remaining on foliage.

Chemical Control

Chlorothalonil, copper, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl provide fair control of the leaf spot phase of this disease only. Apply fungicides according to label recommendations.

Apple Scab

Apple scab lesions on the foliage of crabapple. (Courtesy of E.M. Dutky)

Apple scab (caused by the fungus *Venturia*) is the most common disease of apple and crabapple. **Apple scab** and related **scab** diseases can also be a problem on other rosaceous ornamentals such as mountain ash, hawthorn, cotoneaster, and pyracantha. Olive-colored spots (1/4 inch in diameter) with fuzzy borders can be seen on leaves and petals. Corky-looking lesions may appear on twigs and fruit. Severely infected leaves, petals, and fruit may turn brown and drop prematurely. There are cultivars of crabapple and other ornamentals with good resistance to this disease.

Proper Management

Improve plant vigor, use resistant cultivars, and remove leaf litter to reduce inoculum.

Chemical Control

Apply chlorothalonil, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl according to label recommendations.

Cedar-Apple and Quince Rusts

Gall and spore clusters of cedar-apple rust on eastern red cedar.

Hawthorn fruits infected with quince rust. (Courtesy E.M. Dutky)

Rust diseases are unique because the fungi that cause them often require more than one host plant to survive. **Cedar-apple rust** and **quince rust** affect two groups of highly utilized landscape plants.

The **cedar-apple rust** fungus overwinters in galls that may grow to several inches in diameter on eastern red cedar and several other junipers. In the spring, brightly-colored, gelatinous horns emerge from the galls during wet weather. These horns consist of masses of spores that are spread by wind to newly-emerging apple, crabapple, and hawthorn leaves and fruit. By mid-summer, rusty or orange-colored spots appear on infected leaves. In mid- to late summer, spores produced in these spots are carried by the wind to cedar and juniper. On susceptible crabapple cultivars, rust causes premature defoliation, stunted growth, and poor quality fruit.

The disease cycle of **quince rust** is similar to cedar-apple rust. The galls of **quince rust** on eastern red cedar and other junipers are small and spindle-shaped. **Quince rust** affects fruit, young stems, and petioles on rosaceous hosts such as apple, crabapple, hawthorn, quince, mountain ash, and cotoneaster. Fruits are stunted and killed, and twigs and petioles become swollen and distorted, often resulting in death.

Proper Management

On coniferous hosts, prune affected branches 6 to 8 inches below galls during dry weather with sterilized pruning tools. Use cultivars of crabapple and other rosaceous plants that are resistant to **rusts**. If practical, remove the alternate host within a 1/4-mile radius.

Chemical Control

On juniper, apply mancozeb or mancozeb plus thiophanate-methyl according to label recommendations. On rosaceous hosts, apply chlorothalonil, mancozeb, triadimefon, or mancozeb plus thiophanate-methyl according to label recommendations.

Dogwood Anthracnose or Decline

Foliar symptoms of dogwood anthracnose. (Courtesy J.L. Peterson)

Dogwood anthracnose or decline, caused by the fungus *Discula*, is primarily a disease of flowering dogwood (*Cornus florida*). Tan-colored leaf spots with purple margins form on developing leaves and flower bracts. These spots grow together, forming large blotches on leaf blades and along leaf margins. Infected leaves eventually die. The fungus may continue to grow down into the petioles and branches, resulting in the death of twigs and branches. Brown, elliptical cankers may form at the base of dead branches. Drought, winter injury, and environmental stress predispose dogwood to anthracnose. Kousa dogwood (*Cornus kousa*) is resistant to this disease.

Proper Management

Improve plant vigor, avoid moisture stress, avoid wounding, and prune affected branches 6 to 8 inches below diseased tissue during dry weather with sterilized pruning tools. Avoid planting dogwoods in shady or crowded areas.

Chemical Control

Chlorothalonil and propiconazole (Banner) provide fair control of the leaf spot phase of this disease only. Apply fungicides according to label recommendations.

Rhabdocline Needlecast

Rhadbocline needlecast on previous year's growth of Douglas fir. (Courtesy B.B. Clarke)

Rhabdocline needlecast, caused by the fungi *Rhabdocline pseudotsugae and R. weirii*, affects only Douglas fir. Irregularly shaped, reddish-brown spots surrounded by green tissue appear on the previous year's needles by early spring. Orange fruiting bodies containing spores are produced on the lower surface of affected tissue at budbreak. These spores are splashed to the current season's needles in late April and early May. In early summer, older infected needles are shed (or cast). Symptoms on newly infected needles do not appear until the following fall or winter. Lower branches are more severely affected. The development of **Rhabdocline needlecast** is favored by abundant moisture and cool temperatures. Trees on north-facing slopes or in low-lying areas with poor air circulation are more likely to become infected. Close plant spacing and poor weed control contribute to conditions of high humidity that are favorable for disease development.

Proper Management

For optimal control, improve plant vigor, increase plant spacing, and control weeds.

Chemical Control

Apply chlorothalonil when candles are 1/2-inch long. Repeat fungicide applications at 3- to 4-week intervals until conditions are no longer favorable for disease development. Apply fungicides according to label recommendations.

Juniper Tip Blights

Twig dieback of juniper caused by the fungus *Kabatina*. (Courtesy B.B. Clarke)

Juniper tip blights are caused by the fungi *Phomopsis* and *Kabatina*. Tips of newly developing branches become infected with *Phomopsis* in the spring and turn brown by summer. Infected growth is killed back to the previous season's wood. Mature tissue is resistant to **Phomopsis tip blight**. **Kabatina blight** symptoms can occur throughout the year and only on wounded twigs older than one year. Plants stressed by moisture extremes, insect infestations, and winter injury are susceptible to *Kabatina*. Environmental stress and high humidity in the canopy due to close spacing increase the severity of **tip blight.**

Proper Management

Improve plant vigor, avoid wounding, prune affected tissue, and space plants adequately to ensure good air circulation. Control insect pests when present.

Chemical Control

To control *Phomopsis*, apply thiophanate-methyl or thiophanate-methyl plus mancozeb at budbreak according to label recommendations. There are no fungicides recommended for the control of *Kabatina*.

Diplodia (or Sphaeropsis) Shoot Blight and Canker

Symptoms of Diplodia shoot blight of pine usually begin on the lower branches.
(Courtesy J.L. Peterson)

Diplodia (or Sphaeropsis) Shoot Blight and Canker affects 2- and 3-needle pines and is most devastating on Austrian, mugo, and Scots pines. The fungus *Sphaeropsis* infects and kills developing needles, resulting in dead candles that are much shorter than healthy ones. Sunken cankers may form on branches and stems, killing the tissue beyond the cankers. The lower branches of pines are usually affected first. Tiny, black, spore-producing structures called "fruiting bodies" can be seen with the aid of a hand lens at the base of dead needles and on cones. Spores are released from these fruiting bodies in cool, rainy weather and are transmitted to susceptible tissue. This disease is more severe on trees that are stressed. Japanese black pine is tolerant of this disease and offers an attractive alternative where **Diplodia shoot blight** has been a problem in the past.

Proper Management

Improve plant vigor and prune affected branches 6 to 8 inches below diseased tissue during dry weather with sterilized pruning tools. Remove as much plant debris as possible and use tolerant species.

Chemical Control

Apply Tersan 1991 WP or Cleary 3336 WP according to label recommendations.

Nectria cankers

Nectria canker on improperly pruned maple. Note orange spore clusters in affected bark.
(Courtesy B.B. Clarke)

Nectria cankers are common on a wide variety of shade trees and other woody ornamentals. *Nectria* is an opportunistic fungus that infects twigs, branches, and trunks through wounds and at the base of dead branches. This fungus can cause both annual and perennial cankers. Annual cankers are common on twigs and branches injured by freezing, drought stress, mechanical injuries, or other diseases. As cankers enlarge, twigs are girdled and killed in a single season. A cut made into the wood with a pocket knife reveals a sharp transition between white, healthy wood and brown, infected wood. Perennial cankers enlarge yearly, encircling the branches and eventually killing tissue beyond the canker. With each successive year of infection, a "bulls-eye" pattern may develop. **Nectria canker** can be identified by the bright orange fruiting bodies that form in the center of the cankers.

Proper Management

Improve plant vigor and avoid moisture stress, wounding, winter injury, and mechanical injury. Prune affected branches (when practical) 6 to 8 inches below infected tissue, during dry weather, with sterilized pruning tools.

Chemical Control

None recommended.

Pachysandra Leaf and Stem Blight

Pachysandra leaves infected with *Volutella*. Note the target-shaped lesions. (Courtesy S. Davis)

Pachysandra leaf and stem blight is caused by the fungus *Volutella*. Pachysandra is most susceptible to this disease when it has suffered from winter injury, moisture or heat stress, mechanical injury, or has a problem with scale insects. Large leaf spots, which have a "bulls-eye" pattern, appear on leaves. Cankers form on petioles and stems that produce characteristic pink-colored fruiting bodies within several weeks in wet weather. Occasional thinning and removal of leaf litter reduces humidity and helps to keep disease severity to a minimum.

Proper Management

Improve plant vigor and avoid moisture stress, winter injury, and mechanical injury. Remove leaf litter to reduce humidity and control scale insects, if present.

Chemical Control

To control **leaf and stem blight**, apply chlorothalonil or mancozeb plus thiophanate-methyl according to label recommendations. If scale insects are present, apply 2 percent dormant oil, acephate, malathion, diazinon, or dimethoate according to label recommendations.

Ovulinia Petal Blight

Dried flowers infected by Ovulinia petal blight cling to rhododendron foliage. Note the dark, round sclerotia (resting structures) imbedded in the dried flower.
(Courtesy J. L. Peterson)

Ovulinia petal blight, one of the most common diseases of rhododendrons and azaleas, affects only the flowers. Small, water-soaked spots appear on infected petals. These spots rapidly enlarge until the flower becomes slimy, limp, and turns prematurely brown. Entire trusses may become diseased almost simultaneously. Most infected petals adhere to the plant but some may fall to the ground. Six to eight weeks following infection, small, black sclerotia (resting structures) develop on infected petals. These sclerotia germinate in the spring and produce fruiting structures called apothecia. Spores are forcibly ejected from the apothecia, striking blossoms close to the ground. Wet weather at flowering time enhances disease development.

Proper Management

Remove dead trusses and fallen petals as soon after bloom as possible to reduce disease spread. Maintain plant vigor.

Chemical Control

Mist chlorothalonil, triadimefon, thiophanate-methyl, or mancozeb plus thiophanate-methyl onto plants from the time flowers begin to show color until flowering has ceased at intervals stated in label recommendations.

Atropellis Canker

Atropellis canker on eastern white pine.
(Courtesy B. B. Clarke)

Atropellis canker (caused by the fungus *Atropellis*) is most noticeable on Scots and eastern white pine in the spring. The fungus enters trees predominantly at the branch nodes via small wounds or cracks in the bark. Elliptical, resin-soaked cankers form and enlarge over a period of several years, girdling small twigs and branches. Infected wood beneath the bark is stained dark-chocolate brown to black. Weakened or stressed trees are most susceptible to *Atropellis* infection.

Proper Management

Maintain plant vigor. Prune affected branches 6 to 8 inches below diseased tissue during dry weather with sterilized pruning tools. Remove severely infected trees.

Chemical Control

None recommended.

Phytophthora Root Rot

Container grown rhododendron displaying varying stages of Phytophthora wilt.

Phytophthora root rot (caused by the fungus *Phytophthora cinnamomi*) affects a wide variety of nursery crops including azalea, rhododendron, pieris, aucuba, camellia, dogwood, Japanese holly, juniper, hemlock, false cypress, white pine, and yew. This soil-borne fungus attacks the roots of susceptible plants, resulting in root rot and death. Affected plants become yellow and stunted and will eventually wilt and die. A cut made into the stem of an infected plant at the soil-line will reveal a red-brown discoloration of the wood just beneath the bark. Plants in low, wet, and poorly drained soils are susceptible to **Phytophthora root rot**.

Proper Management

Utilize good sanitation practices during propagation and production. Plant only in well-drained soilless media, preferably amended with composted hardwood bark. Bark improves drainage and releases compounds that are antagonistic to the fungus. Ensure proper drainage and prevent over-watering. Plant resistant cultivars.

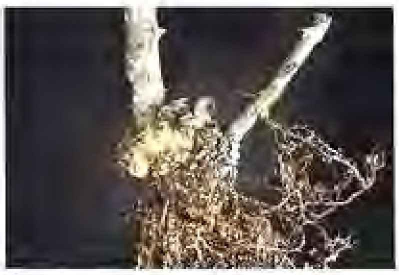
Chemical Control

Apply a drench of propamocarb, ethazole, metalaxyl, or fosetyl-Al in May and repeat at 4-6 week intervals as needed. Apply chemicals according to label recommendations.

Specific Spring-Time Diseases Caused By Bacteria

Crown Gall

Crown gall is caused by the soil-borne bacterium *Agrobacterium tumefaciens*. More than 600 species of plants are susceptible to **crown gall**. This bacterium enters plant roots and stems through wounds near the soil line. Infection by the bacterium causes tissue at the crown area to grow rapidly, resulting in the formation of galls. These galls consist chiefly of host tissue. Following the initial infection, galls soon form on other parts of the plant. To prevent **crown gall**, avoid wounding during transplanting and cultivation. Once the disease is present in a plant, pruning individual galls will not prevent galls from forming on other parts of the plant.



Crown gall on rose. (Courtesy B. B. Clarke)

Proper Management

Improve plant vigor, avoid wounding, and remove entire plants when galls are observed. Utilize resistant plants in spots where diseased plants have been observed previously.

Chemical Control

Galltrol-A may be used as a pre-plant dip at transplanting.

Fire Blight

Fire Blight on mountain ash. (Courtesy B. B. Clarke)

Fire blight, caused by the bacterium *Erwinia amylovora*, can occur on many rosaceous plants, including crabapple, cotoneaster, hawthorn, mountain ash, pyracantha, and pear. In the spring, bacteria ooze from existing cankers on infected plants. The bacteria are carried to healthy blossoms and branches by insects that are attracted to the ooze. The bacteria are also spread by splashing rain. Twigs and branches infected with the **fire blight** bacterium die rapidly and appear scorched. Cankers form at the base of infected branches.

Proper Management

Improve plant vigor, avoid heavy spring fertilization, and use resistant cultivars. Prune affected branches during dry weather. Remove branches 6 to 8 inches below diseased tissue using sterilized pruning tools.

Chemical Control

Copper or streptomycin according to label recommendations.

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Plant Disease Control

RUTGERS COOPERATIVE EXTENSION · NEW JERSEY AGRICULTURAL EXPERIMENT STATION

Common Spring-Time Diseases of Woody Ornamentals in the Landscape

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Many disease problems in the landscape and on home grounds occur in the spring. These problems are worse when plants are under stress, have suffered extensive winter damage, or when the weather is cool and rainy. The sections that follow briefly describe some common problems that occur in the spring in the landscape.

It is important to remember that trees and shrubs in poor health are more susceptible to disease. **Improving plant vigor** is the most important aspect of disease control in the home landscape.

May 1992

THE STATE UNIVERSITY OF NEW JERSEY
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Specific Spring-Time Diseases Caused By Fungi

Leaf Spots

Fungal leaf spot on sugar maple.
(Courtesy B. B. Clarke)

Leaf spots are very common and can occur on many species of ornamental plants. **Leaf spots** are caused by leaf-inhabiting fungi that discolor and kill small, discrete regions of tissue between or on the leaf veins. Frequently, these spots have a light-colored center with a distinct dark-colored border. Individual spots may grow together to form larger **leaf blotches**. Most leaf spot fungi produce spores in dead leaf litter on the ground. Spores are splashed or carried by wind to developing leaf tissue at budbreak. The development of **leaf spots** is favored by abundant moisture and cooler temperatures. Severe spotting can occur when moisture remains on leaf surfaces for long periods of time. Fungicides are effective only if they are present on leaf surfaces at the time the fungi are producing spores. Fungicides applied after **leaf spots** are visible are ineffective. Most damage caused by the fungi that cause **leaf spots** is merely cosmetic.

Proper Management

Improve plant vigor and reduce inoculum by removing leaf litter. Irrigate in the early morning hours and avoid overhead watering to prevent excessive moisture from remaining on foliage.

Chemical Control

Apply chlorothalonil, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl according to label recommendations.

Anthracnose

Anthracnose lesions on veins of sycamore leaves.

Anthracnose is a common disease of many shade tree species, particularly sycamore, ash, oak, maple, and walnut. Diseased leaves appear "scorched" along veins and leaf margins. Twigs and branches may die back if infection is severe or if the tree is in poor health. Leaves infected with **anthracnose** are often shed. As with leaf spot diseases, **anthracnose** is more severe when moisture remains on leaf surfaces for long periods of time. Since **anthracnose** does not usually cause serious damage to healthy trees, application of fungicides is recommended only when it is necessary to keep trees as blemish-free as possible.

Proper Management

Improve plant vigor, prune dead branches, avoid planting highly sensitive plants, and remove leaf litter to reduce inoculum. Irrigate in the early morning hours and avoid overhead watering to prevent excessive moisture from remaining on foliage.

Chemical Control

Chlorothalonil, copper, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl provide fair control of the leaf spot phase of this disease only. Apply fungicides according to label recommendations.

Apple Scab

Apple scab lesions on the foliage of crabapple. (Courtesy E.M. Dutky)

Apple scab (caused by the fungus *Venturia*) is the most common disease of apple and crabapple. **Apple scab** and related **scab** diseases can also be a problem on other rosaceous ornamentals such as mountain ash, hawthorn, cotoneaster, and pyracantha. Olive-colored spots (1/4 inch in diameter) with fuzzy borders can be seen on leaves and petals. Corky-looking lesions may appear on twigs and fruit. Severely infected leaves, petals, and fruit may turn brown and drop prematurely. There are cultivars of crabapple and other ornamentals with good resistance to this disease.

Proper Management

Improve plant vigor, use resistant cultivars, and remove leaf litter to reduce inoculum.

Chemical Control

Apply chlorothalonil, mancozeb, thiophanate-methyl, or mancozeb plus thiophanate-methyl according to label recommendations.

Cedar-Apple and Quince Rusts

Gall and spore clusters of cedar-apple rust on eastern red cedar.

Hawthorn fruits infected with quince rust. (Courtesy E.M. Dutky)

Rust diseases are unique because the fungi that cause them often require more than one host plant to survive. **Cedar-apple rust** and **quince rust** affect two groups of highly utilized landscape plants.

The **cedar-apple rust** fungus overwinters in galls that may grow to several inches in diameter on eastern red cedar and several other junipers. In the spring, brightly-colored, gelatinous horns emerge from the galls during wet weather. These horns consist of masses of spores that are spread by wind to newly-emerging apple, crabapple, and hawthorn leaves and fruit. By mid-summer, rusty or orange-colored spots appear on infected leaves. In mid- to late-summer, spores produced in these spots are carried by the wind to cedar and juniper. On susceptible crabapple cultivars, rust causes premature defoliation, stunted growth, and poor-quality fruit.

The disease cycle of **quince rust** is similar to cedar-apple rust. The galls of **quince rust** on eastern red cedar and other junipers are small and spindle-shaped. **Quince rust** affects fruit, young stems, and petioles on rosaceous hosts such as apple, crabapple, hawthorn, quince, mountain ash, and cotoneaster. Fruits are stunted and killed, and twigs and petioles become swollen and distorted, often resulting in death.

Proper Management

On coniferous hosts, prune affected branches 6 to 8 inches below galls during dry weather with sterilized pruning tools. Use cultivars of crabapple and other rosaceous plants that are resistant to **rusts**. If practical, remove the alternate host within a 1/4-mile radius.

Chemical Control

On juniper, apply mancozeb or mancozeb plus thiophanate-methyl according to label recommendations. On rosaceous hosts, apply chlorothalonil, mancozeb, triadimefon, or mancozeb plus thiophanate-methyl according to label recommendations.

Juniper Tip Blights

Twig dieback of juniper caused by the fungus *Kabatina*. (Courtesy B.B. Clarke)

Juniper tip blights are caused by the fungi *Phomopsis* and *Kabatina*. Tips of newly developing branches become infected with *Phomopsis* in the spring and turn brown by summer. Infected growth is killed back to the previous season's wood. Mature tissue is resistant to **Phomopsis tip blight**. **Kabatina blight** symptoms can occur throughout the year and only on wounded twigs older than one year. Plants stressed by moisture extremes, insect infestations, and winter injury are susceptible to *Kabatina*. Environmental stress and high humidity in the canopy due to close spacing increase the severity of **tip blight.**

Proper Management

Improve plant vigor, avoid wounding, prune affected tissue, and space plants adequately to ensure good air circulation. Control insect pests when present.

Chemical Control

To control *Phomopsis*, apply thiophanate-methyl or thiophanate-methyl plus mancozeb at budbreak according to label recommendations. There are no fungicides recommended for the control of *Kabatina*.

Oak Leaf Blister

Oak leaf blister symptoms on pin oak. (Courtesy E.M. Dutky)

Light green pockets or blisters, about 1/4 inch in diameter, occur on the leaves of many different species of oak. These blisters resemble galls caused by insects; however, with **oak leaf blister**, the upper leaf surface is swollen and the underside of the blister is depressed. As the blisters age, they become dry and brown, resembling leaf spots. The development of **oak leaf blister** is favored by wet weather. This disease does not seriously harm healthy trees and control with fungicides is not recommended.

Proper Management

Improve plant vigor.

Chemical Control

None recommended.

Dogwood Anthracnose or Decline

Foliar symptoms of dogwood anthracnose. (Courtesy J.L. Peterson)

Dogwood anthracnose or decline caused by the fungus *Discula*, is primarily a disease of flowering dogwood (*Cornus florida*). Tan-colored leaf spots with purple margins form on developing leaves and flower bracts. These spots grow together, forming large blotches on leaf blades and along leaf margins. Infected leaves eventually die. The fungus may continue to grow down into the petioles and branches, resulting in the death of twigs and branches. Brown, elliptical cankers may form at the base of dead branches. Drought, winter injury, and environmental stress predispose dogwood to anthracnose. Kousa dogwood (*Cornus kousa*) is resistant to this disease.

Proper Management

Improve plant vigor, avoid moisture stress, avoid wounding, and prune affected branches 6 to 8 inches below diseased tissue during dry weather with sterilized pruning tools. Avoid planting dogwoods in shady or crowded areas.

Chemical Control

Chlorothalonil provides fair control of the leaf spot phase of this disease only. Apply the fungicide according to label recommendations.

Nectria Cankers

Nectria canker on improperly pruned maple. Note orange spore clusters in affected bark.
(Courtesy B.B. Clarke)

Nectria cankers are common on a wide variety of shade trees and other woody ornamentals. *Nectria* is an opportunistic fungus that infects twigs, branches, and trunks through wounds and at the base of dead branches. This fungus can cause both annual and perennial cankers. Annual cankers are common on twigs and branches injured by freezing, drought stress, mechanical injuries, or other diseases. As cankers enlarge, twigs are girdled and killed in a single season. A cut made into the wood with a pocket knife reveals a sharp transition between white, healthy wood and brown, infected wood. Perennial cankers enlarge yearly, encircling the branches and eventually killing tissue beyond the canker. With each successive year of infection, a "bulls-eye" pattern may develop. **Nectria canker** can be identified by the bright orange fruiting bodies that form in the center of the cankers.

Proper Management

Improve plant vigor and avoid moisture stress, wounding, winter injury, and mechanical injury. Prune affected branches (when practical) 6 to 8 inches below infected tissue, during dry weather, with sterilized pruning tools.

Chemical Control

None recommended.

Verticillium Wilt

Dark streaking in vascular tissue is diagnostic of Verticillium wilt.
(Courtesy S. Davis)

Verticillium wilt is a disease of many species of shade trees in the landscape, particularly maple. The fungus *Verticillium* lives in the soil and penetrates small roots. Spores of the fungus are carried up to developing tissue in the canopy via water-conducting vessels in the wood. The vessels become clogged and affected branches wilt and die. **Verticillium wilt** is part of a syndrome known as **maple decline**, where environmental stress, attack by insects, and poor growth contribute to an over-all decline in older trees.

Proper Management

Improve plant vigor and avoid moisture stress.

Chemical Control

None recommended.

Diplodia (or Sphaeropsis) Shoot Blight and Canker

Symptoms of Diplodia shoot blight of pine usually begin on the lower branches.
(Courtesy J.L. Peterson)

Diplodia (or Sphaeropsis) shoot blight and canker affects 2- and 3-needle pines and is most devastating on Austrian, mugo, and Scots pines. The fungus *Sphaeropsis* infects and kills developing needles, resulting in dead candles that are much shorter than healthy ones. Sunken cankers may form on branches and stems, killing the tissue beyond the cankers. The lower branches of pines are usually affected first. Tiny, black, spore-producing structures called "fruiting bodies" can be seen with the aid of a hand lens at the base of dead needles and on cones. Spores are released from these fruiting bodies in cool, rainy weather and are transmitted to susceptible tissue. This disease is more severe on trees that are stressed. Japanese black pine is tolerant of this disease and offers an attractive alternative where **Diplodia shoot blight** has been a problem in the past.

Proper Management

Improve plant vigor and prune affected branches 6 to 8 inches below diseased tissue during dry weather with sterilized pruning tools. Remove as much plant debris as possible and use tolerant species.

Chemical Control

Apply Tersan 1991 WP or Cleary 3336 WP according to label recommendations.

Pachysandra Leaf and Stem Blight

Pachysandra leaves infected with *Volutella.* Note the target-shaped lesions. (Courtesy S. Davis)

Pachysandra leaf and stem blight is caused by the fungus *Volutella.* Pachysandra is most susceptible to this disease when it has suffered from winter injury, moisture or heat stress, mechanical injury, or has a problem with scale insects. Large leaf spots, which have a "bulls-eye" pattern, appear on leaves. Cankers form on petioles and stems that produce characteristic pink-colored fruiting bodies within several weeks in wet weather. Occasional thinning and removal of leaf litter reduces humidity and helps to keep disease severity to a minimum.

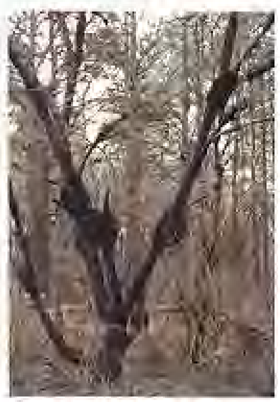
Proper Management

Improve plant vigor and avoid moisture stress, winter injury, and mechanical injury. Remove leaf litter to reduce humidity and control scale insects, if present.

Chemical Control

To control leaf and stem blight, apply chlorothalonil or mancozeb plus thiophanate-methyl according to label recommendations. If scale insects are present, apply 2% dormant oil, acephate, malathion, diazinon, or dimethoate according to label recommendations.

Black Knot of Plum and Cherry



Black knot on wild cherry.

Black knot of plum and cherry. This disease is widespread on garden plums, sweet and sour cherries, and chokecherry. Knot-like swellings, which are black, roughened, and spindle-shaped, form on twigs and branches. These knots, which live for many years, continually increase in size. Spores of the causal fungus *Apiosporium* are released from the knots during rainy weather in the spring and infect green, susceptible tissue. These new swellings will grow for two seasons before producing spores of their own.

Proper Management

Improve plant vigor and prune infected limbs 6 to 8 inches below all visible knots before new shoots develop. Remove and destroy the clippings.

Chemical Control

None recommended.

Ovulinia Petal Blight

Dried flowers infected by Ovulinia petal blight cling to rhododendron foliage. Note the dark, round sclerotia (resting structures) imbedded in the dried flower. (Courtesy J. L. Peterson)

Ovulinia petal blight, one of the most common diseases of rhododendrons and azaleas, affects only the flowers. Small, water-soaked spots appear on infected petals. These spots rapidly enlarge until the flower becomes slimy, limp, and turns prematurely brown. Entire trusses may become diseased almost simultaneously. Most infected petals adhere to the plant but some may fall to the ground. Six to eight weeks following infection, small, black sclerotia (resting structures) develop on infected petals. These sclerotia germinate in the spring and produce fruiting structures called apothecia. Spores are forcibly ejected from the apothecia, striking blossoms close to the ground. Wet weather at flowering time enhances disease development.

Proper Management

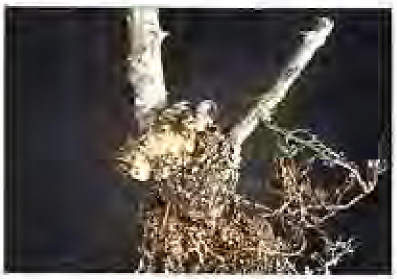
Remove dead trusses and fallen petals as soon after bloom as possible to reduce disease spread. Maintain plant vigor.

Chemical Control

Mist chlorothalonil, triadimefon, thiophanate-methyl, or mancozeb plus thiophanate-methyl onto plants from the time flowers begin to show color until flowering has ceased at intervals stated in label recommendations.

Specific Spring-Time Diseases Caused By Bacteria

Crown Gall



Crown gall on rose. (Courtesy B. B. Clarke)

Crown gall is caused by the soil-borne bacterium *Agrobacterium tumefaciens.* More than 600 species of plants are susceptible to **crown gall**. This bacterium enters plant roots and stems through wounds near the soil line. Infection by the bacterium causes tissue at the crown area to grow rapidly, resulting in the formation of galls. These galls consist chiefly of host tissue. Following the initial infection, galls soon form on other parts of the plant. To prevent **crown gall**, avoid wounding during transplanting and cultivation. Once the disease is present in a plant, pruning individual galls will not prevent galls from forming on other parts of the plant.

Proper Management

Improve plant vigor, avoid wounding, and remove entire plants when galls are observed. Utilize resistant plants in spots where diseased plants have been observed previously.

Chemical Control

None recommended.

Fire Blight

Fire blight on mountain ash. (Courtesy B. B. Clarke)

Fire blight, caused by the bacterium *Erwinia amylovora*, can occur on many rosaceous plants, including crabapple, cotoneaster, hawthorn, mountain ash, pyracantha, and pear. In the spring, bacteria ooze from existing cankers on infected plants. The bacteria are carried to healthy blossoms and branches by insects that are attracted to the ooze. The bacteria are also spread by splashing rain. Twigs and branches infected with the **fire blight** bacterium die rapidly and appear scorched. Cankers form at the base of infected branches.

Proper Management

Improve plant vigor, avoid heavy spring fertilization, and use resistant cultivars. Prune affected branches during dry weather. Remove branches 6 to 8 inches below diseased tissue using sterilized pruning tools.

Chemical Control

Copper or streptomycin according to label recommendations.

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Plant Disease Control

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Needlecasts and Common Needle Diseases of Christmas Trees

Ann Brooks Gould, Ph. D.
Extension Specialist in Plant Pathology

Christmas trees in New Jersey plantations are susceptible to needle diseases, especially when environmental conditions are conducive for disease development. There are three types of needle diseases—**needle rusts, needlecasts**, and **needle blights**—that are caused by living organisms. These living organisms are called pathogens. Pathogens obtain nutrients by parasitizing living tissue.

Needle Rusts

Needle rusts are unique because they are caused by fungi that often require more than one host plant species to survive. These fungi initially spend part of their life cycle on one host and the remainder on another. Needle rusts on conifers are characterized by yellow, orange, red-brown, or white pustules that are filled with powdery, brightly-colored spores. There are a variety of rust fungi that affect Christmas trees in New Jersey. Two of the more common needle rusts are **Spruce Needle Rust** and **Pine Needle Rust**.

Spruce Needle Rust

Spruce needle rust, caused by the fungus *Chrysomyxa*, affects white, black, and Colorado blue spruce. On spruce, white blisters containing yellow spores appear on the current season's needles in mid-summer. These spores are blown by the wind in the summer to the alternate hosts, Labrador tea and leatherleaf. The fungus colonizes the alternate hosts and overwinters in leaf tissue. In the spring, spores are produced in pustules on lower leaf surfaces. These spores are carried by the wind to developing spruce needles. By mid-summer, symptoms are again present on spruce. Infected needles are shed by the end of the summer, and severely infected trees may lose up to 75 percent of their new needles. Spruce needle rust rarely kills spruce trees, but repeated infections may limit growth and render trees unfit for sale.

Needle Rust. (Courtesy J.L. Peterson)

To prevent spruce needle rust, avoid planting susceptible spruce trees near swamps that harbor leatherleaf or Labrador tea. Norway or Black Hills spruce are fairly resistant to this disease. Fungicides are ineffective and are, therefore, not recommended.

Pine Needle Rust

Pine needle rust, caused by the fungus *Coleosporium*, can be a problem on red and Scotch pines. The alternate hosts of this fungus are goldenrod and aster. *Coleosporium* overwinters in pine needles. In the early summer, orange blisters erupt from needles on the lower branches. Spores from these pustules are carried by the wind to goldenrod and

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aster. By late summer, orange, cushion-like pustules appear on the lower leaf surfaces of the herbaceous host. Spores produced in these pustules are blown back to pine. New blisters are formed on the pine needles the following summer.

Severe infection can disfigure or kill young trees. For optimal control of pine needle rust, the life cycle of the fungus must be disrupted by removing the alternate host. Mow goldenrods and asters before August, or apply a registered herbicide. Triadimefon (Bayleton) is registered for control of this disease and is effective when used according to label directions.

Needlecasts

Most needlecast diseases are caused by fungi that infect young developing shoots. Generally, symptoms do not appear on infected needles until the winter or spring following infection. At that time, tan or reddish-brown spots appear. Structures of the fungus that produce spores, called fruiting bodies, develop in these discolored regions. The spores are carried by wind or splashing water to susceptible tissue. Three common needlecast diseases in New Jersey are **Rhabdocline**, **Lophodermium**, and **Cyclaneusma** needlecasts.

Rhabdocline Needlecast

Rhabdocline needlecast affects previous year's growth of Douglas-fir. (Courtesy B.B. Clarke)

Rhabdocline needlecast, caused by the fungus *Rhabdocline pseudotsugae*, affects only Douglas fir. Irregularly shaped, reddish-brown spots surrounded by green tissue appear on the previous year's needles by early spring. Orange fruiting bodies containing spores are produced on the lower surface of affected tissue at budbreak. These spores are splashed to the current season's needles in late April and early May. By early summer, infected needles are shed (or cast). Symptoms on newly infected needles do not appear until the following winter. The disease is more severe on lower branches.

The development of Rhabdocline needlecast is favored by abundant moisture and cool temperatures (53° to 59°F). Trees on north-facing slopes or in low-lying areas with poor air drainage are more likely to become infected. Close spacing and poor weed control contribute to conditions of high humidity that are favorable for disease development.

To identify Rhabdocline needlecast, look for reddish-brown spots on last year's needles in late winter and early spring. For optimal control, increase spacing, control weeds, and apply chlorothalonil at 10% budbreak. Repeat fungicide applications one and three weeks after the first spray. Apply the fungicide a fourth time if cool spring weather persists.

Lophodermium Needlecast

***Lophodermium* fruiting body on pine.** (Courtesy P. R. Bachi)

Lophodermium needlecast, caused by the fungus *Lophodermium seditiosum*, is most severe on 2- and 3-needle pines, particularly Scotch and Austrian pines. In the spring, brown spots with yellow margins appear on needles that had been infected the previous growing season. By mid-summer, these needles turn completely yellow, then brown, and finally are cast from the tree. "Football-shaped" fruiting bodies are produced in the brown needles just before or after they drop. These fruiting bodies are easy to spot even without a hand lens because of their characteristic shape. From August through October, windblown spores infect the current season's growth on branches close to the ground. Symptoms on newly infected needles do not appear until the following spring. Although the disease appears first on lower branches, in severe cases the entire tree may be affected. This disease can be troublesome on nursery seedlings as well as on mature trees.

Like Rhabdocline needlecast, the development of Lophodermium needlecast is enhanced by abundant moisture, high humidity, poor air drainage, and poor weed control. For optimal control in the nursery, avoid keeping needles wet for prolonged periods by irrigating early in the morning. Application of a registered fungicide such as chlorothalonil should commence in early June and may be repeated until late fall at 6- to 8-week intervals.

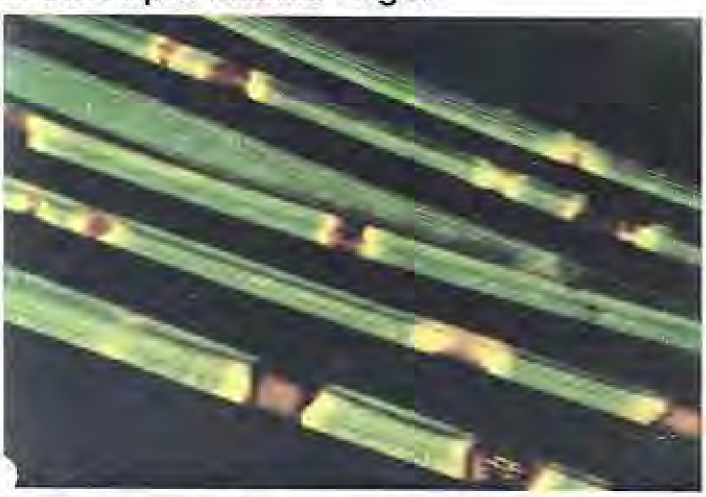
Cyclaneusma Needlecast

Scotch pine needle infected with *Cyclaneusma*. (Courtesy J. L. Peterson)

Cyclaneusma needlecast, like Lophodermium needlecast, also occurs on 2- and 3-needle pines. Scotch and Austrian pines are particularly susceptible. This needlecast disease is caused by the fungus *Cyclaneusma minus*. Dark-brown, horizontal bands appear in the fall on affected needles. White, waxy fruiting bodies develop within the bands and produce spores in the spring. These spores are carried by the wind to infect needles of all ages. Most trees become infected between April and June, although infection is possible through December. Symptoms do not appear until the following growing season, and severely infected needles hang on the tree for several months before dropping.

To identify Cyclaneusma needlecast, look for yellow needles with dark-brown bands on 2- and 3-year old needles. Needles anywhere on the tree may be affected. To control this disease, apply a registered fungicide, such as chlorothalonil, prior to budbreak. Repeat the application at 6- to 8-week intervals until late fall.

Brown Spot Needle Blight



Brown spot needle blight. (Courtesy of B. B. Clarke)

The most common needle blight disease, **brown spot needle blight**, is caused by the fungus *Scirrhia acicola*. This disease, although a problem on many pines, is most severe on Scotch pine.

In the spring, black fruiting bodies develop in dead needle tissue. Spores are splashed from these structures to developing needles. Symptoms progress from small spots to larger, reddish-brown, resin-soaked lesions with yellow margins. These lesions girdle the needle, causing the tip of the needle to die and the base of the needle to remain green. The fungus overwinters in dead needle tissue. Needles on lower branches are more likely to be affected. Infected needles are shed the following summer once the fungus has produced spores.

The development of brown spot needle blight is favored by prolonged periods of wet weather, particularly during June and July. Close spacing and poor air circulation increase disease severity. Since individual trees differ in susceptibility to needle blight, utilize seedlings derived from resistant seed. Avoid cultivating when the foliage is wet to limit disease spread. Chlorothalonil may be applied in the spring when new growth is 1/2 to 2 inches in length, and repeated at 3- to 4-week intervals until conditions are no longer favorable for disease development.

Some diseases of Christmas trees are not caused by living organisms. These diseases, often called **abiotic** or **non-infectious** diseases, are caused by non-living agents such as air pollutants, temperature and moisture extremes, nutritional toxicities and deficiencies, exposure to salt, and other site-related stresses.

Fluoride Pollution

Fluoride injury on Austrian pine. (Courtesy of E. Brennan)

Fluoride is a natural component of soil, rocks, and various minerals. Toxic quantities of fluoride may be released into the atmosphere when materials containing fluorides are heated or treated with acid during industrial processing.

The primary sources of fluoride pollution in New Jersey are glass and brick manufacturing plants.

In conifers, fluoride injury is most evident on first-year needles. The tips of these needles first turn reddish-brown from the tip toward the needle base. With continued exposure to fluoride, affected needles may be cast from the tree. Older needles rarely exhibit visual symptoms. Conifers that are particularly sensitive to fluoride include Douglas fir, mugo pine, Scotch pine, white pine, and blue spruce. Gladiolus, one of the most susceptible plants to fluoride, is often used as an "indicator plant" for fluoride pollution.

Injury due to fluoride commonly results from a gradual accumulation of the pollutant in plant tissue over time. Unlike deciduous species which shed affected leaves, toxic levels of fluoride can accumulate in conifers because affected needles may persist for several years. Ultimately, the type of injury that results from fluoride depends on the dose (concentration x time), the time of year, the plant species involved, the genetic make-up of the vegetation, and the age of the foliage affected.

The symptoms produced on conifer needles in response to fluoride injury may be easily confused with symptoms caused by other pollutants and environmental stresses. To diagnose fluoride injury, compare the affected vegetation with other vegetation in the area. Generally, more than one plant species will be affected if fluoride is involved. Visually examine the affected plants and compare the symptoms with those described in the literature. Since fluoride problems typically occur within a few miles of the source, verify the existence of a pollution source in the area. Symptoms due to fluoride pollution are more prominent and uniformly distributed on the side of the plant facing the pollution source.

Drought stress on white pine. (Courtesy of B. B. Clarke)

Drought Stress

Drought stress occurs when the foliage loses water at a faster rate than the roots can absorb water from the soil. Symptoms of early drought stress in conifers appear at the top of the tree as a wilting and drooping of the needles. If drought stress persists, needles may become discolored and permanently bent. In conifers, the oldest needles may turn yellow and drop prematurely. Drought stress may also predispose affected trees to attack by other pathogens and insects.

Yellowing of needles on Japanese black pine due to soil compaction and poor drainage. (Courtesy of B. B. Clarke)

Site-related Stresses

Poor site conditions, such as compaction, poor drainage, and low fertility, are common problems associated with Christmas tree production. Symptoms on trees affected by poor site conditions often include needle yellowing, stunting, and premature needle drop. Although Christmas trees can be grown on marginal sites, optimum growth and improved pest and stress tolerance can only be attained when site-related stresses are corrected or avoided.

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Plant Disease Control

RUTGERS COOPERATIVE EXTENSION · NEW JERSEY AGRICULTURAL EXPERIMENT STATION

An Integrated Approach to Necrotic Ring Spot Control in Turf

Bruce B. Clarke, Ph. D.
Extension Specialist in Turf Pathology

Karen Kackley-Dutt, Ph. D.
Coordinator, Plant Diagnostic Laboratory

Necrotic ring spot is a newly described disease of cool-season turfgrasses that has been observed with increasing frequency in the northeast, upper midwest, and Pacific northwest regions of the United States. Prior to 1983, it was considered a component of the Fusarium blight complex. Necrotic ring spot is a serious disease of Kentucky bluegrass and has also been reported on bentgrass, fine fescue, annual bluegrass, and rough bluegrass.

Symptoms

Circular patches of infected turf may develop whenever periods of cool, wet weather occur. On Kentucky bluegrass, patches first appear as small, light green spots 2 to 4 inches in diameter. In some cases, patch diameters may exceed 3 feet, but they usually remain in the 4- to 12- inch range. As turf succumbs to infection, the leaves turn reddish-brown to bronze and then fade to a light straw color. Under conditions of thick thatch, all of the plants in a patch may die, resulting in a sunken or crater-like depression. Frequently, however, plants survive or recolonize infection centers and the patch takes on a ring or frog-eye appearance. Symptoms may also appear as diffuse patterns of yellow or brown-colored turf that coalesce into larger blighted areas.

Necrotic ring spot is often confused with yellow patch and pink snow mold since they exhibit similar foliar symptoms and occur in the fall and spring. Unlike the latter two diseases whose symptoms usually subside in late spring, necrotic ring spot can occur throughout the growing season and is characterized by the presence of dark brown fungal strands (hyphae) on dying roots, rhizomes, and crowns. In the later stages of infection, black fruiting bodies (pseudothecia) may occasionally be found on these tissues in the field.

Causal Agent

Leptosphaeria korrae J. C. Walker & A. M. Sm., the causal agent of necrotic ring spot, was formerly named *Ophiobolus herpotrichus* (Fr.) Sacc. The fungus forms brown, septate, runner hyphae on infected turfgrass roots and crowns. Dark brown, flattened resting structures (sclerotia) and black, flask-shaped fruiting bodies (pseudothecia) later develop on infected plant parts.

Disease Cycle

The causal agent is believed to survive unfavorable periods as sclerotia or hyphae in plant debris. Although little is known about the development of *L. korrae* in the soil, it attains maximum growth in the laboratory at 68° to 82°F and is inhibited at temperatures above 86°F or below 50°F. Symptoms can occur throughout the growing season during cool, wet weather, but generally appear in late spring and early autumn. Patches often fade with the advent of warmer temperatures in the summer, but may reappear in response to heat and drought stress. Infection centers develop again in early autumn and may persist through the winter and early spring. Recovery is slow and severely infected plants are easily pulled up due to the extensive root, crown, and rhizome rot. Infected sod and mechanical equipment may spread the disease.

Epidemiology

Conditions that favor necrotic ring spot are similar in many respects to those that favor take-all patch. The growth of the fungus is stimulated by cool, wet weather; however, heat and drought stress have been shown to intensify symptom expression. Since the fungus is more tolerant of soil moisture extremes than Kentucky bluegrass, drought stress may play a more important role in the development of necrotic ring spot than in take-all or

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summer patch. Necrotic ring spot can occur over a wide range in soil pH (5.0 to 8.0) and is intensified on compacted soils. The disease is most prevalent on 2- to 4-year-old lawns that were established with sod, although seeded areas and young turf can also sustain damage.

Control

Since necrotic ring spot is a relatively new disease, information regarding its control is limited. Most researchers agree that keeping infected turf adequately fertilized and well watered to avoid drought stress will promote recovery. The benefits attributed to specific nitrogen sources or the application of sulfur to modify soil pH have not been consistently demonstrated. Overseeding infected turf with perennial ryegrass, tall fescue, or more resistant cultivars of Kentucky bluegrass will reduce disease severity.

Several currently registered fungicides have proven effective in reducing the incidence and severity of necrotic ring spot when applied on a preventive basis in early- to mid-spring. Systemic fungicides, such as fenarimol (Rubigan), benomyl (Tersan 1991), or thiophanate-methyl (Fungo, Topsin M, or Cleary 3336) applied at high label rates as a spray or drench, have been most effective. For best results, apply fungicides in early April and then repeat in early May. Control is enhanced when products are applied in at least 4 gallons of water per 1000 square feet. Contact fungicides have not provided adequate control in most laboratory and field tests.

Severe outbreak of necrotic ring spot on a Kentucky bluegrass lawn.

Necrotic ring spot on an annual bluegrass putting green.

Crater-like depressions in a Kentucky bluegrass lawn, caused by *Leptosphaeria korrae*.

Fruiting body of *Leptosphaeria korrae* on a Kentucky bluegrass stem.

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Plant Disease Control

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An Integrated Approach to Summer Patch Control in Turfgrass

Bruce B. Clarke, Ph. D.
Extension Specialist in Plant Pathology

Karen Kackley-Dutt, Ph. D.
Coordinator, Plant Diagnostic Laboratory

Summer patch was first recognized as a disease of cool-season turfgrasses in 1984. Prior to that time, it was an unidentified component of Fusarium blight. Summer patch has been reported in North America on fine fescue and Kentucky bluegrass. The causal agent has also been isolated on occasion from bentgrass and perennial ryegrass. The disease generally occurs on turf that has been established for more than two years.

Symptoms

On Kentucky bluegrass, symptoms first appear in early sum-summer as small, circular patches of wilted turf 1.5 to 3.5 inches in diameter. Patches may enlarge to more than 24 inches, but generally remain in the 2- to-12- inch range. Affected leaves rapidly fade from a grayish-green to a light straw color during sustained hot weather (daytime highs 82° to 95°F and nighttime temperatures exceeding 68°F). Irregular patches, rings, frog-eye, and crescent patterns may also develop and coalesce into large areas of blighted turf.

In mixed stands of bentgrass and bluegrass maintained under putting green conditions, patches are circular and range from 1 to 12 inches in diameter. As annual bluegrass yellows and declines, bentgrass species frequently recolonize patch centers. On fairways and lawns, rings or frog-eye patches may not develop. In such cases, symptoms may appear as diffuse patterns of yellowed or straw-colored turf that are easily confused with heat stress, insect damage, or other diseases. Infected roots, rhizomes, and crowns turn brown as they are killed. Examination of these tissues typically reveals a network of sparse, dark brown to black, fungal strands (hyphae) from which clear penetration hyphae invade the underlying vascular tissue. In the latter stages of infection, vascular discoloration and cortical rot are extensive. No fruiting structures have been observed under field conditions.

Causal Agent

Magnaporthe poae Landschoot & Jackson, the causal agent of summer patch, is a newly described fungus whose asexual stage had previously been misidentified as *Phialophora graminicola* (Deacon) J. Walker. The fungus forms dark brown to black, septate, runner hyphae on roots, crowns, and rhizomes of turfgrass hosts. Sexual fruiting bodies, which have only been observed in culture, are black, spherical, and have long cylindrical necks.

Disease Cycle

The pathogen is believed to survive the winter months as hyphae in previously colonized plant debris and in perennial host tissue. Colonization and suppression of root growth has been shown to occur between 70° and 95°F under controlled environmental conditions, with optimum disease development at 82°F. In the field, infection commences in late spring when soil temperatures stabilize between 65° and 68°F. The fungus moves from plant-to-plant by growing along roots and rhizomes. Symptoms develop during hot (86° to 95°F), rainy weather or when high temperatures follow periods of heavy rainfall. Patches may continue to expand through the summer and early autumn and are often still evident the following growing season. The summer patch fungus may be spread by aerification and dethatching equipment as well as by the transport of infected sod.

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Epidemiology

Summer patch is most severe during hot, wet years and on poorly drained, compacted sites. Although heat stress plays an important role in disease development, drought stress is usually not a predisposing factor. Under ideal conditions, the causal agent can spread along roots, crowns, and stem tissue at a rate of up to 1.5 inches per week. Symptom expression has been shown to increase with the use of nitrate-based fertilizers, arsenate herbicides, and many commonly used contact fungicides. The disease is frequently stimulated when turfgrass is maintained under conditions of low mowing height, high pH (> 6.0), compaction, and frequent, light irrigation.

Control

Because summer patch is a root disease, cultural practices that alleviate stress and promote root development will reduce disease severity. Since low mowing enhances symptom expression, avoid mowing turf below recommended heights, particularly during periods of heat stress. In the northeast, symptoms are less apparent when lawns are maintained at a height of 2 to 3 inches and golf greens and fairways are cut at or above 5/32 and 3/8 inches, respectively. Fertilize turf with ammonium sulfate or a slow-release nitrogen source such as sulfur-coated urea. Irrigate deeply and as infrequently as possible without inducing drought stress. Aerification, improving drainage, reducing compaction, and syringing to reduce heat stress are other practices that will aid in the control of this disease.

Overseeding affected areas with bentgrass, perennial ryegrass, tall fescue, or resistant cultivars of Kentucky bluegrass represent one of the most cost-effective means of controlling summer patch. Use mixtures or blends of resistant turf cultivars or species for best results. Conversion of golf areas from bluegrass to bentgrass will also reduce disease incidence.

Fungicides are available that can effectively control summer patch. Applications should commence on a preventative basis in late spring or early summer when soil temperatures stabilize between 64° and 68°F. Systemic fungicides, such as fenarimol (Rubigan), propiconazole (Banner), triadimefon (Bayleton), and the benzimidazoles (i.e., Tersan 1991 and Cleary 3336), have proven to be most effective but must be applied at high label rates. Repeat two to three times at 21-28 day intervals for best results. Efficacy is enhanced when products are applied in at least 4 gallons of water per 1000 square feet. The continued use of contact fungicides at high label rates may stimulate symptom severity.

Summer patch symptoms on an annual bluegrass golf fairway.

Growth of perennial ryegrass within a patch of Kentucky bluegrass killed by *Magnaporthe poae*.

Pigmented hyphae of *Magnaporthe poae* on the surface of a turfgrass root. (Courtesy P. J. Landschoot)

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DIAGNOSTIC SERVICES:

- Disease and insect pest diagnoses.
- Plant and weed identification.
- Insect identification.
- Nematode detection.
- Screening for turfgrass endophytes
- Screening for fungal resistance to benzimidazole fungicides.
- Other services available by contract.

WHERE TO SEND SAMPLES:

Sample submission forms are available from County Extension Offices throughout the State of New Jersey or by FAX (908-932-1270). Send samples with the appropriate submission form and payment to:

Via U.S. Postal Service:

Plant Diagnostic Laboratory
Rutgers Cooperative Extension
P. O. Box 550
Milltown, NJ 08850-0550

Via Other Delivery Services:

Plant Diagnostic Laboratory
Rutgers Cooperative Extension
Building 6020, Dudley Road
Cook College
New Brunswick, NJ 08903

FEES:

All in-state samples (except fine turf)	**$20**
Fine turf samples	**$50**
All out-of-state samples	**$75**

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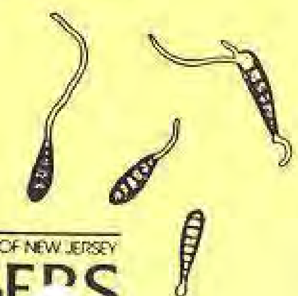
PLANT DIAGNOSTIC LABORATORY

AND

NEMATODE DETECTION SERVICE



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PLANT DIAGNOSTIC LABORATORY

MISSION:

The mission of the Plant Diagnostic Laboratory is to provide the citizens of New Jersey with accurate and timely diagnoses of plant problems. These goals are achieved in cooperation with New Jersey Experiment Station/Cook College Extension personnel and research faculty.

HISTORY:

The Plant Diagnostic Laboratory is a diagnostic service available to the residents of the State of New Jersey. The laboratory was established in 1991 on the Cook College campus of Rutgers University. The lab is fully operational, and there is a fee for its services.

STAFF and COOPERATORS:

The Plant Diagnostic Laboratory is staffed with two full-time diagnosticians who are trained in all aspects of plant health. Seasonal employees and students assist in the laboratory. The Plant Diagnostic Laboratory staff works in close cooperation with Rutgers Cooperative Extension specialists, county faculty, and other university personnel to provide accurate diagnoses and up-to-date control recommendations.

HOW TO SUBMIT A SAMPLE:

1) Sample submission forms may be obtained at your local county Rutgers Cooperative Extension office. Forms may also be requested by FAX (908-932-1270). There are four different forms. The green form is for plant identification. The yellow form is for golf course and landscape turf. The brown form is for home grounds and landscape samples, and the pink form is for commercial growers.

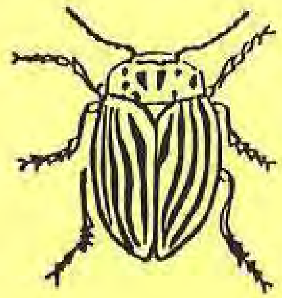
2) Completely fill out the sample submission form.

3) Collect the appropriate samples, carefully following all directions found on the back of the sample submission form.

4) Properly package the sample, including the submission form and appropriate payment.

5) Mail the sample to the appropriate address.

6) The laboratory will respond with the diagnosis by mail in a timely manner.



TIPS FOR SAMPLE SUBMISSION:

1) Carefully follow all directions on the sample submission form.

2) Send samples early in the week. Samples mailed on a Friday will sit in the post office over the weekend.

3) Collect samples before applying pesticides.

4) Avoid packaging samples in plastic unless they are to be shipped overnight. Keep the sample and all paperwork separate to keep the paperwork dry.

5) Detailed information on the submission form is essential for an accurate diagnosis. Take the time to completely fill out the submission form.

Rutgers University

Diagnostic Lab Provides Answers for Turf Growers

By MICHELLE DOMANGUE

THE YEAR-OLD Plant Diagnostic Laboratory at Rutgers University offers diagnosis of all sorts of plant problems. But special expertise in turfgrass is "our unique legacy," said Dr. Karen Kackley, lab coordinator.

"Because of his national reputation, many golf course superintendents around the country submit samples to Dr. Bruce Clarke (New Jersey Extension turf pathologist)," Kackley said. Now that the new lab is up and running at full speed, Clarke forwards the samples to Kackley.

TURFGRASS PULLED by a cup cutter makes an ideal sample, says Dr. Karen Kackley, coordinator of the Plant Diagnostic Laboratory at Rutgers University.

Kackley herself holds a doctorate in turfgrass pathology from the University of Maryland. Before taking on the post of lab coordinator, she had worked with plant protection compounds for turf and ornamentals in Monsanto's product development department.

Her assistant, Richard Buckley, earned a master's degree in turfgrass pathology from Rutgers. Together, they do their best to identify sources of problems in the plant samples they receive.

During the lab's first six months, nearly one in five samples came from outside the state—some from as far afield as California.

The lab is a full-service agricultural diagnostic facility, equipped to deal with ornamentals and field-crop specimens, along with turf.

Turfgrass accounted for nearly half of all samples received from June to December 1991.

Once the samples arrive, "we look for pathogens—fungus, bacteria; we'll even make a stab at viruses," Kackley said. "We look for insect pests in samples. We can't test for pesticide residue. But we may be able to say, 'It looks like a chemical burn.' We can suggest cultural problems."

What may be most important of all, though, "I can tell you what it isn't."

From the samples sent in during 1991, she found the most serious disease problems in turf were brown patch, pythium blight, anthracnose and summer patch. Often, they appeared in combination.

Accurately diagnosing diseases can reduce chemical use, since many of the chemical treatments are very specific, Kackley noted. If the diagnosis is wrong, the chemical treatment may be wasted.

But the person sending in the sample can help insure the diagnosis is accurate.

"The quality of diagnosis depends on the quality of the sample submitted and the quality of information. For instance, someone could send in a branch of a tree, but the problem might be present only in the roots. With turf, it's nice because we can have the whole plant."

RICHARD BUCKLEY is diagnostician/program associate at the Plant Diagnostic Laboratory. He earned a master's degree in turfgrass pathology from Rutgers and has worked in diagnostics, soil testing and field research.

Weather information is also important, she added.

A cup cutter plug makes an ideal turf sample.

Kackley shared some tips for taking samples:

- Select from the transition zone between healthy and affected turf.
- Collect several samples representing different stages of symptom development from each location to be analyzed.
- Take samples at least 5 inches long by 5 inches wide by 3 inches deep.
- Obtain samples right before shipping to insure freshness; dried turf is difficult to analyze.
- Place dry insects in a sturdy container stuffed with paper to prevent damage. Put soft-bodied insects in unbreakable containers filled with alcohol. Don't use tape to secure insects to paper.

Lab services aren't free, and the cost of analyzing a sample is higher for clients living outside New Jersey. The lab's goal is to become self-supporting within five years, Kackley said.

But money expended on lab analysis can be money well-spent.

She gave the example of one golf course superintendent who brought in 18 samples, one from each fairway.

Flaberghasted, she asked, "Do you realize how much this will cost?"

"Do you realize how much I spend on treatment?" he retorted.

The Rutgers lab welcomes inquiries by mail or fax. The address is: *Plant Diagnostic Laboratory, Rutgers Cooperative Extension, P.O. Box 550, Milltown, N.J. 08850-0550 (street address: Building 6020, Dudley Road, Cook College, New Brunswick, N.J. 08903). Fax: 908-932-1270.* □

CHANGE

Gerald T. Knight has been named vice president and chief financial officer of The Toro Company. □

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