Pest e-alerts

Entomology and Plant Pathology, Oklahoma State University
127 Noble Research Center, Stillwater, OK 74078
405.744.5527

Vol. 9, No. 12 http://entoplp.okstate.edu/Pddl/ **Apr 20, 2010**

Spring Dead Spot Season

Damon L. Smith, Extension Horticulture Pathologist
Nathan R. Walker, Turfgrass IPM Scientist

While many turfgrass managers, landscape maintenance professionals, and homeowners have been focusing on evaluating the severity of winterkill in bermudagrass stands in the past weeks, there have also been many questions regarding spring dead spot (SDS). Along with large brown areas that are void of live tillers as a result of winterkill, there are areas with the distinct sunken circular patches indicative of SDS. Spring dead spot is a common disease of bermudagrass. It is only occasionally a disease of buffalograss or zoysiagrass in Oklahoma. Spring dead spot can be found frequently each year in Oklahoma and primarily occurs in highly managed residential, commercial, and recreational bermudagrass lawns. Stands of bermudagrass that are seldom fertilized, irrigated, or receive herbicide treatments are less likely to develop severe levels of SDS. In Oklahoma, SDS is most noticeable in spring as bermudagrass breaks dormancy and into the early summer. New symptoms of SDS do not appear in growing stands of bermudagrass in mid-to-late summer or fall in Oklahoma. Spring dead spot is not a disease of newly established bermudagrass stands (1-2 years since establishment), but can be common in mature stands (3+ years since establishment).

Affected areas may range from inches to many yards in length or diameter. The turf in affected areas will be dead and tan-brown in color. Large circular, semi-circular, or arcs of dead turf will be apparent. Patches appear in the same areas each year. They will typically expand in size and often can reach several yards in diameter. Affected plant parts, such as rhizomes, crowns, roots or stolons are dark black and rotted when removed from the soil. As the patches expand

and more dead area develops, weed growth within the patches during spring and summer months is common and may require the use of pre- or post- emergent herbicides.

Fungi in the taxonomic group *Ophiosphaerella* are the principal causal agents of spring dead spot in Oklahoma. Infection of the turf begins when soil temperatures are mild (<70°F). In Oklahoma, infection of susceptible grasses begins in late September and will continue as long as soil temperatures are above 50° F. Fungal growth and plant infection resumes in early spring, may slow growth of plants, and will eventually subside once soil temperatures climb above 75°F. Turfgrass grown under high nitrogen fertility or that receive late-season applications of nitrogen to extend green color, are more prone to the development of SDS.

Balanced soil fertility programs in early summer will increase the speed of recovery and aid bermudagrass in outcompeting weed encroachment (see OCES fact sheet HLA-6420). Avoid late season applications of nitrogen fertilizers. It is generally recommended to not fertilize after September 15th in Oklahoma. For small lawns and when patches are identified before they are large, symptomatic areas can be dug and the soil and plant material removed in and around the dead area. Few effective fungicides are directly available for the homeowner to use for spring dead spot control. However, fungicides are available to commercial turfgrass maintenance operators for SDS control. Fungicides in the Fungicide Resistance Action Group (FRAC) 3 (e.g. demethylation inhibitors or DMIs) should be applied in the fall before soil temperatures drop to 70°F for chemical control to be effective. A second fungicide application 28-30 days later will result in better control than a single fall application. Some fungicides and their relative efficacy on spring dead spot, which have been evaluated in trials at Oklahoma State University, are included in Table 1. It is highly recommended that the affected areas are photographed or otherwise mapped in the spring and that fall fungicide applications are targeted to only those areas where the disease was present. Recent research has indicated a single spring application is generally not effective, while a single spring plus a single fall application is as effective as two fall applications. Research has documented that fungicides mostly provide suppression of disease symptoms and complete control is often not achievable without the incorporation of other cultural strategies described above.

If confirmation of SDS is desired, samples of turfgrass can be sent to the Oklahoma State University Plant Disease and Insect Diagnostic Laboratory (PDIDL). The sample should be taken from the edge of the patch after they become apparent. Samples should be collected so that half of the sample area includes the apparently dead portion of turf, and the remaining half includes living grass immediately adjacent to the patch. Circular plugs or squares of 3-4" in diameter or width are sufficient. The sample should be double-bagged in ziptop bags with no added moisture, placed in a box with padding, and mailed to the PDIDL. Be sure to include a completed sample form with your sample. Sample forms can be found at http://entoplp.okstate.edu/PDDL/turf.pdf. Samples can be submitted at your county extension office or directly to PDIDL. Please call the PDIDL at 405-744-9961 if you need additional information regarding sampling or testing. Any pertinent digital pictures should be sent to jen.olson@okstate.edu and damon.smith@okstate.edu.

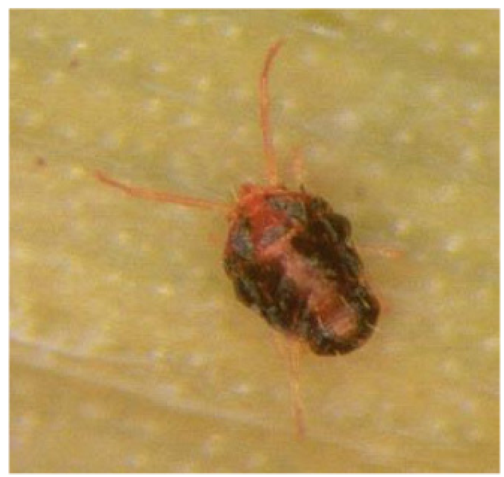
Table 1. Relative performance of select fungicides for controlling spring dead spot of bermudagrass.

Fungicide (Trade Name)	Performance [A]
Propiconazole (Banner Maxx)	4
Fenarimol (Rubigan)	4
Myclobutanil (Eagle)	4
Triticonazole (Trinity, Triton Flo)	2
Chlorothalonil (Daconil)	1
Azoxystrobin (Heritage)	1

[A] Performance is based on a scale of 1 - 4 where 1 = not effective, 2 = little to no control, 3 = some control, 4 = fairly good control.

Brown Wheat Mite Showing Up in Winter Wheat

Tom A. Royer, Extension Entomologist



Despite the Million Dollar Rain that we received this past weekend, Ron Hayes, Director of Farm Programming with Radio Oklahoma Network reported that there are still some small pockets of dry conditions in the northern parts of Woods and Alfalfa counties as well as parts of the Panhandle. This means that pests, such as brown wheat mite have not been knocked out in all locations. Roger Gribble, area agronomy specialist in Enid reported brown wheat mite infestations in western OK (Ellis and Harper counties). Producers need to remain alert to these problems so that their wheat is not suffering dual problems of dry growing conditions PLUS brown wheat mite.

This mite is small (about the size of this period.) with a metallic brown to black body and 4 pair of yellowish legs. The forelegs are distinctly longer that the other three pair. Brown wheat mites can complete a cycle in as little as 10-14 days. They will undergo up to 3 generations each year, but have probably already completed at least one or two by now. Numbers will likely decline if a hard, driving rain occurs. Spring populations begin to decline in mid-late April when females begin to lay “diapause” eggs.

Brown wheat mite causes problems in wheat that is stressed from lack of moisture. They feed by piercing plant cells in the leaf, which results in "stippling". As injury continues the plants become yellow, then dry out and die. These mites feed during the day, and the best time to scout for them is in mid-afternoon. They do not produce webbing and will quickly drop to the soil when disturbed. They are very susceptible to hard, driving rains, but until then they can cause yield loss when present in large numbers

Research suggests that a treatment threshold of 25-50 brown wheat mites per leaf in wheat that is 6-9 inches tall is economically warranted. An alternative estimation is "several hundred" per foot of row. However, make sure you check your field before deciding to spray your field, because that "Million Dollar Rain" may have already spread some wealth your way by killing off some of those pesky mites!

Check CR-7194, Management of Insect and Mite Pests in Small Grains for registered insecticides, application rates, and grazing/harvest waiting periods. It can be obtained from any County Extension Office, or found at the OSU Extra Website at http://pods.dasnr.okstate.edu/docushare/dsweb/Get/Document-2601/CR-7194web2008.pdf.

Those Aren't Aliens Hanging from Your Red Cedar Trees

Damon L. Smith, Extension Horticulture Pathologist

No those aren't alien life forms hanging from your cedar trees! It's cedar rust season! This is the time of year when rusts including cedar-apple rust caused by the fungus *Gymnosporangium juniperi-virginianae*, hawthorn rust caused by the fungus *Gymnosporangium globosum*, and quince rust caused by the fungus *Gymnosporangium clavipes*, cause the formation of telia (orange gelatinous structures) from galls on eastern red cedar trees (*Juniperus virginiana*). While these rusts are different species and cause slightly different symptoms, the life cycle is very similar. All require two hosts to complete their life cycles with the common host being *Juniperus* species. Alternate hosts include apple, hawthorn, crabapple, and quince among other rosaceous plants. On cedar hosts, typical symptoms include galls and twig dieback. In the spring, cedar–apple rust telia will typically appear as large masses of orange tendrils that erupt from, round growths (galls) after periods of rain and high humidity (Figs. 1 and 2). Galls formed by hawthorn rusts are generally smaller, more rounded and brownish in color. The gelatinous spore structures (telia) that develop after spring rains are brownish in color and have rounded tips (Fig. 3). Quince rust produces a spindle-shaped swelling on branches of *Juniperus* species. Gelatinous spore producing structures (telia) are orange in color, cushion-like, and appear to "ooze" from cracks in the gall (Fig. 4). Spores (basidiospores) that are released from telia serve as inoculum, which infect rosaceous plants. Damage to rosaceous plants includes lesions on leaves and young twigs and fruit. Under severe infection defoliation can result. In summer spores (aeciospores) form on rosaceous plants and will infect Juniperus plants. Galls form during the next year after infection and telia may not form until 22 months after infection.

Management strategies for the rusts described above include the use of resistant junipers, hawthorn, apples, crabapples, and quince varieties. In addition, spatial separation of rosaceous plants and junipers can help reduce the cycling of spores between alternate hosts so that the fungus cannot complete its life cycle. This latter strategy can be difficult to achieve as spores can move considerable distances (up to tens of miles). Fungicides should be used preventatively on desirable rosaceous plants. Fungicides should be applied to these plants prior to, or as soon as telia (orange, gelatinous structures on junipers) are observed. Multiple applications of fungicide may be necessary if wet weather persists. Consult the publication E-832 "Extension Agents' Handbook of Insect, Plant Disease and Weed Control" for fungicide recommendations.

Fig 1. Hundreds of cedar-apple rust galls on eastern red cedar trees.

Fig 2. Close-up of a cedar-apple rust gall with mature gelatinous spore tendrils.

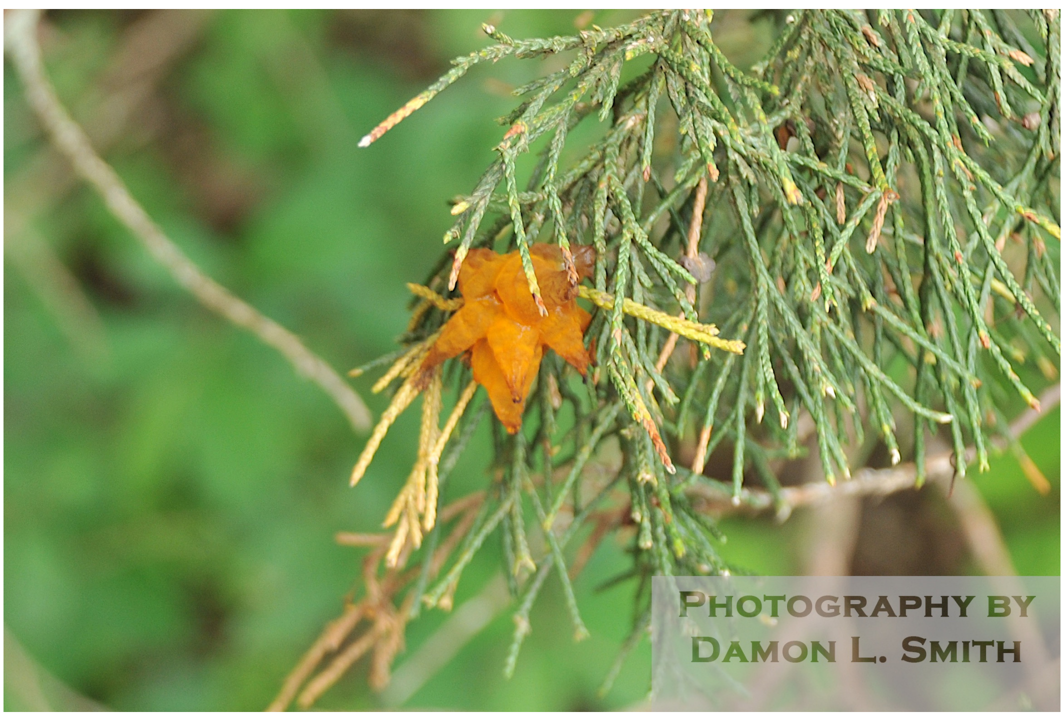


Fig 3. Close-up of a hawthorn rust gall with mature gelatinous spore tendrils on an eastern red cedar tree.

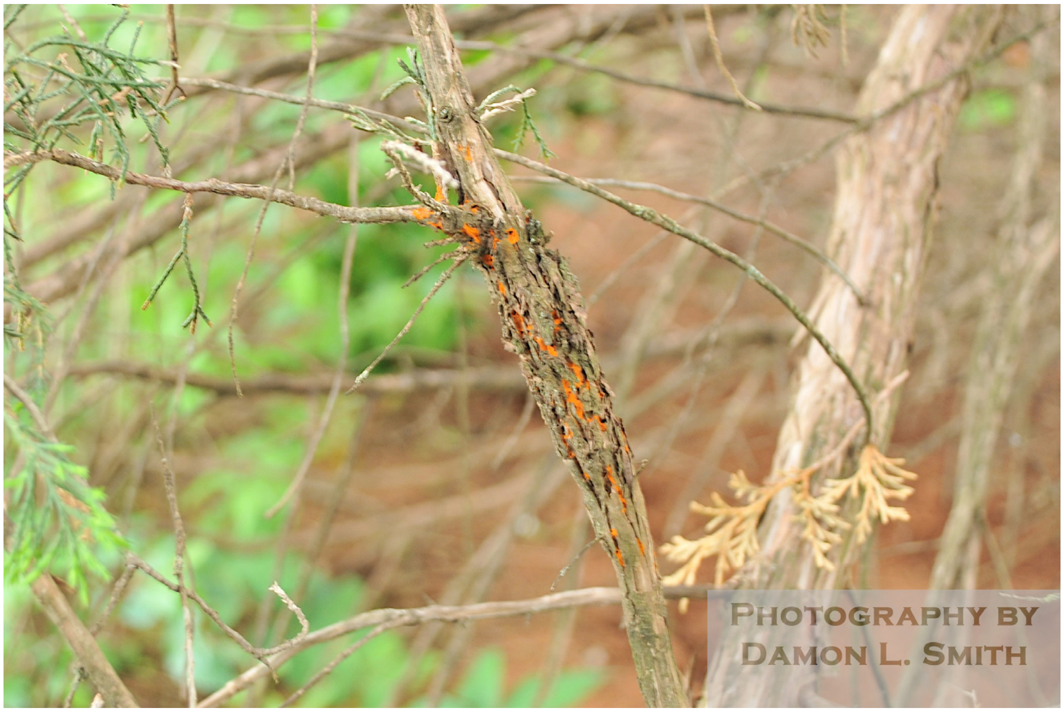


Fig 4. Close-up of spindle-shaped stem swellings (galls) caused by quince rust on an eastern red cedar tree.

Dr. Richard Grantham
Director, Plant Disease and Insect Diagnostic Laboratory

Issued in furtherance of Cooperative Extension work, acts of May 8 and June 30, 1914, in cooperation with the U.S. Department of Agriculture, Robert E. Whitson, Director of Oklahoma Cooperative Extension Service, Oklahoma State University, Stillwater, Oklahoma. This publication is printed and issued by Oklahoma State University as authorized by the Vice President, Dean, and Director of the Division of Agricultural Sciences and Natural.