Integrated Pest & Crop Management

Mizzou Plant Diagnostic Clinic

by Patti Hosack and Lee Miller

The Mizzou Plant Diagnostic Clinic (PDC) is open all year to receive plant samples that are affected by a disease or disorder. The PDC can also identify pesky weeds, plants of interest, mushrooms, and insects or spiders. Last year the Clinic processed 445 samples: 46% of these were agronomic crops, mostly soybeans and corn. A large number of soybean samples were diagnosed with Sudden Death Syndrome in 2014, as the cooler than normal weather promoted high disease pressure. In corn, Northern Corn Leaf Blight was most problematic and we had the first positive confirmation, via molecular testing, of Goss's Wilt. Goss's Wilt was diagnosed in several northern Missouri counties ranging from the Northwest corner of the state to St. Charles County. More information on Goss's Wilt can be found here: http://ipm.missouri.edu/IPCM/2014/8/Goss_s-Bacterial-Wilt-and-Leaf-Blight-of-Corn/.

Figure 1: Sudden Death Syndrome (Fusarium virguliforme) on soybean (Glycine max). Photo by Daren Mueller, Iowa State University, Bugwood.org.

Figure 2: Goss' bacterial wilt (Clavibacter michiganensis ssp. nebraskensis) on corn (Zea mays). Photo by Connie Tande. SDSU Plant Science Dept. Bugwood.org.

The PDC is open all year. It is encouraged that you get a diagnosis before applying pesticides, as this allows for selection of a product that will most effectively control the precise pest problem. The PDC is open for sample drop off, Monday through Friday from 9am to 4pm. A sample can also be mailed directly to the PDC or dropped off at your local extension office. If possible, take a picture of the sick plant before digging it up; if several plants are affected a picture of the entire planting is also encouraged. Pictures may be submitted in an email to plantclinic@missouri.edu, printed and submitted with the sample or supplied on a flash drive. As always, please include a Submission Form, which has been filled out as completely as possible, with the sample. Submission Forms and information on how to collect and ship samples can be found on the website: www.plantclinic.missouri.edu, or at your local Extension Office.

MU Plant Diagnostic Clinic • 28 Mumford Hall • Columbia, MO 65211 phone: (573) 882-3019 • email: plantclinic@missouri.edu

In This Issue

Mizzou Plant Diagnostic Clinic 1

Weed of the Month: Carolina Foxtail and Little Barley, Native, Cool-season Grasses........2

New Coordinator for Extension Nematology Lab........4

Soil Sampling for Soybean Cyst Nematodes to Prevent Potential Yield Losses........5

Alfalfa Weevil Larval Management Options for 2015 6

Weather Data for the week ending March 29, 2015 8

Weed of the Month: Carolina Foxtail and Little Barley, Native, Cool-season Grasses

by Mandy D. Bish & Kevin Bradley, Division of Plant Science, Weed Science

Carolina foxtail (Alopecurus carolinianus) and little barley (Hordeum pusillum) are cool-season grasses that can be seen in Missouri croplands, pastures, roadsides, etc. **(Figure 1)**. Both are native species and can be found in most continental U.S. states. Carolina foxtail, which is not directly related to the summer foxtails, is commonly found in moist areas of fallow agronomic fields and has been shown to be a potential host for fungi that cause diseases such as Rice blast[1]. Little barley is most commonly found in wheat-producing areas of the state, mostly north of the Missouri river. However, the grass can also be found in pastures, where its dense mat can suppress and delay the greening of more desirable forage grasses.

Figure 1: Carolina foxtail (A) and little barley (B) can be found in most of the continental United States, including Missouri.

Both grasses tend to have erect stems, although the Carolina foxtail stem can occasionally be bent. Carolina foxtail will grow from 4 to 6 inches in height while little barley can range from 4 inches to 2 feet. The leaf blades of Carolina foxtail are rough and may reach 6 inches in length; the leaf tips are pointed and can be sharp. Little barley leaf blades may be glabrous (lacks hairs) or have short hairs on both the upper and lower leaf surface, and the blades can reach up to 8 inches in length. The leaf blades of both grasses are less than ¼ inch in width. Both plants lack auricles, and have thin membranous ligules at the junction where the leaf blade meets the stem **(Figure 2)**. However, the ligules on little barley tend to be much smaller than those on Carolina foxtail, which are also small and approximately $1/8^{th}$ of an inch. The leaf sheath of Carolina foxtail lacks hairs while little barley may or may not have short hairs present on the sheath.

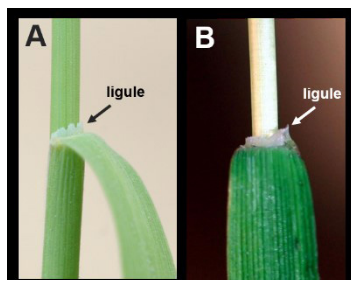

Figure 2: Both Carolina foxtail (A) and Little Barley (B) have membranous ligules at the junction where the leaf blade meets the stem

Figure 3: The Carolina foxtail seed head is cylindrical in shape and resembles timothy.

The seed head of Carolina foxtail has a cylindrical shape that can be up to 2 inches long **(Figure 3)**. Little barley has a seed head that is made up of flattened spikes and is more grain-like. The stiff awns tend to be less than ½ inch in length **(Figure 4)** and can injure grazing livestock.

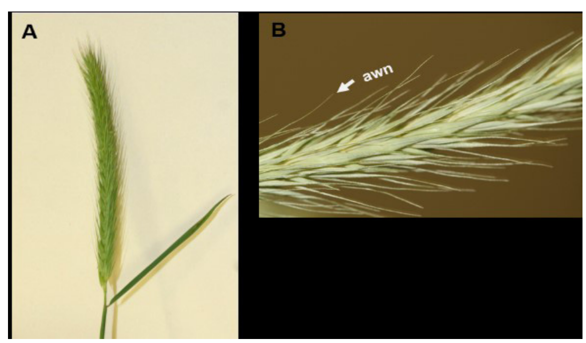

Figure 4: Little barley seed head is grain-like (A) in appearance. The awns can reach 1/2" in length (B).

Carolina foxtail seed viability is short-lived; however, the grass is capable of producing seed in spring or autumn, and this helps to ensure persistence of the grass population[2]. In general, Carolina foxtail is easy to control, and currently no known instances of herbicide-resistant Carolina foxtail are known. The best time to control the grass is in the fall or early spring, prior to seed set. Glyphosate and paraquat provide effective non-selective control. Spring applications of atrazine (4 pts/A) following wheat harvest and prior to weed emergence in field going to corn or sorghum or Canopy EX (chlorimuron + tribenuron) (1.1 to 3.3 oz/A) prior to soybean planting have been shown to be effective. Applications of Basis (0.33 to 0.5 oz/A) or Simazine (1 qt/A) after fall harvest and prior to emergence of Carolina foxtail have also shown good control.

Little barley is also best controlled in the fall or early spring. Gramoxone (paraquat) can provide effective burndown of little barley; however, continual use of the herbicide is likely to lead to paraquat-resistance as has occurred in a closely related species, smooth barley. Spring applications of Olympus (0.91 oz/a) or Maverick (0.67 oz/a) have shown 98 to 100% control of little barley 28 days after treatment[3]. A fall application of Olympus (0.91 oz/a) with Axiom (10 oz/a) or a Maverick (0.67 oz/a) application each provided only 50% little barley control when observed 28 days after treatment; however, control had increased to 95-97% when observed again the following April[3]. Sencor is another option for post-emergence control in select winter wheat varieties with metribuzin tolerance and can be applied at rates of 2 to 3 oz/A when wheat is at the 2-leaf to the 2-tiller stage; 5 to 6 oz/A from 3 to 4-tiller stage; and 5 to 8 oz/A when wheat has more than 4 tillers.

With regards to little barley control in pastures and hay fields, researchers in Alabama have found that late winter/early spring treatments including Accent (nicosulfuron) at 1 oz/A provided greater than 90% control of little barley 6 weeks following herbicide application. With the addition of metsulfuron (0.4 oz/A) and 2,4-D Amine (1 lb active ingredient/A), little barley control reached 97% six weeks after treatment4. Glyphosate at a low rate of 14 fl oz/A provided 90% or greater control at 3 weeks following treatment and over 95% control 6 weeks after the application[4].

Atrazine can also be used for little barley control; however, an 18-month grazing and/or foraging restriction will likely apply to almost any subsequent crop following atrazine treatment. As in all weed management programs, tank mixes and herbicide rotations should be implemented in the control of these grasses to avoid the development of herbicide resistance.

For more information on Carolina foxtail, little barley, or other grassy weeds:

Download a copy of Mizzou's Identifying Grass Seedlings (IPM1024):
http://weedscience.missouri.edu/publications/ipm1024.pdf.

Visit our weed identification web page: http://weedid.missouri.edu/
Download the free ID Weeds App in the Apple or Google Play Stores

Find the University of Missouri Weed Science extension on social media at:

Mizzou Weed Science
@ShowMeWeeds

[1] Jiay, Gealy D, Lin MJ, Wu L, and H Black. (2008) Plant Disease 92:504-507.
[2] Baskin CC, Baskin JM, and EW Chester. (2000) Journal of the Torrey Botanical Society 127(4): 280-290.
[3] Young B (2002) Determine the Effectiveness of various herbicides for control/suppression of little barley in winter wheat: www.siu-weeds.com/research/2002/502.pdf
[4] Enloe SF, Dorough H, Ducar JT, and JS Aulakh. (2012) Forage and Grazing lands: doi: 10.1094/FG-2012-0828-01-RS.

New Coordinator for Extension Nematology Lab

by Manjula Nathan

Amanda Howland joined MU as the new Coordinator for the Extension Nematology Lab effective Dec 1, 2014. She replaced Bob Heinz, who retired after 35 years of outstanding work at MU on Dec 31st. Amanda received her MS in Horticulture from Oregon State University and her masters research was focused on determining spatial distribution of plant-parasitic nematodes in Vineyards of the Pacific Northwest and determining the host status of the northern root-knot nematode on grapevines.

Amanda in her role as the coordinator for the Extension Nematology lab will be receiving and processing soil samples for soybean cyst nematodes and other plant parasitic nematodes, provide identification of plant parasitic nematodes, offer quarantine tests, HG Race tests, and respond to clients questions on submitting samples, and provide technical assistance to the regional agronomy and horticulture specialists, researchers, state and government, and industry personnel on when and how to samples for nematodes testing .

The Extension Nematology lab provides timely and accurate testing of soil and plant samples for the presence of plant parasitic nematodes. Recommendations are provided for management strategies to reduce the effect of nematodes on plant growth and yield for the citizens of Missouri. You can reach Amanda at 573-884-9118 or email her nematodelab@missouri.edu. Samples can be dropped off or mailed to the lab at 23 Mumford Hall, University of Missouri. Columbia, MO 65211.

Soil Sampling for Soybean Cyst Nematodes to Prevent Potential Yield Losses

by Manjula Nathan & Amanda Howland

Soybeans planting is just weeks away, and it is important that you test your fields for Soybean Cyst Nematodes (SCN) now before planting. SCN is a major concern to growers throughout the state. These parasitic round worms invade the plant roots and suck nutrients from the plants, decreasing their ability to produce adequate yields. The challenge with preventing SCN is that infected plants do not easily express symptoms. Fields can sustain up to 30% yield loss due to SCN without displaying any symptoms, making sampling the only way to identify a problem that you might not actually be seeing. Producers often ignore the possibility of SCN because they plant resistant varieties, but it is important to realize that SCN can adapt to the resistance lines if the same source is used year after year. Amanda Howland, the new coordinator for the MU Extension Plant Nematology labs, says 97% of the soil samples received since January 1, 2015 tested positive for having SCN, with egg counts ranging from 100 to over 35,000 eggs per cup of soil (250 cc3). Roughly 25% of the total samples tested had more than the threshold value of 10,000 SCN eggs/cup of soil. This proves it is important to sample soybean fields and check SCN egg counts periodically (every three years) to monitor whether the egg counts are increasing. Although typically fall is a good time to check fields for SCN because the results will be available for use in making decisions and plans for the next growing season, especially in terms of crop rotation and soybean variety selection, it is still not too late to sample the fields now ahead of planting.

Since SCN egg counts are only as good as the sample taken, here are a few tips for sampling for SCN:

1. Limit the size of the area being sampled: 10 - 20 acres is a good target.
2. Using a bucket and probe or shovel, walk the area in a W or Z pattern, sampling about 8 inches deep in the root zone between the rows. Take about 20 cores (with a shovel take ¼ cup of soil from near the shovel tip). Mix the cores well into a composite sample, and bag about a pint of it for submission. Do not let samples dry out! Nematodes are sensitive to heat. Do not leave samples in the sun or other areas of high temperature.
3. Label the plastic bag and ship it as soon as possible.
4. Fill out a submission form (available from our Website or your local extension agent) or on a piece of paper indicate:
    - Name, address, phone, and email (if you have email, results can be sent quickly)
    - County and cropping history
    - Type of test: SCN egg count ($20), HG Type test modified (In state: $75; Out of state: $125) or full (In state: $100; Out of state: $150), or Complete Nematode Analysis which includes plant parasitic nematodes identification ($30)
    - The mailing address for the lab is: Plant Nematology Lab, 23 Mumford Hall, University of Missouri, Columbia, MO 65211.

The SCN Egg Count test is what most soybean growers would need. If you notice a field that is slipping in yield, had high egg counts years ago, or you haven't had your soils tested for SCN in the last three years, a $20 SCN Egg Count test is a worthwhile investment that can offer peace of mind and save considerable yield loss. The HG Type test would be for the grower who has high egg counts after growing resistant lines for years. This test indicates the HG type (or race) of SCN in the field, and what sources of resistance would be good to choose when buying seed. The Complete Nematode Analysis test is a count of all the plant parasitic nematodes in the sample. (It does not give an SCN egg count.) This test is used if you feel you may have a nematode problem other than SCN. This test would also be important for growers in SE Missouri who may have the Root Knot nematode as well as SCN.

The Extension Nematology Lab has a website with more information on how to sample, the tests we provide, and how samples are analyzed in the lab. A submission form can also be downloaded from the site http://soilplantlab.missouri.edu/nematode. The turnaround time for the lab is typically 3-5 to working days.

For management decisions regarding SCN please refer the University of Missouri Extension Guide on Soybean Cyst Nematode: Diagnosis and Management. This guide can be downloaded at: http://extension.missouri.edu/publications/DisplayPub.aspx?P=g4450

Alfalfa Weevil Larval Management Options for 2015

By Wayne Bailey

Alfalfa weevil egg hatch is underway in many southern and some central Missouri alfalfa fields. In normal years, alfalfa weevil adults reenter alfalfa fields in late summer after field temperatures begin to cool, laying eggs during fall, winter, and spring seasons when temperatures go above 60oF for a few days or more. Eggs are laid inside plant stems and hatch in the spring depending on when they were laid. Alfalfa weevil eggs develop and eventually hatch after accumulating about 300 degree day heat units based on a 48OF developmental minimum temperature model.

Scouting

Alfalfa weevil larvae grow through 4 larval (worm) stages often referred to as instars, which feed on alfalfa plant tissues as they develop. After emerging from the egg, 1st instars (very tiny in size) move from the hatching sites inside plant stems and crawl to the terminal buds of alfalfa plants. Upon arriving at the terminal buds they burrow into the interior tissues of the green buds and begin feeding with their chewing mouthparts. As the buds expand, early feeding by 1st instars is often seen as small holes in expanding leaf tissues. Carefully opening terminal buds by pinching two sides of an individual bud and carefully pulling the bud in half will generally display the 1st instar if a larva is present. Similar holes in leaf tissue may also be result of feeding by 2nd instars, which due to their expanding size generally leave the interior of the buds to feed on the expanding leaf tissues found on external areas of the terminal buds. The presence of small holes in leaf tissue does provide an early warning that a potential alfalfa weevil larval problem may occur as larvae continue to develop. Numbers of 1st instars are not used in economic threshold calculations, which instead are based mainly on numbers of 2nd, 3rd, and 4th instars. The last two instars feed on all areas of the alfalfa plant, although a majority of larvae will be found in the upper 1/3 to 2/3 of the plant canopy unless numbers of larvae are excessive. As they grow, large larvae consume increasingly greater amounts of plant tissues which often result in economic crop loss through reductions in alfalfa quality and yield.

Although problems with alfalfa weevil have yet to occur this spring, producers in the southern counties of Missouri should scout fields on a weekly schedule beginning now and continue through first harvest. Producers in central and northern counties should begin scouting for alfalfa weevil within the next two weeks. Scouting for 2nd, 3rd, and 4th alfalfa weevil larvae is best accomplished by randomly collecting 50 alfalfa stems (10 stems at 5 different field locations) and tapping them into a white bucket. Larvae will generally be dislodged by this action and allow for an average number of larvae per alfalfa stem to be calculated. Caution should be used when collecting stems as larvae can be easily dislodged from the growing tip of the plant stem by rough handling. It is recommended that the top of the alfalfa stem be cupped in one hand while the plant stem is removed by cutting with a knife near the base of the stem. If an average of one or more larvae per stem is found in the 10 stem sample, then the economic threshold has been reached and control is justified. Infestations often first occur on warmer, south facing slopes of fields.

Management Options

An application of a labeled insecticide is the primary management option used in Missouri for early infestations of alfalfa weevil larvae on alfalfa. Early harvest of the alfalfa by either machine or livestock may be viable options for some producers in Missouri. If early harvest of alfalfa by machine is selected as a control strategy, then the crop is harvested approximately 7-10 days prior to the normal plant growth stage of 1/10 bloom. Data from Missouri studies indicate that alfalfa weevil larval numbers are reduced by about 95% with mechanical harvest and about 90% by cattle grazing in a management intensive grazing system. Most mechanical harvesters increase the rate of forage drying by crushing the plant stems which results in high mortality of alfalfa weevil larvae at the same time. Mob grazing (high numbers of cattle grazing often for short period of time) of alfalfa removes most alfalfa weevil larvae as they feed on the upper 2/3 of the standing plants. Producers using grazing as a control strategy must be aware of the bloat risk to cattle grazing green alfalfa and risk to the alfalfa stand due to trampling during wet conditions. If an insecticide application is selected, a list of insecticides recommended for alfalfa weevil control follows. Rates are given as amount of product applied per acre. The preharvest (PHI) interval lists the minimum number of days before harvest that an insecticide application can be applied. Some fields, especially in southern Missouri, may require two or more applications of insecticide to achieve acceptable control levels of this pest. Multiple insecticide applications are often due to several peaks in larval numbers resulting from numerous weevil eggs laid throughout fall, winter, and spring seasons.

Other factors that affect efficacy of insecticide applications when used to control alfalfa weevil larval in alfalfa include cool temperatures (below 60 degrees F), which slow the metabolic processes of the developing larvae and often slow the onset of larval mortality to levels below what is normally expected when organophosphate and pyrethroid classes of insecticides are applied at warmer temperatures. Steward insecticide, a recent entry into the alfalfa weevil insecticide market, has demonstrated increased larval mortality when used in cool conditions often experienced in the "Ozark Uplift Areas" of Southwest Missouri. Although Steward's performance is better than other insecticides under the cool conditions found in Southwest Missouri, efficacies on alfalfa weevil larvae are generally equivalent at more normal conditions. Some other factors include use of low rates of insecticides when larval populations are very high, less than optimal coverage of the infested crop, or possible development of resistance to the pesticide(s) being used.

Recommended Insecticides for Alfalfa Weevil Larvae in Alfalfa - 2015

Chemical name	Common name	Rate of Formulated Material	Preharvest Interval
Beta-cyfluthrin	*Baythroid XL	1.6 to 2.8 fl oz/acre	7 days
Lambda-cyhalothrin + chlorantraniliprole	*Besiege	6.0 to 10.0 fl oz	1 day forage, 7 day hay
Chlorpyrifos + gamma cyhalothrin	*Cobalt Advanced	19 to 38 fl oz/acre	7-14 days
Chlorpyrifos + gamma cyhalothrin	*Cobalt	19 to 38 fl oz/acre	7-14 days
Dimethoate	Dimethoate/Dimate	see specific label	10 days
Phosmet	Imidan 70W	1 to 1 1/3 lb/acre	7 days
Methomyl	*Lannate	0.9 fl oz/acre	7 days
Chlorpyrifos	*Lorsban Advanced	1 to 2 pts/acre	7 - 21 days
Chlorpyrifos	*Lorsban 4E	1 to 2 pts/acre	7 - 21 days
Chlopyrifos	*numerous products	see specific labels	
Zeta-cypermethrin	*Mustang Maxx	2.24 to 4.0 fl oz/acre	3 days
Permethrin	*numerous products	see specific label	7 - 14 days
Gamma-cyhalothrin	*Proaxis	1.92 to 3.2 fl oz/acre	1 day forage, 7 day hay
Zeta-cypermethrin	*Respect EC	2.24 to 4.0 fl oz/acre	3 days
Carbaryl	Sevin 4F	1 qt/acre	7 days
Carbaryl	Sevin XLR Plus	1 qt/acre	7 days
Zeta-cypermethrin + chlorpyrifos	*Stallion	5.0 to 11.75 fl oz	7 days
Indoxacarb	*Steward 1.25 EC	4 to 11.3 fl oz/acre	7 days
Cyfuthrin	*Tombstone Helios	1.6 to 2.8 fl oz/acre	7 days forage/hay
Lambda-cyhalothrin + chlorantraniliprole	*Volian Xpress	6.0 to 9.0 fl oz	1 day forage, 7 day hay
Lambda-cyhalothrin	*Warrior 1 CS	1.92 to 3.2 fl oz/acre	1 day forage
Lambda-cyhalothrin	*Numerous products	see speciic labels	1 day forage, 7 days hay
Read and follow all label direction, precautions, and restrictions.			
*** Designated a restricted use product.**			

Weather Data for the Week Ending March 29, 2015

		Weekly Temperature (°F)						Monthly Precipitation (in.)		Growing Degree Days‡	
Station	County	Avg. Max.	Avg. Min.	Extreme High	Extreme Low	Mean	Departure from long term avg.	March 1-29	Departure from long term avg.	Accumulated Since Apr 1	Departure from long term avg.
Corning	Atchison	51	32	65	26	41	-5	1.59	-0.36	*	*
St. Joseph	Buchanan	50	33	65	27	41	-6	1.45	-0.51	*	*
Brunswick	Carroll	52	35	68	30	43	-5	1.70	-0.53	*	*
Albany	Gentry	50	33	65	26	41	-5	0.98	-1.11	*	*
Auxvasse	Audrain	52	33	59	29	42	-5	1.99	-0.64	*	*
Vandalia	Audrain	50	32	57	28	41	-5	1.94	-0.82	*	*
Columbia-Bradford Research and Extension Center	Boone	53	34	59	31	42	-6	1.87	-1.00	*	*
Columbia-Capen Park	Boone	55	35	62	30	44	-5	2.02	-0.75	*	*
Columbia-Jefferson Farm and Gardens	Boone	53	34	60	31	43	-5	1.88	-0.98	*	*
Columbia-Sanborn Field	Boone	53	35	61	31	44	-5	2.25	-0.58	*	*
Columbia-South Farms	Boone	53	34	60	31	43	-5	1.86	-1.05	*	*
Williamsburg	Callaway	53	34	63	29	42	-5	1.87	-0.98	*	*
Novelty	Knox	48	31	62	27	39	-7	1.09	-1.27	*	*
Linneus	Linn	49	32	65	27	40	-6	1.47	-0.76	*	*
Monroe City	Monroe	49	32	60	26	40	-7	1.98	-0.46	*	*
Versailles	Morgan	56	36	64	33	46	-3	1.79	-0.98	*	*
Green Ridge	Pettis	55	35	61	32	44	-4	1.10	-1.53	*	*
Lamar	Barton	61	41	74	37	49	-1	2.09	-1.20	*	*
Cook Station	Crawford	59	34	71	28	46	-4	3.23	-0.17	*	*
Round Spring	Shannon	61	33	79	26	46	-3	5.88	+2.36	*	*
Mountain Grove	Wright	57	36	72	29	46	-2	4.14	+0.57	*	*
Delta	Cape Girardeau	57	35	74	28	46	-5	7.30	+3.33	*	*
Cardwell	Dunklin	61	39	74	30	50	-3	5.25	+1.30	*	*
Clarkton	Dunklin	61	38	75	31	50	-2	6.79	+3.25	*	*
Glennonville	Dunklin	61	39	75	31	50	-2	7.82	+4.34	*	*
Charleston	Mississippi	60	38	75	29	49	-2	7.57	+4.01	*	*
Portageville-Delta Center	Pemiscot	61	39	75	32	50	-3	6.78	+3.08	*	*
Portageville-Lee Farm	Pemiscot	61	40	76	32	51	-2	6.53	+2.86	*	*
Steele	Pemiscot	61	39	76	32	50	-3	5.56	+1.60	*	*

‡Growing degree days are calculated by subtracting a 50 degree (Fahrenheit) base temperature from the average daily temperature. Thus, if the average temperature for the day is 75 degrees, then 25 growing degree days will have been accumulated.

Weather Data provided by Pat Guinan
GuinanP@missouri.edu
(573) 882-5908

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Editor: Amy Hess (hessa@missouri.edu)