IPM-1

Kentucky Integrated Crop Management Manual for Field Crops

"ALFALFA"

Agriculture and Natural Resources • Family and Consumer Sciences • 4-H Youth Development • Community and Economic Development

Kentucky Integrated Crop Management Manual for Alfalfa

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For additional and current information please consult the following web sites:

For more IPM information and links to many pest and crop management sites view the IPM web page at:
http://www.uky.edu/Ag/IPM/

For the most current information on pests view the **Kentucky Pest News** at:
http://www2.ca.uky.edu/agcollege/plantpathology/extension/kpnindex.htm

For up-to-date weather, and crop and pest models view Ag-weather at:
http://wwwagwx.ca.uky.edu/Gisproducts.html

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PREFACE

Agriculture is the world's most important industry. This level of importance will continue due to rapidly expanding populations which demand increased amounts of food and fiber. Crop protection problems associated with this increased production have become more complex. A simplistic approach to pest control leads to serious environmental complications. A truly successful pest management program must take a multi- disciplinary, multi-crop approach in order to supply the farmer with reliable pest control information. An approach to crop production based on sound economic, ecological, technical and social considerations is required to assist the farmer to achieve needed production levels, while maintaining food safety and environmental quality.

PHOTO CREDITS

Ric Bessin. University of Kentucky Department of Entomology. Potato leafhopper, pg.6; Pea aphid, pg.7;Armyworm, pg. 9; Fall armyworm, pg. 9; Yellow striped armyworm, pg. 10

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Unknown sources. Parasitoid wasp and its cocoon, pg. 3; Adult alfalfa weevil, pg. 5; Striped blister beetle and Margined blister beetle, pg. 8; Green cloverworm, pg. 10; Spring black stem leaf spot, pg. 26; Phytopthora root rot, pg. 27; Mycoleptodiscus Crown Rot, pg. 28; Bacterial wilt, pg. 28.

Paul Vincelli. University of Kentucky. Sclerotinia crown and stem rot, pg. 25; Lepto leaf spot, pg. 25;

University of Illinois. Fall armyworm head, pg.9. http://bulletin.ipm.illinois.edu/pastpest/articles/v9816f.html

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ALFALFA SCOUTING

Scouting for pests in alfalfa will be quite different from the techniques used in scouting other crops, especially for insects.

Pests	Monitoring Stations	Procedure/Location
Stand Count	Random	Number of crowns/4 square feet
Insects	Random	30 stems or 100 sweeps/field or 1 sq. ft.
Weeds	Random	100 square feet
Diseases	Random	10 to 50 foot radius

Pest	Field Size	No. of Locations
Spittlebugs	---	10
Alfalfa Weevil	---	30/stems/field
Potato Leafhopper	---	5
Weeds & Diseases	Less than 20 acres	3
	Greater than 20 acres	1/10 acres

Stand Counts for Alfalfa

Stand counts should be made when the first visit is made to the field. See page 34 for stand count procedure.

Record: Record the number of crowns per four square feet and the number of stems per four square feet.

Scouting Procedures for Insects in Alfalfa

Lee Townsend

The crop has two main pests: first cutting - the alfalfa weevil; second and third cuttings - the potato leafhopper. Occasionally, there are damaging infestations of grasshoppers and fall armyworms. While a variety of insects, spiders and other arthropods live in alfalfa fields during the growing season, most are not pests. Many, including lacewings, lady beetles, and small beneficial wasps are important natural enemies. These insects help to regulate pest numbers in alfalfa and nearby fields.

Alfalfa Weevil

The grub-like larva of the alfalfa weevil is pale green and legless. It has a dark brown to black head and a thin green line down the center of its back. Weevil larvae feed in plant tips for about 3 weeks, chewing small holes in the leaves.

Alfalfa weevil damage and larva

Note: The clover leaf weevil larva also feeds in alfalfa tips and can be mistaken an alfalfa weevil larva. It has the same legless, green grub appearance but its head is tan instead of dark brown. Also, the white stripe down its back is bordered with some red or pink smudges; the marks are absent in the alfalfa weevil. Clover leaf weevils do not cause economic damage to alfalfa and should not be included in weevil counts.

Management decisions are based on numbers of alfalfa weevil larvae per 30 stems, height of the alfalfa, and the number of degree days (base 48) that have accumulated from January 1 until the time of sampling.

When to Sample:

Fields should be checked for first time during the week when the degree day accumulation in your area reaches 190. When you first enter the field, check several alfalfa tips for feeding damage and larvae. If no damage is apparent, then walk through the field in a "Z" or "U" shaped pattern and check for feeding damage or weevils at several random spots. There is no need to collect stem samples until you can find damage or the insects.

If feeding damage or weevils are found, sample immediately and return every 7 days until the weevil season is over. Check the degree day accumulation each time you return and use the Alfalfa Weevil Pest Management Recommendation Table with the appropriate degree day accumulation for that day.

The following procedure allows you to sample a field for alfalfa weevil larvae to collect information that can be used to evaluate damage potential. You can get the most current alfalfa weevil degree day total from your county extension office or the UK Ag Weather web site at: http://wwwagwx.ca.uky.edu/Gisproducts.html

Field Sampling Procedure

Equipment needed:

1.) Yardstick or other measuring device
2.) 3.5 gallon plastic bucket (white or yellow)
3.) Alfalfa Weevil Pest Management Recommendation Table

Sampling Procedure:

Weevil numbers can vary from field to field; each one should be sampled separately.

Collect a total of 30 stems at widely spaced intervals while you walk through the field in a giant "U" or "Z" shaped pattern. Avoid the edge; walk at least 20 paces into the field before starting.

1. Stop randomly, stoop down and cup one hand around the plant tip so that any larvae present do not fall out. Use your other hand to break the stem at ground level.

2. Place each stem tip first into the bucket. After collecting 30 stems, return to the edge of the field to count larvae and measure stems.

3. Pick 4 or 5 stems at a time and beat them vigorously against the inside of the bucket to knock off the weevil larvae. Set stems aside to be measured. After you have finished, count all of the weevil larvae in the bucket and record this number on the scouting form.

4. Randomly select and measure the length of 10 stems to the nearest inch. Start from the base, where the green tissue is distinguishable from the brown woody tissue, to the growing tip. Record each measurement, then calculate the average stem height and record this number on the scouting form.

5. Note the growth stage of each stem as: pre-bud - no buds are apparent; bud - only buds are present, or flower if any flowers can be seen. Calculate the percentage of the plants in the bud or flower stage and record this.

6. Use the number of larvae per 30 stems and average alfalfa height in Alfalfa Weevil Pest Management Recommendation Table for the current degree day range in your area and determine the recommended action.

Using the Management Tables

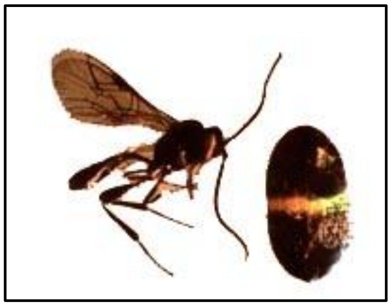
The Alfalfa Weevil Pest Management Tables will help you to make accurate pest management decisions. Use the section with the number of degree days that have accumulated up to that time. Example: Suppose that you took a sample when 200 DD had accumulated and found 24 larvae in the 30-stem sample. The average alfalfa stem length was 4 inches.

According to the 190-225 DD table, application of a long residual insecticide would be needed if you had collected 67 or more larvae on the 4-inch tall alfalfa. There is no need to treat now but there were more than 15 so you should sample again in 2 days. Although the sample is below a treatable level, eggs are hatching and weevils may reach a damaging number soon.

Early harvest, instead of an insecticide application, is an option if 30% or more of the crop has reached the bud stage of development.

Early harvest saves insecticide and application costs and damage caused as the sprayer drives over the field. It also establishes an efficient cutting schedule for the growing season.

A tiny wasp that attacks weevil larvae is present throughout the state and helps to regulate populations. The oblong brown wasp cocoon with a narrow light band around the middle can be found inside the white cocoons of the weevil around the time of first harvest.



Parasitoid wasp and its cocoon

ALFALFA WEEVIL PEST MANAGEMENT RECOMMENDATIONS TABLE

DD 190-225							
#Larvae	27	47	67	85	100	115	130
Alfalfa Height	2"	3"	4"	5"	6"	7"	8"

- Apply a long residual insecticide if the larval number you find is greater than the number in the table for the height of alfalfa sampled. Spraying now protects alfalfa weevil parasites emerging later.
- Sample again in two days if larval number is not exceeded but is above 15 (Expect to spray at that time) OR sample again in seven days if you find less than 15 larvae per 30 stems.

DD 226-275							
#Larvae	15	17	19	20	20	20	20
Alfalfa Height	2"	3"	4"	5"	6"	7"	8"

- Spray a long residual insecticide if the larval number you find is greater than the number in the table above for the height of alfalfa sampled.
- Sample again in seven days if you find less than the number of larvae for the appropriate alfalfa height.

DD 276-325									
#Larvae	37	45	60	67	83	94	105	125	135
Alfalfa Height	4"	5"	6"	7"	8"	9"	10"	11"	12"

- Spray a medium residual insecticide if the larvae number you find is greater than the number in the table above for the height of alfalfa sampled.
- Sample again in seven days if you find less than the number of larvae for the appropriate height.

DD 326-375									
#Larvae	82	95	105	105	105	105	105	105	105
Alfalfa Height	8"	9"	10"	11"	12"	13"	14"	15"	16"

- Spray with a short residual insecticide if larvae number is exceeded for the size of alfalfa sampled.
- Sample again in two days if alfalfa is less than 16" and the number of larvae found is more than 20 but less than 82 per 30 stems OR sample in seven days if alfalfa is taller than 16" and the number of larvae found is less than 20.

DD 376-525							
* #Larvae	52	60	64	68	72	76	80
Alfalfa Height	12"	13"	14"	15"	16"	17"	18"

- Spray with a short residual insecticide if the number of larvae are exceeded for the size alfalfa sampled or **harvest now if the alfalfa is in the 30% bud stage or greater.**
- Resample in two days if larval number is less than but within 20 of the number justifying a treatment.

Evaluating Post-Harvest Alfalfa Weevil Larval And Adult Damage

If early harvest was used as a weevil management tool, there may be enough feeding by larvae or newly-emerged adults to slow the recovery of the stand. The alfalfa weevil is an approximately 3/16 inch long brown snout beetle with a dark stripe down the back. It causes "notch-like" feeding holes on leaves giving them a feathery appearance. Watch fields carefully for normal green-up after harvest and sample for weevils if there appears to be a delay.

Adult alfalfa weevil

The table below can be used to evaluate larval numbers and plant height to determine if control is necessary. Treatment may be justified if adult weevils can be seen feeding on 50% or more of the crowns and regrowth is prevented for 3 to 6 days. Use low rates if an insecticide application is necessary.

Assessing Feeding by Alfalfa Weevil On First Cutting Regrowth

(5-7 days after harvest)

*#/Larvae	20	25	33	40	47	54	60
Alfalfa Height	2"	3"	4"	5"	6"	7"	8"
**#/Larvae	17-20	17-24	17-32	20-39	23-46	23-53	23-59

*Spray with a short residual insecticide if the number of larvae/30 stems exceeds the number in the table above for the appropriate alfalfa height.
**Sample again in 2 days if the number of larvae/30 stems is in this range for the appropriate alfalfa height. If numbers are below this level, no treatment should be necessary.

Potato Leafhopper

Potato leafhopper

The potato leafhopper is the key pest of 2nd and 3rd cuttings. The adult is a 1/8- inch long, pale green, wedge-shaped insect with 2 pairs of small wings. The nymphs (immature stages) have the same shape and color of the adults but are smaller and wingless.

Potato leafhoppers are sap feeders so they do not cause holes in the leaves like those from weevil larvae. They cause "hopperburn", a distinctive yellow wedge shape at the tip of the leaf. Feeding by nymphs and adults causes the injury that reduces alfalfa quality and may weaken stands. Both stages feed on the undersides of leaves and move quickly when disturbed. Because of their size and subtle damage, a sweep net is used to sample fields accurately.

Alfalfa height and number of leafhoppers per 100 sweeps are used to make a management decision.

When to sample:

Sample 7 to 10 days after harvest, then weekly to determine if leafhopper numbers are increasing. Fall and spring seedings of alfalfa are especially susceptible to injury and should be sampled weekly, beginning in late April or mid-May and continuing until the first harvest. These fields then should be checked 7 to 10 days after harvest and weekly after that.

Hopperburn caused by potato leafhoppers.

Alfalfa fields can be sampled anytime during the day as long as the foliage is not wet. Sweeping alfalfa when it is wet from dew or rain is difficult and the samples are almost impossible to count.

Field Sampling Procedure

Equipment needed:

1. Yardstick or other measuring device.
2. 15-inch diameter sweep net.

Sampling procedure:

1. Use the net to take groups of 20 sweeps at 5 different locations over the field (100 total).

2. Count all potato leafhoppers (adults and nymphs) collected in each 20-sweep sample and record the number before going to the next area of the field.

3. Randomly collect 10 stems as you walk back out of the field using the selection procedure described in the "Alfalfa Weevil" section.

4. Measure the 10 length of each stem from tip to ground level. Calculate the average stem height, examine each of the ten stems for buds or flowers and record this information.

5. Add up the total number of potato leafhoppers per 100 sweeps, then use the numbers with the following table to determine if potato leafhopper numbers are great enough to justify a control measure.

Determining the need for leafhopper control:

Use average alfalfa stem height and number of insects per sweep or 100 sweeps to determine if an insecticide treatment is justified.

Economic Threshold Table for Potato Leafhopper

Average Stem Length (inches)	Number of Leafhoppers per sweep	Number of Leafhoppers per l00 sweeps
Less than 3	0.2	20
3 – 6	0.5	50
8 - 10*	1.0	100
12 - 14*	2.0	200

*Note: If the alfalfa is at 30% bud or more you should plan on cutting within 7-10 days. If this is the case, you should not spray. However, soon after cutting you should resample the field to determine the need for control.

Aphids

Aphids are small, light green pear-shaped insects that feed on sap from alfalfa plants in the spring and fall. The pea aphid is the most common species in Kentucky alfalfa but very high populations are needed to cause concern. Natural enemies, a fungus disease, or hot weather usually reduces their numbers before plants are stressed. Pea aphids may become very abundant during long, cool springs but many are needed before a control measure should be considered.

Pea aphid

<u>Sampling Procedure</u>

Pea aphids may be found when checking first cutting alfalfa for weevil feeding. A close examination of plant stems in several parts of a field and counting numbers of aphids per stem will help in making a management decision.

Black legume aphid

An average of 5 or more aphids per seedling stem, 40+ on alfalfa less than 10 inches tall, and 75 + on alfalfa greater than 10 inches tall would be needed before considering an insecticide application.

<u>Note:</u> There are some aphid species in other parts of the US that can cause serious damage to alfalfa. These include the blue alfalfa aphid, cowpea or black legume aphid, and the spotted alfalfa aphid. The black legume aphid can appears in late summer and can survive mild winters in Kentucky. If you see aphid-infested alfalfa that is wilted, discolored or distorted, take a sample of the plants and aphids to your county extension office for identification.

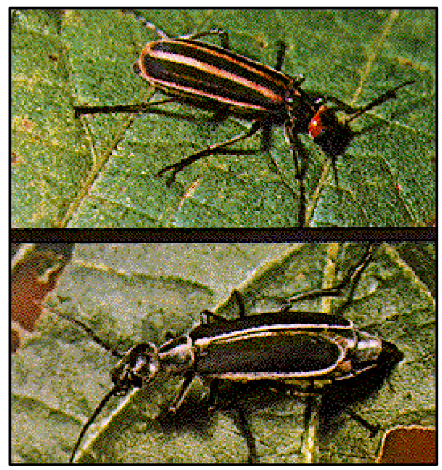
Grasshoppers

Grasshoppers feed on a wide range of plants but usually do not damage alfalfa unless conditions are very dry and alfalfa is one of the few succulent plants available to them. In general, alfalfa needs to be checked for grasshopper damage only during very dry periods. If enough are present, they can cause extensive damage in a short time.

Sampling Procedure

Grasshopper control may be justified if there are more than 15 grasshoppers per sq. yard in alfalfa that is less than 6" tall. Field border treatments should be made if there are 20 to 40 nymphs or adult grasshoppers per square yard in adjacent non-crop land fields. Visualize a square yard area on the ground and estimate, as accurately as possible, the numbers that jump from the area as you walk through it.

Blister beetles



Top - Striped blister beetle; Bottom – Margined blister beetle

Blister beetles have long (3/4 to 1-1/4 inch), narrow bodies, broad heads, and long antennae. The segment between the head and abdomen is narrow and resembles a “neck”. The front wings are flexible in contrast to the hard front wings of most beetles. Three species are relatively common in Kentucky. The striped blister beetle has orange and black stripes on its wing covers: the black blister beetle is jet black, and the margined blister beetle is black with a thin gray stripe around wing covers.

Blister beetles are active from mid-July through August. They tend to occur in group with the striped and margined blister beetles preferring to feed on flowering plants. They do not damage alfalfa but can be a lethal contaminant in hay consumed by horses. Blister beetles contain a toxic chemical (cantharidin) that can cause severe sickness and sometimes death if horses eat dead beetles trapped in cured hay. Cattle are not as susceptible to poisoning as are horses.

Sampling Procedure

Blister beetles are attracted to flowers plants so they can be found in fields with blooming weeds or alfalfa. Areas of a field with many flowering plants should be inspected visually or sampled with a sweep net. There are no thresholds for these insects but steps can be taken to greatly reduce the chances of them contaminating hay.

Blister beetles will fly out of fields as cut hay wilts and cures. However, if beetles are present at harvest, and the hay goes through a crimper or conditioner as it is being cut, *Striped, margined, and black blister beetles* many of them can be crushed and remain in the hay where they are still toxic, especially to horses.

Caterpillars

Some caterpillars will feed on alfalfa during the growing season. The larval feeding stage for each species lasts for 4 to 5 weeks.   Most have two or more generations each year. Usually they are of no consequence other than providing food for natural enemies. Occasionally, they can be abundant enough to cause serious defoliation, leaving only plant stems.

Sampling Procedure

If caterpillar defoliation is noticed, use a 15-inch diameter sweep net to sample the field. Take 1 set of 20 sweeps per 5 to 10 acres. Record the number and length in inches of caterpillars found, or a representative sample of each species.

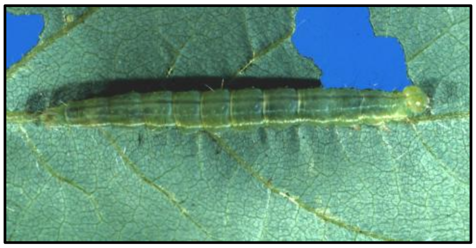
Common name	Description
Alfalfa caterpillar	Velvety green body with white lines along side. 1.5 inches long when full grown
Armyworm	Dark green-brown to black with pale with, orange and dark brown stripes along body. Yellow brown head with dark network of markings. 1.5 inches long
Fall armyworm	Similar to armyworm but with distinct pale inverted "Y" on head. 
Green cloverworm	Light green caterpillar with 3 pairs of thin white stripes along body. They thrash violently when handled. 1.5 inches long
Variegated cutworm	Gray to black caterpillar with single pale yellow spot on back of 4 to 7 body segments. Curls into "C" when disturbed. 1.75 inches long
Yellow striped armyworm	Green to black caterpillar with cream yellow to orange stripes along back. Dark spot on side of the first legless body segment. 2 inches long

The green cloverworm and yellowstriped armyworm are most common during the summer.
Treatment may be justified if there are more than 6 caterpillars of any species per sweep and most larvae are 75% of less of their mature length.

Alfalfa caterpillar

Armyworm

Fall armyworm


Green cloverworm


Yellow striped armyworm

Variegated cutworm

Meadow Spittlebug

Meadow spittlebug nymphs are small, light green, sap-feeding insects that develop in frothy masses on alfalfa and grass stems in the spring. Under normal growing conditions infestations are rarely severe enough to affect plants. Some stunting or wilting may occur under dry conditions if there is one (1) or more masses on each alfalfa stem and plants are less than 6 inches tall. Even if numbers justify treatment, spittle masses protect the insects from spray droplets and reduce the effectiveness of an insecticide application.


Spittlebug

ALFALFA KEY INSECT CALENDAR FOR KENTUCKY

Insect	March	April	May	June	July	August	September
Alfalfa Weevil	* ** ** ** **********						
Potato Leafhopper			** ** ** ** ** ** ** ** *				

BENEFICIAL INSECTS PICTURE SHEET

Green Lacewing Larva

Green Lacewing

Lady Beetle Larva

Lady Beetle

Syrphid Fly Larva

Syrphid Fly

Spined Soldier Bug

Damsel Bug

Scouting Procedures for Weeds in Alfalfa

J. D. Green and James R. Martin

Weeds compete with alfalfa for water, light, nutrients and space. This competition decreases yield, lowers forage quality, increases disease and insect problems, causes premature loss of stand, and creates harvesting problems. Some weeds that are present in alfalfa fields are unpalatable to livestock or may irritate the animals consuming the forage. A few plants are even potentially poisonous.

In most grain and fiber crops weeds are separated from the crop at harvest, but with forage crops such as alfalfa, the weeds are gathered with the crop. The addition of weeds to alfalfa forage may decrease per acre yields, but more importantly, may lower the quality of the forage. The decrease is often in the form of lowered protein and digestibility of the feed; the latter causing livestock to reduce forage consumption. However, a vigorous stand of alfalfa usually will not have a serious weed problem.

Weeds can be grouped into three categories based on their life cycle.

-Annuals - plants which complete the germination, vegetative and reproduction process in one growing season or year. The terms "winter annual" and "summer annual" refer to that portion of the year when these weeds grow.

-Biennials - plants that live during two growing seasons. The first season is vegetative growth and the second season is vegetative and reproductive growth.

-Perennial - plants that live for more than two years. Perennial usually have more than one reproductive system. Some of the vegetative reproductive systems of perennial include fleshy taproots, tubers, rhizomes, stolons, bulbs, and bulblets.

An understanding of the life cycle and reproductive methods of plants is necessary for determining a weed control program for alfalfa. As a general rule, perennial weeds are more difficult to control because of their multiple reproductive mechanism.

Weed seeds are usually very small and can be easily dispersed by wind, water and animals, including man. Weeds in a forage crop, such as alfalfa harvested for hay, can be widely distributed during harvesting, transporting and feeding. Weeds can eventually be spread over a large area, not only from the physical spreading of the hay during feeding, but also by livestock manure after the weed seeds have passed through an animal's digestive tract. Alfalfa harvested from a field with serious problem weeds should not be fed in a field that is not infested.

The presence of weeds in alfalfa indicates that less than optimum production could be expected; however, determining the extent of yield reduction due to various weeds is difficult. Economic threshold levels have not been established for most weeds in alfalfa. Attempts to establish economic threshold levels are extremely complex and difficult because of the various competitive effects of weeds on forage yield and quality.

<u>Cultural Control</u>

Vigorous, dense stands of forage legumes seldom have serious weed problems. Management practices to establish and maintain such stands include:

1. liming and fertilizing the field as recommended by soil analysis;
2. seeding well adapted, vigorous, long-lived varieties;

3. buying weed-free seed;
4. providing proper soil drainage;
5. cutting the forage at first flower;
6. timely control of insects and disease;
7. crop rotation to interrupt the buildup of certain weeds.

Following these practices usually keeps the forage highly competitive with most weeds. Nevertheless, the aggressive nature of some weed species allows them to become established in spite of preventative efforts. Herbicide treatments or returning to a cultivated row crop for several years may be necessary at times. It should be mentioned that an herbicide treatment without good cultural practices will seldom give the expected results. Maximum benefits are obtained when herbicides are used in conjunction with recommended cultural practices. Preplant tillage does not eliminate weeds, but it will weaken them and reduce their numbers. Adequate seedbed preparation allows alfalfa to compete more effectively with weeds. Mowing can be used to control tall growing weeds and the best time to begin mowing is in early summer when underground root reserves are low. This practice not only prevents weed seed production, but encourages re-growth from the roots of perennial weeds. Repeated mowing of perennial weeds gradually starves out their underground parts and they die.

Herbicide Treatments

Herbicides are available to control many of the common annual weed problems in established legumes. However, several perennial broadleaf weeds are visible in the spring of the year when no herbicide treatment can be made. Also, most herbicides labeled for alfalfa must be used only on pure legume stands since they have the potential to injure or kill grasses seeded with the legume. Consult Extension publication AGR-148 "Weed Control Strategies for Alfalfa and Other Forage Legume Crops" for herbicide recommendations.

Scouting Methods

Weed density, or infestation level, will be determined by estimating the percent of ground area occupied by weeds. This can easily be accomplished by randomly selecting areas in the field (refer to the section of this manual on sampling sites). An area of approximately 30 ft by 30 ft should be selected and the percentage of weeds present recorded. Record the predominant species and their size at the time of sampling.

Two distinct weed complexes will be encountered in alfalfa fields. The winter weed complex will be present during the fall and spring through the first harvest and sometimes into the second harvest. The summer weed complex will be present after the first harvest through a killing frost in the fall.

The following weeds are examples of ones commonly found in Kentucky alfalfa fields. Obviously, these do not include all those of economic importance. However, the sampling techniques should apply to most weeds encountered in alfalfa.

Keep in mind that fields which appear almost weed free could have a 5% weed density. Only in extremely poor stands will weed infestations in excess of 50% occur. Record percentages in multiples of 5%, i.e. 15% or 20%, not 17% or 21%. This technique will work equally well in both pure alfalfa and alfalfa-grass mixtures.

Recording Data

Record the estimated percent of the ground covered by all weeds. List the predominate species of weeds and record the estimated percent of the ground covered by each weed. **The percentage of the ground covered by all weeds should equal the sum of the percentages of ground covered by each weed.** See pages 19 and 20 for assistance in estimating the total percent of a field covered by weeds.

WINTER WEEDS	SUMMER WEEDS
common chickweed	large crabgrass
shepherdspurse	giant foxtail
yellow rocket	yellow foxtail
fleabane	goosegrass
field pennycress	johnsongrass
wild garlic	pigweeds
dandelion	lambsquarters
henbit	curly dock
deadnettle	broadleaf dock
musk thistle	buckhorn plantain
	broadleaf plantain

MAPPING FIELDS FOR WEEDS

One of your most important duties as a scout is to prepare a "weed map" of each field that you survey. This map will be of benefit to the grower in planning his weed control program for the coming years.

Steps in preparing a "weed map".

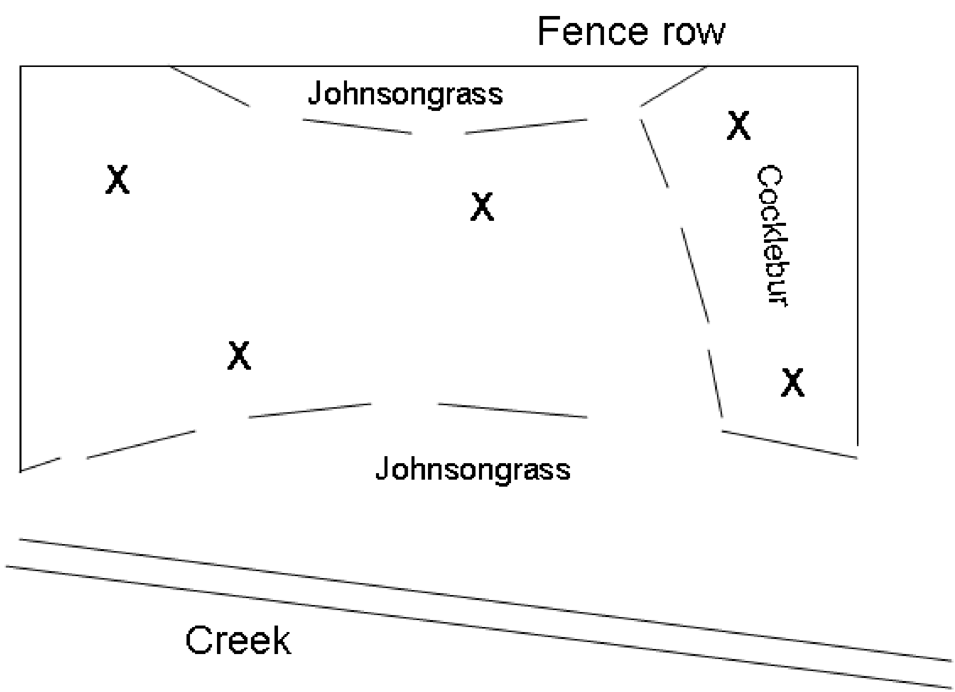
1) Outline the shape of the field on the report form. Make notations as to locations of fences, roads, woods, etc.

2) Mark the approximate locations of severe weed infestations or weeds not listed on the survey form and mark the locations where you make your counts.

3) This map should be drawn each time you scout the field.

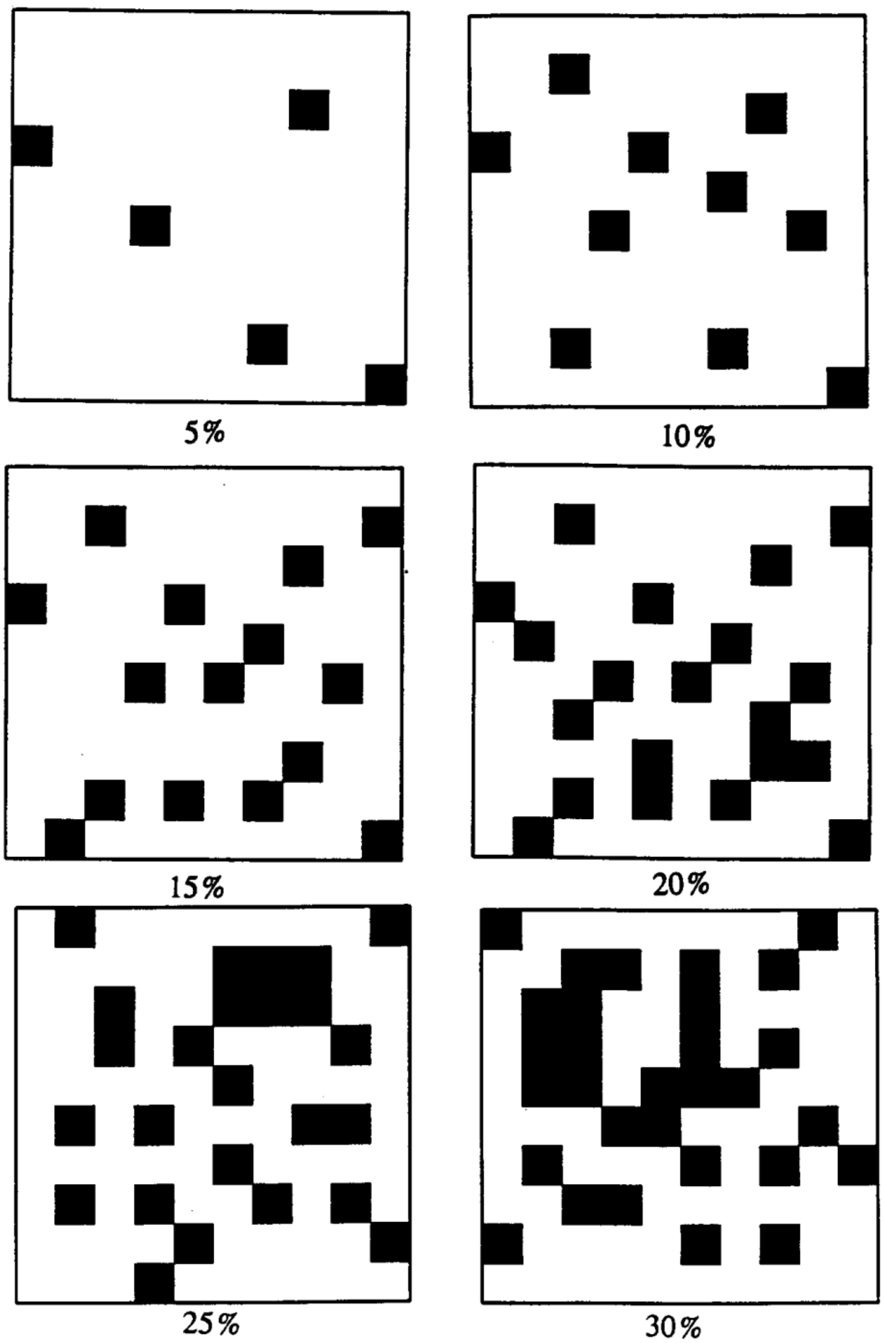
4) Be sure and indicate any weed problems on the map that would assist the grower in making management decisions.

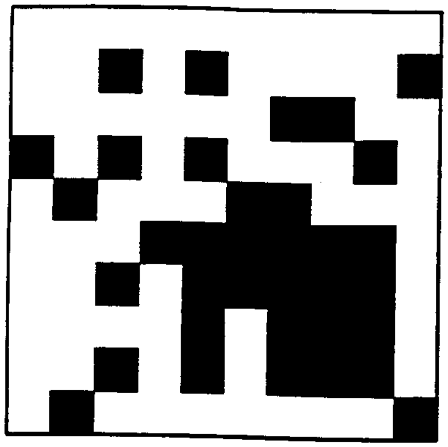
The following example can be used as a guide in preparing a "weed map" of your fields.



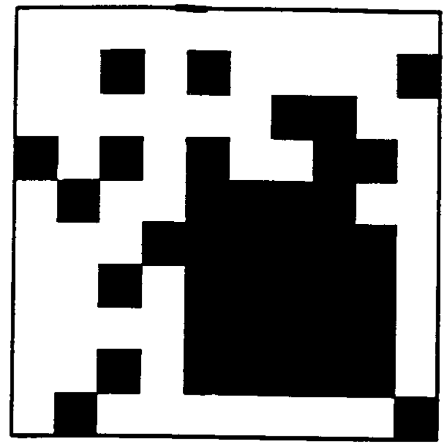
Estimated Percent Coverage for Weeds

Percent of coverage for the black area is given below each box.

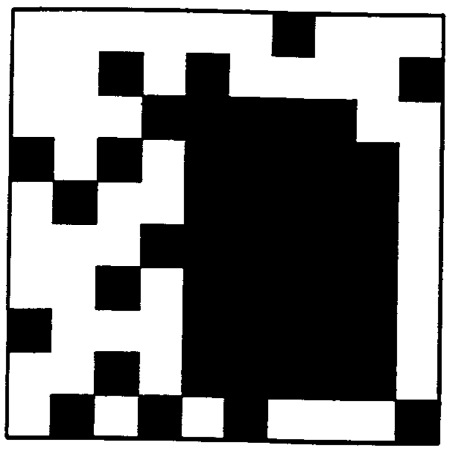




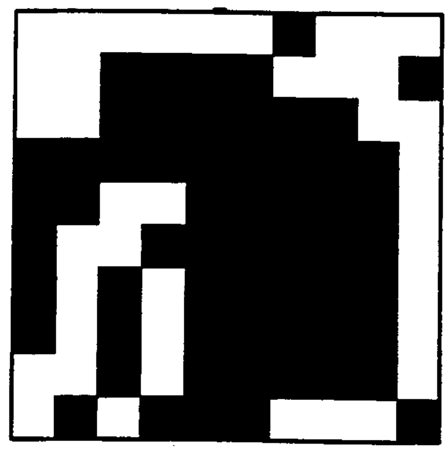
35%



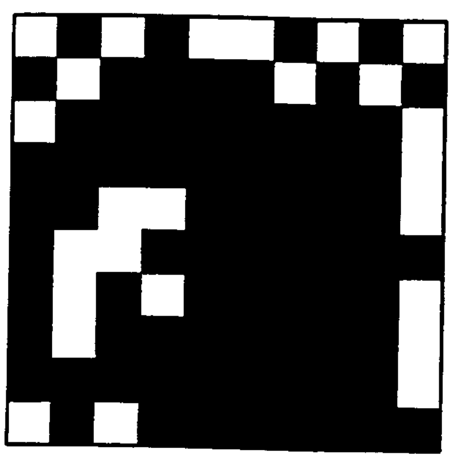
40%



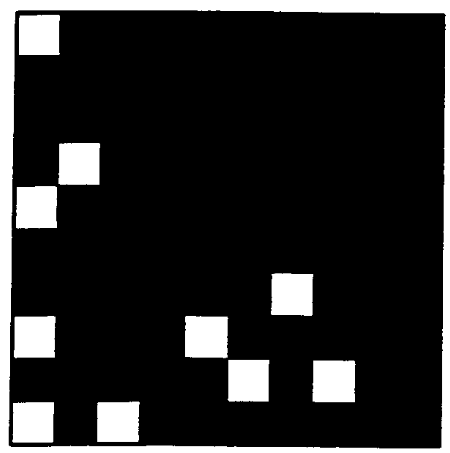
50%



60%



75%

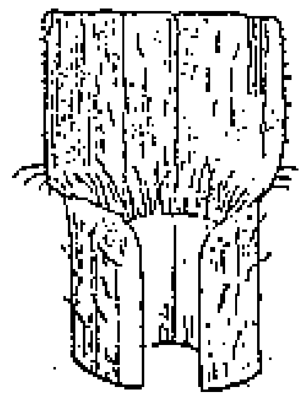


90%

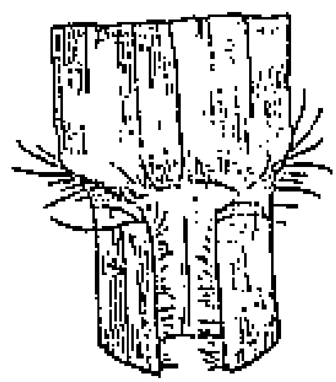
Identification of Common Weedy Grasses by Vegetative Characteristics

Grass	Vegetative Characteristics							
	Ligule			Sheath		Blade		
	None	Hairy	Membrane	Smooth	Hairy	Smooth	Hairy	Rough
Barnyardgrass	X			X				X
Large crabgrass			X		X		X	
Smooth crabgrass			X	X		X	at base	
Giant foxtail		X		X				X
Green foxtail		X		X				X
Yellow foxtail		X	X	X		X	at base	
Goosegrass			X		at top	X	at base	
Johnsongrass		X fused	X	X		X		
Fall panicum		at base		X		X		

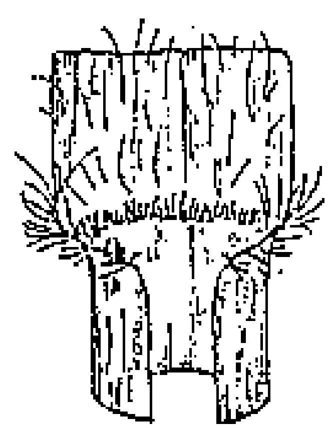
Note: These are the usual characteristics; however, there may be variations.



Giant Foxtail

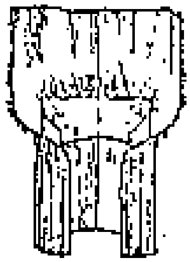


Green Foxtail

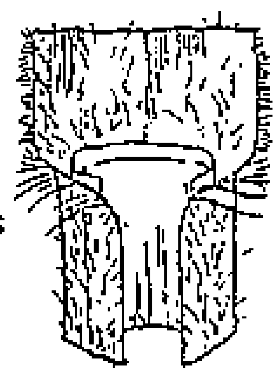


Yellow Foxtail

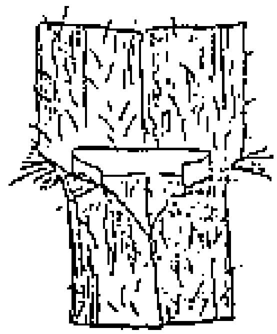
Wild Cane



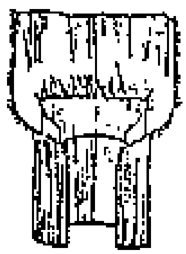
Large Crabgrass



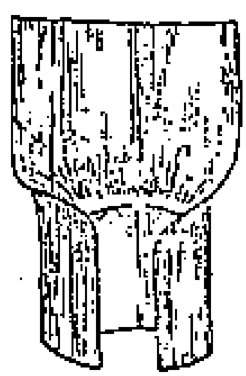
Smooth Crabgrass



Goosegrass



Johnsongrass



Fall Panicum

IDENTIFYING CHARACTERISTICS FOR CERTAIN SEEDLING BROADLEAF WEEDS

	Cotyledon	Leaf	Other
1. Common Chickweed	Small and thick Oval shaped Pointed tip	Oval shaped Pointed tip Opposite	
2. Cocklebur	Thick Long and Narrow	Oblong Toothed edges Alternate	
3. Burcucumber,	Thick Oblong	Somewhat lobed Alternate	Viney Stem
4. Eastern Black Nightshade	Small and Spoon shaped	Oval shaped Alternate	Lower surfaces of leaves often purple
5. Henbit	Round	Round shaped Toothed margins Deep crevices in surface Opposite	Square stem
6. Honeyvine Milkweed	Heart-shaped Opposite	Viney stem Long stem	
7. Hophornbeam Copperleaf	Oval shaped Toothed margins Opposite		
8. Jimsonweed	Thick Long and narrow	Heart-shaped with smooth edges near base and irregular edges at tip Alternate	Pungent odor
9. Lambsquarters	Small and Narrow	First 2 leaves are opposite and subsequent leaves are alternate	Leaves appear white, especially on underside
10. Morningglory Bigroot	Butterfly shaped with long narrow blades	Heart-shaped Hairless Alternate	Viney stem Established plants develop large perennial root
11. Morningglory, Entireleaf	Butterfly shaped	Heart-shaped Hairy Alternate	Viney stem

	Cotyledon	Leaf	Other
12. Morningglory, Ivyleaf	Butterfly shaped with prominent veins	3-lobed Hairy Alternate	Viney stem
13. Morningglory, Pitted	Butterfly shaped with long narrow blades	Shape is variable Hairless Alternate	Stem and leaf margin often purple Viney stem
14. Morningglory, Tall	Butterfly shaped with prominent veins	Heart shaped Alternate	Viney stem
15. Pigweed, Redroot	Narrow and about 1/4 inch in length	Oval Shaped Alternate	Taproot is red Stems are hairy
16. Prickly sida	Oval shaped 3 veins on upper surface	Oval shaped Toothed margins Alternate	2 to 3 spiny projections below each node
17. Ragweed, Common	Thick, spoon-shaped and small	Deeply divided Hairy Opposite	Emits a strong odor when crushed
18. Ragweed, Giant	Thick Spoon-shaped	Develop lobes with growth Opposite	
19. Shepherdspurse	Fleshy Small (2-3 mm) Round shaped	First leaves are round, other leaves are somewhat lobed	
20. Smartweed, Ladysthumb	Fleshy Narrow 3/4 inch long	Oblong and pointed Alternate	Membrane sheath at node is hairy
21. Smartweed, Pennsylvania	Fleshy Narrow 3/4 inch long	Oblong and pointed Alternate	Membrane sheath at node is hairy
22. Velvetleaf	Fleshy and oval shaped Small hairs	Pubescent on leaf and stem Alternate	Pungent odor

Alfalfa Diseases

Paul Vincelli

Alfalfa Disease Calendar for Kentucky

Disease	Observation Radius	Feb.	March	April	May	June	July	Aug	Sept	Oct	Nov
Sclerotinia Crown and stem rot	50'		***************								*****
Leaf Spots	10'			***********************							
Aphanomyces root rot	50'				**********						
Anthracnose	20'							***********			
Phytophthora root rot	100'			*****************************							
Bacterial wilt	100'			**********************************							
Crown and root rot Complex	100'			**********************************							
Problems, Other		***									

DESCRIPTION OF COMMON ALFALFA DISEASE

Sclerotina Crown and Stem Rot

Examination Period: Every two to four weeks from March l5 to June l. Examine any plants within a 50 foot radius that have symptoms described below.

Symptoms: Crowns and lower stems become soft, mushy and discolored. Leaves and stems may yellow and die. White aerial mold growth may be produced in light brown, soft mushy stems . Hard, black, small, round to irregular-shaped bodies (sclerotia) ranging from small to larger than pea size may be found on or embedded in lower stems and crowns. These black bodies have a white to silvery interior when cut. Patches of dead and dying plants appear as if they have been scalded.

Field infected with Sclerotinia crown and stem rot (top); sclerotia, which are initially white, beginning to form in plant tissue (left); hardened, black sclerotia laying on the soil surface (right); black sclerotia in plant tissue.

Occurrence: Symptoms are most common in early Spring, especially in no-till fields, areas of high moisture, wet soils, rank growth and poor air circulation occur. Damage can be extensive to fall-seeded stands in the spring following seeding, especially with extended snow cover and in no-till stands. Do not confuse with winter kill, which is common in Kentucky. Loss of fall-seeded stands in winter are more commonly due to Sclerotinia crown and stem rot.

Rating Scale:
0 = no affected plants;
l = only l to 2% plants showing typical symptoms;
2 = 2 to l5% of plants affected;
3 = more than l5% of plants affected.

Record: Record a rating of 0 to 3 for each sample site.

Leaf Spots

Examination Period: Every one to four weeks from May l through September. Examine leaves of plants within a l0 foot radius.

Symptoms: A wide variety of different leaf spots may be present. Leaf spots can be tan, brown, or black, circular to oblong, and may be raised above leaf. Smaller spots are scattered over entire surface of leaf; larger spots may be oblong, and show concentric rings of light to dark colors. Yellow areas may surround some spots. Leaves with several or many spots may turn yellow and fall to ground. Some foliar pathogens also infect stems and may produce similar spots there.

Lepto leaf spot

Occurrence: Moist weather conditions and dense stands favor all leaf spots. Certain leaf spots are favored by moderate to cool temperatures (Lepto Leaf Spot, Spring Black Stem) while others prefer warmer temperatures (Stemphylium leaf spot, Summer Black Stem). Leaf spots may occur soon after cutting (Lepto Leaf Spot) or after several weeks of growth (others) depending on the leaf spot and the environmental conditions

Rating Scale:
0 = no spots observed on leaves or stems;
l = a few lower leaves contain spots, but little or no defoliation;
2 = many leaves contain spots, some premature defoliation;
3 = leaves and stems peppered with spots, many leaves fallen on ground, all plants affected.

Record: Record a rating of 0 to 3 for each sample site.

Stemphyllium leaf spot

Spring black stem leaf spot

Common(Pseudopeziza) leaf spot

Action: If the field receives a rating of 2 or more, prompt cutting of the alfalfa will reduce loss in yield and quality from foliar disease. However, do not harvest fields that have not reached the mid-bud stage in order to allow sufficient time for replenishment of root reserves.

Aphanomyces Root Rot

Examination Period: Examine five sites at least 10 - 20 feet from field edge, especially in wetter areas of the field.

Symptoms: Seedlings affected by this syndrome exhibit severe stunting of internodes, petioles, and leaf blades. The seed leaves (cotyledons) commonly are yellow, and may exhibit reddening, especially on the underside. Leaf blades exhibit a bluish-green cast. Seedlings typically do not wilt or collapse, as they might from a damping-off disease. Roots of affected plants exhibit light brown discoloration. Commonly, the problem affects most or all of the field, although it may be worse in more poorly drained areas.

Occurrence: This disease is a common stand-establishment problem when spring-seeded alfalfa is followed by extended periods of wet weather, especially within 4-6 weeks of seeding. Disease pressure sometimes is higher in no-till fields, although severe outbreaks can occur in conventional fields. About two thirds of Kentucky alfalfa fields are infested with the pathogen. If disease pressure is severe, the disease can attack alfalfa varieties with some resistance, although these usually outgrow the symptoms better than susceptible varieties.

Rating Scale:
0 = no plants diseased;
1 = less than 25% of plants in the site have modest stunting and yellowing;
2 = over 25% of plants are showing symptoms; some scattered plants may show severe stunting;
3 = over half of the plants at the site are extremely stunted (less than two inches tall) six weeks after seeding.

Record: A rating of 0 to 3 for each sample site.

Phytophthora Root Rot

Examination Period: Every four weeks from April l5 until September l5. Examine any suspicious looking plants within l00 feet of monitoring site.

Symptoms: Stands are stunted and thin in Phytophthora affected areas. Leaves on severely affected plants turn yellow or tan and often drop early. Regrowth is slow after harvest. Dig (not pull) surviving plants and look for reddish-brown rotted areas on roots that may extend to the crown . Affected areas of roots later turn black in color. Severely affected tap roots become rotted off. Affected plants sometimes show reddening typical of phosphorus deficiency.


Phytophthora root rot

Occurrence: Cool, wet periods in heavy, poorly drained soils are most favorable for Phytophthora development. Excessive rainfall or soil compaction can increase the root rot problem. Look in waterlogged areas for the disease. Disease can cause great seedling loss.

Rating Scale:
0 = no infected plants, good healthy stand;
l = one plant has symptoms, appearance of some missing plants in stand;
2 = from 2 to l0% of plants infected, several; areas where stand slightly thinned;
3 = more than l0% of plants infected, severe thinning of stands.

Record: Record a rating of 0 to 3 for each sample site.

Anthracnose

Examination Period: Every four weeks in late summer and fall (August l to October l). Examine plants within a 20 foot radius.

Symptoms: Light to dark brown diamond-shaped lesions with dark borders form on lower parts of stem. Small, black bodies of the fungus may sometimes be observed in the center of lesions. A "shepherd's crook" may develop on young dead shoots (see Plate 3, Alfalfa Diseases II picture sheet). As disease development progresses, the fungus may girdle and kill stems, crown buds and the crown. Dead, straw-colored stems become scattered throughout the field and a thinning of the stand becomes evident. Look for a single stem killed as well as entire crown. Crown infections are characterized by a dry rot with blue-black coloration of the internal tissues and lower stems.

Occurrence: Hot, moist weather conditions in late summer and fall are favorable for anthracnose development. Dense, rank stands reduce air circulation and provide conditions favorable for infection. The crown rot phase of this disease is often serious in hot, dry weather.

Rating Scale:
0 = no symptoms on stems or crowns of any plants;
l = a few lesions found on some stems of a few plants, but no crown infections, little or no death of plants;
2 = lesions found on a number of plants, some dead, straw-colored stems scattered throughout the area;
3 = lesions observed on most stems of most plants, numerous dead stems found, thinning of stand apparent with anthracnose symptoms associated with crowns of dead of dying plants. (If entire crowns are dead, indicate as such.)

Record: Record a rating of 0 to 3 for each sample site.

Bacterial Wilt

Examination Period: Every four weeks from April l to October l. Examine any suspicious looking plants within l00 feet of monitoring site.

Symptoms: Affected plants first appear stunted with slow regrowth after cutting. Leaves turn yellow at margin first, advancing over the whole stem or plant. Best diagnostic symptoms occur in the tap roots. Dig (not pull) suspicious plants and look for yellowish-tan discoloration of the vascular tissue. Overall the internal root tissues are yellow compared to white in healthy tap roots._

Bacterial wilt

Occurrence: Most probable in older stands; seldom seen in seedling year. Affected plants are usually scattered, not clustered. Not particularly common in recent years.

Rating Scale:
0 = no infected plants, good healthy stand;
1 = a few plants have suspicious symptoms, appearance of some stunted or missing plants in stand;
2 = from 2 to l0% of plants infected;
3 = more than l0% of the plants infected, with thinning of the stand evident.

Record: Record a rating of 0 to 3 for each sample site.

Crown and Stem Rot Complex

Mycoleptodiscus crown rot

Examination Period: Every four weeks from April l to October l. Examine suspicious plants within l00 feet of monitoring site.

Symptoms: Infected plants are stunted with bleached, yellow leaves that wilt under moisture stress. Reddish to black rotted areas develop anywhere in the crown and upper tap root. Generally rot extends from the crown toward the roots. Sometimes the crown is hollowed. The rot develops slowly and seldom kills plants in a single crop season.

Occurrence: Some root and crown rot complex can be found in most alfalfa plantings. Most damage is confined to aging stands, or stands weakened by other biological, environmental or cultural stresses. Weeds easily invade areas with the disease. Fields pastured or harvested while wet often have a higher incidence of the disease.

Rating Scale:
0 = no infected plants, good healthy stand;
l = one plant has suspicious symptoms, appearance of some missing plants in a stand;
2 = from 2 to l0% of plants infected, several areas where stand slightly thinned;
3 = more than l0% of plants infected, severe thinning of stand.

Record: Record a rating of 0 to 3 for each sample site.

Sources of additional information:

Kentucky Plant Disease Management Guide for Forage Legumes
http://www2.ca.uky.edu/agcollege/plantpathology/ext_files/PPFShtml/PPFS-AG-F-08.pdf

Alfalfa Diseases Caused by Rhizoctonia Fungi (PPFS-AG-F-06)
http://www2.ca.uky.edu/agcollege/plantpathology/ext_files/PPFShtml/PPFS-AG-F-6.pdf

Crown Rots of Alfalfa (PPFS-AG-F-05)
http://www2.ca.uky.edu/agcollege/plantpathology/ext_files/PPFShtml/PPFS-AG-F-05.pdf

Risk Factors for Sclortinia Crown and Stem Rot in Fall-seeded Alfalfa (PPFS-AG-F-12)
http://www2.ca.uky.edu/agcollege/plantpathology/ext_files/PPFShtml/PPFS-AG-F-2.pdf

UK Forage Variety Trials
http://www.uky.edu/Ag/Forage/ForageVarietyTrials2.htm

SOIL SAMPLING AND SOIL TESTING

Lloyd Murdock

The most important factor of soil testing and fertility recommendations is obtaining a good soil sample. There is more room for error in this step than any other in getting reliable soil test results and recommendations.

Method:

The proper procedures for obtaining a good soil sample are well-established. Publication AGR-16 contains a complete explanation of these procedures.

Time of Sampling:

Recent fertilizer applications can distort the soil test results and fertilizer recommendations. To ensure proper sampling, soil samples should be taken at least six weeks after the last fertilizer application. The best time to sample is in the late winter or spring for that years fertilization. This gives all the nutrients in the soil and those released by the plant a chance to equilibrate. Fall sampling is also good but needs attention paid to sampling location to ensure a more representative sample in row crops. An equal number of samples should be taken from the row middle and from next to the row.

Corn presents a special problem if the soil is sampled when the crop is growing. As the height of the plants reach three feet it becomes increasingly difficult to walk through the field and view the terrain for any areas that need separate sampling. In the process of early season scouting notes should be recorded and areas needing special attention should be out lined on the field map. The best overall suggestion when special notes are lacking is to wait at least six weeks but sample while the general crop condition and field are still visible.

Late application of nitrogen to corn and the use of anhydrous ammonia require some special attention. When nitrogen is applied four to six weeks after planting, the best time to sample would be just before the delayed application. When anhydrous ammonia is applied between the row anytime after planting, do not sample within six inches of the application slit.

Soil cores should not be taken from where fertilizers were banded in or beside the row. If it is not know where the fertilizer band is located, then no soil cores should obtained within 4 to 6 inches of corn rows if row fertilizer was used.

Soil samples for alfalfa, small grains and soybeans can be taken anytime in summer or fall as long as it is at least six weeks after the last fertilizer application. It is always best to sample after the final harvest for the year or before spring planting or re-growth begins. In a double-cropping system (small grains and soybeans), sampling a small grain can be accomplished early so that recommendations for the second crop can be obtained.

Soil Sampling Depth:

Crop	Depth of Sample
Alfalfa and pastures	4"
No-till corn or soybeans	4"
Conventional corn or soybeans	6-8"

Identifying Compacted Soil

Lloyd Murdock

Most compaction results from the use of machinery on soil which is too wet to work well, or from overworking soil and destroying its natural structure. Pressure from tires and tillage tools compress more soil into a given volume. In the process, the natural soil aggregates are broken down and large pores become smaller. This generally causes the soil to be more difficult for plant roots to penetrate.

A soil penetrometer, tiling rod or a three foot length of 3/8-inch diameter steel rod sharpened on one end and having a handle welded to the other end are easy tools to use in identifying compacted layers. Such tools should be marked in six inch increments and should uniformly be pushed into the soil when the moisture content is too wet for tillage. Under these conditions, compacted layers can be "felt" due to resistance in pushing the rod through the soil, and depth to and thickness of the compacted zone can be identified.

The best method for identifying soil compaction is with a soil penetrometer. This is similar to a tiling rod but has a gauge that measures the amount of pressure required to push the rod into the soil. An Annual Field Compaction Record Sheet is on page 43 and gives instructions on how to use the penetrometer and how to make a field recording.

Regardless of the method used, a number of sites in each field should be checked (similar to a soil test) and if severe compaction is found it needs to be confirmed. In addition to a compacted soil, the penetrometer will give high readings for a dry soil and heavy clay layer. Therefore, if severe compaction is found in a field then a soil probe or shovel needs to be used to look at the layer that was found compacted and confirm that high readings were not due to a clay or dry layer.

ANNUAL FIELD COMPACTION RECORD
University of Kentucky
Department of Agronomy

FARM____________________ FIELD_____________ ACRES_______________

MAJOR SOIL TYPE _________________________ YEAR___________________

SITE	READING	Depth of Highest Reading
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		
11		
12		
13		
14		
15		
16		
17		
18		
19		
20		

Site	Reading	Depth of Highest Reading
21		
22		
23		
24		
25		
26		
27		
28		
29		
30		
31		
32		
33		
34		
35		
36		
37		
38		
39		
40		

SUMMARY

% of reading 200 or less _________

% of reading 300 or more ________

Most common depth of readings 300 or more _______

METHOD

Push penetrometer into the soil slowly (do not surge). Note the highest psi reading and the depth at which it occurs. Continue to push until the resistance drops and note the depth where this happens.

Always use the penetrometer when the soil is too wet for proper tillage and when it is not saturated with water. Avoid or test separately field entrances and turn row areas that have excessive traffic and do not represent the field. Readings should be taken in a random manner over the rest of the field.

INTERPRETATION

With readings of 300 psi or above, the compaction is considered severe. If 1/3 of the readings are 300 or more, a corrective action and change in tillage practices should be considered. When 1/2 of the field readings are 300 or more, then changes definitely need to be made. If severe compaction is identified in only a portion of the field, then corrective action should only be considered in that portion.

EXAMPLE RECORD

Site	Reading	Depth of Highest Reading
1	175	6-12
2	200	9-15
3	300	9-15
4	175	6-12
5	300+	9-15
6	225	9-15
7	200	6-12
8	300+	6-15
9	150	3-18
10	250	9-15

SUMMARY

% of readings 200 or less 50

Most common depth of readings 300 or more 9 - 15.

% of readings 300 or more 30

ALFALFA STAND COUNTS

Equipment Needed:

1. Two x two foot sampling quadrant
2. Farmer Report Forms
3. Pencil and paper.

Sampling Procedure:

1. Enter the field and move away from the edge about 20 paces.
2. Drop the sampling quadrant in a randomly selected location.
3. Count all of the alfalfa crowns found within the sampling quadrant. Record this figure on separate sheet of paper. Also, separately count the number of stems/quadrant.
4. Repeat this procedure in five locations in each alfalfa field that is less than 100 acres. If the field is over 100 acres add one sample location for every additional 20 acres.
5. Calculate the number of crowns per four square feet by adding up the number of crown in each location and dividing by the number of locations sampled. Repeat the procedure for stems/four sq ft.
6. Record this figure on your Farmer Report Form as the number of crowns per four square feet, repeat for number of stems per four sq ft.
7. Perform this procedure each spring and fall.