APPENDIX A – Monitoring Protocols

Cabbage Seedpod Weevil - Monitoring Protocol

<u>Host plants:</u>
Plants belong to the family Brassicaceae and include canola, mustard, broccoli, cauliflower and weeds such as wild mustard, flixweed and stinkweed.

<u>Identification, Life cycle and Damage</u>:

Adult: Adults overwinter in soil beneath leaf litter in shelter belts and roadside ditches and emerge from these sites in spring when soil temperatutes warm to approximately 15ºC. Adult weevils are ash-grey and approximately 3 to 4 mm long (Figure 1). They have a prominent curved snout that is typical of most weevils. Adults can be found on early flowering hosts (wild mustard, flixweed, hoary cress, stinkweed, and volunteer canola). **Weevils move to canola fields when the crop is in the bud to early flower stage** and feed on pollen and buds, causing the flowers to die. Yield loss due to this feeding is more evident in dry years when the canola crop can't compensate for the loss. After a pollen meal, mating occurs on the plant. When small pods develop, the female will deposit an egg through the pod wall onto, or adjacent to a developing seed.

Eggs: Eggs are very small, oval, and opaque white (Figure 2). Most often, only a single egg is deposited per pod; however, two or more eggs can be laid per pod when cabbage seedpod weevil densities are high.

Figure: 1- Adult- 16 days

Figure 2: Eggs: 6-7 days

Larva: Larvae are white and grub-like and consist of four larval instars. They can reach 5 to 6 mm in length (figure 3). The first instar larva feeds on the cuticle on the outside of the pod. The second instar bores into the pod and feeds on the developing seeds. A single larva consumes about five canola seeds. Larval feeding on the seeds is the most severe type of injury and infested pods are more prone to shattering that causes seeds to be un-harvestable. Infested pods are often misshapen as a result of the larval feeding. An indirect form of damage can occur when fungus enters the pod through the larval exit hole and infects the pod.

Pupa: Pupation takes place in the soil ((Figure 4). Mature larvae chew small, circular exit holes in the pod walls (Figure 5), drop to the soil surface, burrow in, and pupate within earthen cells. New generation adults emerge about 10 days later and feed on maturing canola pods causing further losses and crop quality. Late in the season, they migrate to overwintering sites.

Figure 3: Larva- 6 weeks

Figure 4: Pupa: 10 days

Figure 5: Exit holes on canola pods

Monitoring

Timing: The risk of infestation can be predicted based on the size of the adult population. **Begin sampling when the crop first enters the bud stage and continue through the flowering.**

Sweep net sampling: Sweep net samples should be taken at ten locations within the field with ten 180° sweeps per location. Count the number of weevils at each location**. Samples should be taken in the field perimeter as well as throughout the field. Adults will invade fields from the margins and if infestations are high in the borders,** application of an insecticide to the field margins may be effective in reducing the population to levels below which economic injury will occur. **An insecticide application is recommended when three to four weevils per sweep are collected**. Timing of the insecticide when the crop is in the 10 to 20% bloom stage (2-4 days after flowering starts) has been shown to be the most effective. Consider making insecticide applications late in the day to reduce the impact on pollinators,

A high number of adults in the fall may indicate the potential for economic infestations the following spring. A trap crop border of an early maturing *Brassica* (e.g. *B. rapa*) can be planted seven to ten days prior to the canola crop to attract and concentrate adult weevils in the field borders. The trap crop should be monitored for the presence of weevils and when weevil populations are high an insecticide can be applied to the trap crop prior to bud formation.

An additional benefit to monitoring for cabbage seedpod weevil is that *Lygus* bugs can be sampled, provided the sampling continues thorough the early pod stage.

Monitoring Protocol for Diamondback Moth

Purpose of Monitoring Program

Diamondback moths can be serious pests of canola and cruciferous vegetables. It is the larval stage that is damaging to the plants, however the adult stage appears before the larvae are present. This provides an opportunity to assess in advance the risk of a potential outbreak.

The pheromones used by female diamondback moths to attract male moths are known and are synthesized for use in monitoring programs. These synthetic pheromone lures are very effective at attracting male moths to traps, which have been designed to capture the moths. Use of these traps provides a relatively simple and effective method of detecting an influx of the moth in advance of the damaging larval stage of diamondback moth. This gives farmers, agronomists, chemical dealers, and others involved in pest management some advanced warning of a potential outbreak, and enables them to place proper emphasis on monitoring for the larvae.

At no time should a decision to use insecticides to control diamondback moth be made based only on information from the traps for adults of this insect. Such decisions need to be made after later sampling for the damaging (larval) stages of diamondback moth and determining if the levels of larvae present in the field are above the economic threshold. Weather can affect the success of mating and laying eggs, and many mortality factors could reduce the numbers of eggs and larvae before they develop to the damaging stage.

When to Monitor

Diamondback moth do not overwinter in the Canadian prairies in significant numbers, but high populations may be blown in on winds from the south. They are at a higher risk of being a problem in years when high populations blow in and establish early (May or early June). Thus the primary goal of this monitoring program is to determine when diamondback moth populations arrive, and in what number. This, combined with considering what the environmental conditions are like when they arrive, will determine the risk of them being a significant problem.

Diamondback moth traps should be placed in fields during the first week in May, or as close to this date as possible, and first moth counts should be made the following week.

Traps for diamondback moth can be placed in fields regardless of whether canola has, or will be, seeded into the field. Having traps near fields that are or will be seeded to canola is ideal, but it is more important to place the traps at the proper time than to wait and see where canola is seeded.

Assembling the Traps

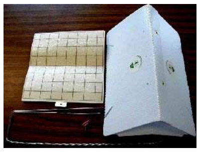

Figure 1
Trap components

Figure 2
Inserting wire

Figure 3
Pheromone placed in trap

Figure 4
Sticky bottom placed in trap

1. Insert the wires through the holes in the top of the trap as in **Figure 2**. Bend the wires to secure them to the trap.
2. Insert the T-shaped plastic attachment on the lure (rubber stopper containing pheromone) through the middle hole in the top of the trap (**Figure 3**). The rubber stopper containing the pheromone should hang from the top of the trap. Remember not to touch the pheromone with your hands.
3. Unfold the sticky insert and place in the bottom of the trap (**Figure 4**).
4. Press in the front and back flaps until they clip into the slits in the sides of the trap (**Figure 5**). This will secure the sticky bottom in place.

Figure 5
Trap with front and back flaps closed

Figure 6
Completed trap set-up

5. T bars can be inserted into hollow metal poles, which are hammered into to ground, so there is a horizontal surface to hang the traps from.
6. The wire that extends from the top of the diamondback moth trap can be wrapped around one of the horizontal ends of the T-shaped bar to secure the trap in place (**Figure 6**).

Trap Height

Traps should either be positioned at 50cm above the ground or moved with the crop canopy. Research has shown that traps positioned at 50cm or moved with the crop canopy attracted more moths than those placed higher (1.5m).

Tips for Setting Up the Traps

If more than one trap is being placed in a field, leave at least 100 meters between traps.

Do not handle the lures with your bare hands. Please wear rubber or latex gloves if handling the lures.

Checking the Traps

Traps should be checked and the number of diamondback moths counted once a week over a six-week period. If a late influx of diamondback moths appear, the diamondback moth traps may be left out for a period longer than six weeks. The pheromone lures for diamondback moth can remain attractive for at least 8 weeks.

If there are only a few moths on the insert of the diamondback moth traps when they are checked, these moths can be removed from the sticky surface with tweezers and the insert reused. If the insert is heavily covered with insects, it should be replaced with a new insert.

Bertha Armyworm: *Mamestra configurata* Monitoring Protocol

The purpose of this monitoring program is to determine the regional risk of an outbreak in advance before the appearance of the damaging (larval) stage. This gives farmers, farm production advisors, agronomists, chemical dealers, and others involved in pest management some advanced warning of a potential outbreak, and enables them to place proper emphasis on monitoring for the larvae. It also provides time for agriculture retailers to have the appropriate insecticides in place.

At no time should a decision to use insecticides to control bertha armyworm be made based only on information from the traps for adults of this insect. Such decisions need to be made after later sampling for the damaging (larval) stages of bertha armyworm and determining if the levels of larvae present in the field are above the economic threshold. Weather can affect the success of mating and laying eggs, and many mortality factors could reduce the numbers of eggs and larvae before they develop to the damaging stage.

Host Plants:

Bertha armyworm is a general feeder on broadleafed plants, and has been especially harmful to canola and flax. They also feed on a range of secondary hosts including peas and potato.

Identification, Life Cycle and Damage:

Adult: The adult moths (Figure 1) begin emerging from the overwintering pupae in **early to mid-June and continue until early August**. The moth has a wing span of about 4 cm., and is active only at night. The forewing is predominantly gray, and flecked with patches of black, brown, olive and white scales. There is a prominent, white, kidney-shaped marking near the middle of the forewing, towards the wing margin. White and olive-colored fringe on the forewing is also a characteristic of the species.

Eggs: Females lay eggs with the size of a pinhead, usually on the underside of the leaves and these are deposited in single layer clusters of 50 to 500 (Figure 2). When first laid, they are white but become darker as they develop.

Figure 1: Adults- 1-5 days

Figure 2: Eggs- 7 days

Larva: Newly emerged larvae are 0.3 cm long, pale green with pale yellowish stripe along each side. The larva has six instar stages and passes through color phases of green and pale brown before becoming large black caterpillars (Figure 3). Mature larvae are 4-5 cm long with a light brown head and a broad pale orange stripe along each side. However, some larvae remain green or pale brown throughout their larval life. Large larvae may drop off the plants and curl up when disturbed, a defensive behavior typical of cutworms and armyworms. Young larvae chew irregular holes in leaves, but normally cause little damage. The fifth and sixth instars cause the most damage by defoliation and seed pod consumption. Crop losses due to pod feeding will be most severe if there are few leaves. Larvae eat the outer green layer of the stems and pods exposing the white tissue. At maturity, in late summer or early fall, larvae burrow into the ground and form pupae.

Pupa:
Bertha armyworms survive the winter as pupae in the ground at depths of 5 to 16 cm. Pupa is reddish brown and about 1.8 in size (Figure 4).

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Figure 3: Larvae- 6 weeks

Figure 4: Pupa- overwinter

<u>Monitoring</u>

<u>Pheromone Traps for Adult Monitoring- Unitrap</u>:
Adult bertha armyworm moths are monitored using pheromone-baited traps that attract male moths. The number of adults collected by these traps provides an indication of the risk of larval damage. We use the all-green model of the unitrap.
Timing:
First week in June – last week in July. The traps should be checked and the number of moths should be counted once a week.

Site Selection:
Select a canola field in your area. Use two traps per field. The trap should not be placed next to a shelterbelt, steep ditch or within ½ kilometer of a strong light source (such as a farm yard light). **The traps should be located 2 m from the field edge and a minimum of 50 m apart.**

Assembly and Trap Set Up:

1. Place vapona insecticidal strip inside the bucket of the trap, so that the moths are killed when they fly in (Figure 1). This is an insecticide and should not be handled with bare hands..
2. For new-model traps, there are four holes drilled in the barrel part of the trap. Two can be used for attaching the trap to the stand, so that it does not swing in the wind.
3. Place the lure into the lure basket, and insert the cap. Do not handle the rubber stopper with your hands. Oil from your skin can reduce the effectiveness of the lure. Lures should be stored in sealed containers at temperatures below 0℃ until you are ready to use t hem. Store only lures for one type of insect per container. Wear disposable gloves when handling lures and use a new pair of gloves between handling lures of different types. This will avoid cross-contamination of the pheromones and possible interferences with their attractiveness.
4. **Mount the traps at about three feet off the ground on a sturdy stake**. The trap is hung directly by a wire hangar from the “L” shaped metal post supplied (Figure 4a) or it can be attached to a stand (Figure 4b)

Figure: 1

Figure: 2

Figure: 3 **Figure**: 4a **Figure**: 4b

Larval monitoring:

Generally, **higher moth numbers during mid June-July indicate greater risk of larval damage in July and August**. Larval sampling should commence **once the adult moths are noted**. Sample at least three locations, a minimum of 50 m apart. At each location, mark an area of 1 m^2 and beat the plants growing within that area to dislodge the larvae. Count them.

Data from monitoring adults with pheromone lures indicates regions where more intensive larval monitoring is needed (as indicated in the Table). By about mid-July, the monitoring data should indicate whether there is a risk of larval infestation. Data from larval monitoring is expected to influence pest control decisions.

Table: Cumulative moth counts

Cumulative moth catch	**Risk Level**	**Interpretation**
0 to 300	Low	Infestations are unlikely to be widespread, but fields should be inspected for signs of insects or damage.
300 to 900	Uncertain	Infestations may not be widespread, but fields that were particularly attractive to egg-laying females could be infested. Check your fields.
900 to 1200	Moderate	Infestations likely, canola fields should be sampled regularly for larvae and for evidence of damage.
1200+	High	Infestations very likely, canola fields should be sampled frequently for larvae and for evidence of damage.

Swede Midge
Monitoring Protocol

Host Plants: Plants belong to the family Brassicaceae such as canola, mustard, cabbage, cauliflower and Brassica weeds.

Identification, Life Cycle and Damage:

Adults: Adults are small (1.5-2 mm in size), light brown flies. They have long filiform antennae (Figure 1). Male antennae have 12 flagellomeres; each is divided into two separate nodes surrounded by a looped sensillum. Female antennal segments are cylindrical. Wing venation is reduced. Cross veins are absent, radial vein is straight or nearly so and cubital fork is present. **Adults appear in the spring from pupae which have spent the winter in the soil.** Eggs are laid in clusters of about 2-50 on the growing point of the plant.

Eggs: Eggs are very small (0.3 mm), transparent in color when first laid, but change to creamy white color as they mature (Figure 2).

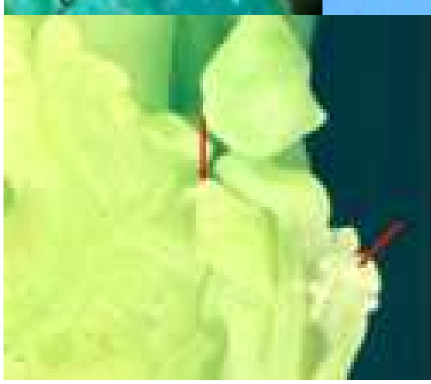


Figure 1: Adults- 1-5 days

Figure 2: Eggs- 3 days

Figure 3: Larva- 14 days **Figure 4:** Pupa-14 days

Larvae: Small maggots can be found feeding in groups near the growing points of the plant. Initially the larva is 0.3 mm in length and transparent. At maturity, it is 3-4 mm in length and lemon yellow in color (Figure 3). Larva spins a cocoon and pupate in the ground

Pupae: Pupation takes place approximately 2" deep in soil near the host plant. Prepupae can go into a state of diapause, overwintering in silken ovoid cocoons in the soil and pupating in the following spring (Figure 4). However, some pupae may overwinter a second season before becoming adults. Larvae and pupae require moist environments to mature. In Ontario, 3-4 overlapping generations have been reported.

Monitoring

Site Selection:

Swede midge adults are not strong fliers and prefer areas of low wind movement, resulting in more damage in sheltered areas, along field edges and buildings.
Because swede midge may be moved in soil, care must be taken when working within a block/field. To prevent the inadvertent movement of swede midge, footwear must be cleaned thoroughly before moving to a new address. Disposable "booties" or rubber boots are recommended.

Pheromone Traps for Adult Monitoring:

Timing: The first emergence of adults in the province of Ontario occurs in mid May.
Traps should be set by May 15 and retrieved just prior to crop harvest**.**

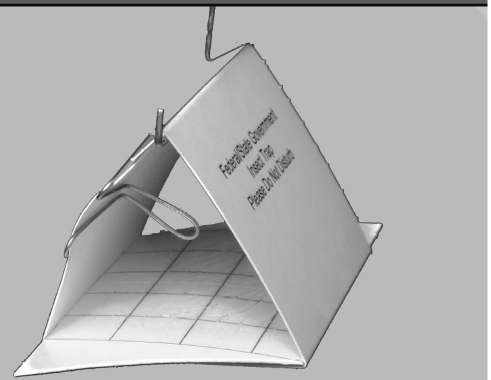
Two (2) traps **will be placed at each field.** Place traps in locations that gives you best coverage, incorporates microhabitats such as those adjacent to shelterbelts and areas of higher humidity, **and has good accessibility. Label all traps using a "wax pencil" and / or lead pencil. Do not use a marker or any other writing device that will emit a volatile chemical. These chemicals may interfere with the pheromone.**

Trap: "Jackson Trap" with removable sticky liners and lures containing swede midge sex pheromone. Trap assembly instructions are given below. All lures should be stored in sealed containers at temperatures below 0℃. Store only one type of lure per container. Wear disposable gloves when handling lures and use a new pair of gloves between handling lures of different types. This will avoid cross-contamination of the pheromones and possible interference with their attractiveness.

Trap Placement: The stake or rod height should be above the predicted final height of the crop and should be flagged to allow easy detection in the field for the inspector and grower. When placed in a field, the **trap should always be within the crop canopy,** therefore, as the crop height increases during the growing period, adjust the trap height accordingly during each servicing. The traps must be attached to the stake or rod in such a manner to keep it from twisting or moving. The trap placed within the field must be orientated in such a way so that the pheromone plume is dispersed down the row. This will maximize its effectiveness. **Note that swede midge adults prefer to fly within the canopy of the crop or just above it**.

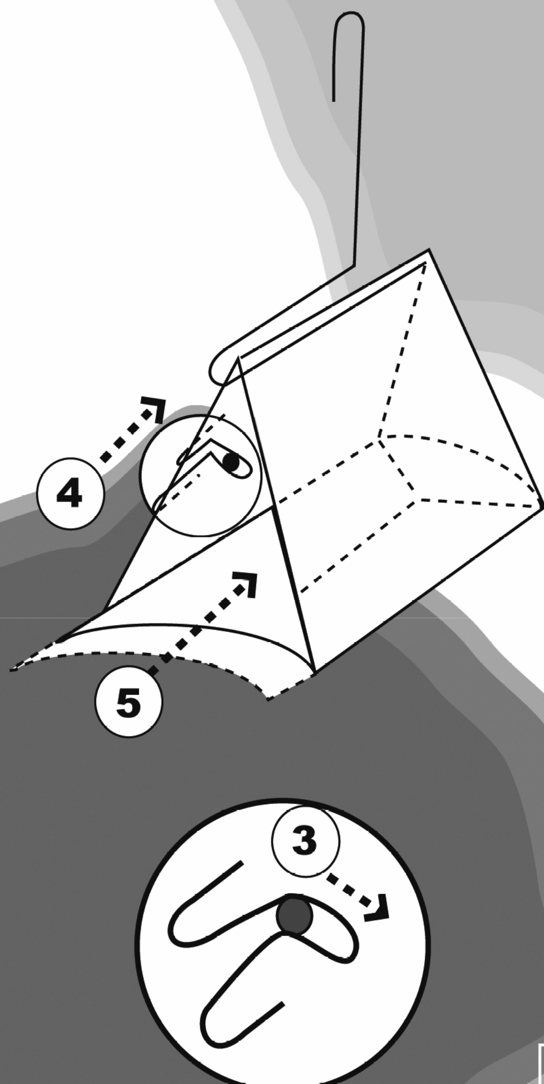
Due to the small size of the trap and the dusty/dirty conditions of the fields the traps have to be checked every week during the growing season. Sticky liners covered in field debris make it almost impossible to see the very tiny swede midge. During the weekly visit to the traps remove and replace the sticky liner. **Replace the lure after 28 days.** Replace damaged or missing traps as necessary. Make sure you replace traps when they become compromised or when badly weathered. Ensure the entrance to the trap remains clear and open.

JACKSON TRAP ASSEMBLY INSTRUCTIONS



Components:

1 Jackson trap
Sticky insert (packed in pairs)
1 wire hanger
1 dispenser holder



How to assemble:

1 Fold open Jackson trap to form a tent shape.
2 Remove lure from foil pack (Avoid touching dispenser surface with fingers.).
3 Insert plastic or rubber dispenser only into the middle notch of the dispenser holder.
4 Slide dispenser holder onto side of trap so that the lure is on the inside and the two legs are on the outside.
5 Peel apart face-to-face sticky liners and slide one insert into trap.
6 Attach metal hanger as illustrated and hang trap in desired location.
7 Replace inserts as required.

NOTE TO USER:
To prolong storage life, refrigerate or freeze unused dispensers in their foil packets.
To avoid contamination, use twee-zers or rubber gloves when handling dispensers.

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Trap

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Instructions for Shipping Sticky Inserts: Ship sticky inserts every 28 days (when you change the lures) for identification of the swede midge. Fold each trap carefully with the sticky surface inside; hold it with a rubber band (Figure 5). Place them in a box as shown in Figure 6 and ship to Dr. Julie Soroka, AAFC-AAC (address is in the data recording sheet).

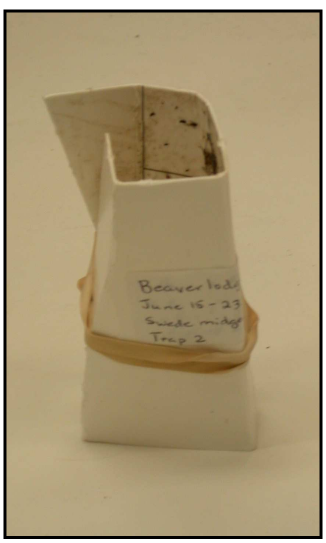


Figure 5: Folding **Figure 6: Packing**

<u>Checking the Crop Damage:</u>

Damage caused by feeding of swede midge larvae results in changes in the physiology of the plant. The growing tip may become distorted and produce several growing tips or none at all, young leaves may become swollen, crinkled or crumpled and brown scarring caused by larval feeding may be seen on the leaf petioles and stems. **Young plants that show unusual growth habits should be examined carefully for damage and larvae. Larvae can be seen with a hand lens.**

Cereal Leaf Beetle - Monitoring Protocol

Host Plants: All cereals, grains and various grasses including; barley, wheat, oats, rye, sorghum, timothy, ryegrass, foxtail grass and bluegrass.

Identification, Life Cycle and Damage:

Adult: Adult cereal leaf beetles (CLB) have shiny bluish-black wing-covers (Figure 1). The thorax and legs are light orange-brown. Females (4.9 to 5.5 mm) are slightly larger than the males (4.4 to 5 mm). Adult beetles overwinter in and along the margins of grain fields in protected places such as in straw stubble, under crop and leaf litter, and in the crevices of tree bark. They favour sites adjacent to shelterbelts, deciduous and conifer forests. They emerge in the spring once temperature reaches 10-5ºC and are active for about 6 weeks. They usually begin feeding on grasses, then move into winter cereals and later into spring cereals.

Egg: Egg laying begins about 14 days after the emergence of the adults. Eggs are laid singly or in pairs along the mid vein on the upper side of the leaf and are cylindrical, measuring 0.9 mm by 0.4 mm, and yellowish in colour. Eggs darken to black just before hatching.

Larva: The larvae hatch in about 5 days and feed for about 3 weeks, passing through 4 growth stages (instars). The head and legs are brownish-black; the body is yellowish. Larvae are usually covered with a secretion of mucus and fecal material, giving them a shiny black, wet appearance (Figure 2). When the larva completes its growth, it drops to the ground and pupates in the soil.

Pupa: Pupal colour varies from a bright yellow when it is first formed, to the colour of the adult just before emergence. The pupal stage lasts 2 - 3 weeks. Adult beetles emerge and feed for a couple of weeks before seeking overwintering sites. There is one generation per year.

Figure 1: Adult **Figure 2**: Larva

Figure 3: Cereal leaf beetle damage to a wheat leaf

Monitoring

Timing:

During the later part of the oviposition period, both adults and larvae can be found in the field at the same time. Depending on the local conditions this is generally from **mid-June to early July.** Recommended time for winter wheat is **mid May to mid June.**

Location:
Give priority to following factors when selecting monitoring sites:

- Choose fields and sections of the fields with past or present damage symptoms.
- Choose fields that are well irrigated (leaves are dark green in color), including young, lush crops. Areas of a field that are under stress and not as lush (yellow) are less likely to support CLB. Monitor fields that are located along riparian corridors, roads and railroads for human aided dispersal**.** Also include farms and hay facilities for this purpose.
- Survey the sides of a field that are close to brush cover or weeds, easy to access, with a sheltered area nearby: hedge rows, forest edges, fence lines, etc.

Focus your site selection on the following host priorities:

- **First** - winter wheat. If no winter wheat is present then;
- **Second** - other cereal crops (barley, wheat, oats, and rye). If no cereal crops are present then;
- **Third** - hay crops. If no hay crops or cereal crops are present then;
- **Fourth** - ditches and water corridors

Sweep net Sampling for Adults and Larvae:

A sweep is defined as a one pass (from left to right executing a full 180 degrees) through the upper foliage of the crop with a 37.5 cm diameter sweep net. A sample is defined as 100 sweeps taken at a moderate walking pace, 4-5 meters inside the border of a field.

Four samples are taken from each site, totalling 400 sweeps per site. The contents of each sample are visually inspected for life stages of CLB and all suspect specimens are to be retained for identification. Please note that because the CLB larvae are covered in a sticky secretion when they are caught in a sweep net they are often covered in debris and are very difficult to see. To help determine the presence of CLB place the contents of the sweep net into a large ziploc bag for observation.

Visual Inspection:

Both the adults and larvae severely damage plants by chewing out long strips of tissue between the veins of leaves, leaving only a thin membrane. When damage is extensive the leaves turn whitish (Figure 3). The plant may be killed or the crop may be seriously reduced. In addition to the feeding damage inspectors should be looking for all life stages of the CLB. In a field of host material the visual survey should be conducted between "sweep samples". Other locations to be examined include grass covered ditch banks and young host crops that are too low to sweep. Experienced surveyors should spend 15 minutes on visual inspection. Less experience surveyors should spend an additional 10 minutes on the visual component.

Optimum conditions for conducting a survey

- **Time of day:** early morning after the dew has evaporated.
- **Temperature**: greater than 10°C, but not midday when the sun is hot and plants are wilting.
- **Wind:** low wind
- In Creston, BC, adults are present once the host cereal crops reach a height of 10 - 20 cm. Larvae are found in the latter half of June into July.

If a positive sample is confirmed in a county or land district the focus of the survey may be switched to a delimitation survey. The delimitation survey may be conducted as a multi-year survey monitoring the growth and movement of the localized CLB populations. Population density within an infested field of host material will be established by examining leaves on 100 tillers. At a positive site, four areas of the field will be sampled. Areas are to be selected at random with 25 tillers selected from each area totalling 100 tillers (25 x 4). The leaves on the tillers are to be examined for CLB eggs and larvae with exact counts taken and recorded.

Grasshopper Monitoring Protocol

<u>Host plants:</u>
A wide range of cultivated crops and rangeland grasses.

<u>Significant Pest Species:</u>

<u>Melanoplus sanguinipes</u> **<u>(Fabricius): Migratory Grasshopper [Orthoptera, Acrididae]</u>**
A grayish-brown species (Figure 1) with a black stripe that usually extends from the eye onto the lateral lobe of the pronotum. Forewings are long, brownish, and bear a raw of dark-brown spots centrally. Hind femora usually have two oblique dark bands. Hind tibia is normally red but sometimes blue. The males are about 20 mm and females are about 28 mm long. Favored habitats are weedy pastures, crops, and similar disturbed areas. They feed both on grass and broad-leafed plants. At high densities, a behavioral change occurs wherein the grasshopper become gregarious, moving as a group; thus the species' common name, "migratory grasshopper".

Migratory grasshoppers overwinter as eggs in the soil. Eggs commence hatching in the midspring. **Hatching occurs from early may to mid July.**
Nymphs feed for about a month before reaching the adult stage. Egg laying begins about a week after the female reaches the adult stage. Females lay 200-300 eggs from late July into the fall in pods 5 cm deep in open soil.

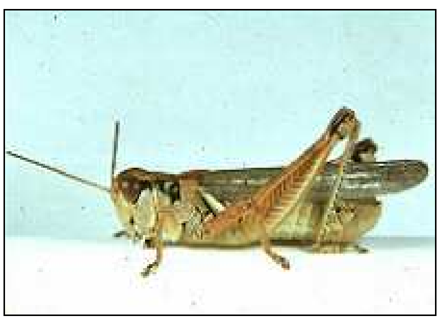


Figure 1: ***M. sanguinipes***

<u>Melanoplus bivittatus</u> **<u>(Say): Twostriped Grasshopper</u>**
Inhabit tall, lush, herbaceous vegetation. Dense populations may reside in tallgrass prairies, wet meadows, roadsides, ditches and crop borders. Hosts include weeds, most crops specially alfalfa and vegetables (broad-leafed plants). It preferentially feed on bean pods. Adults are dark yellowish green in color. This grasshopper derives its name from the two pale yellow stripes extending from the back of the eyes to the tip of the forewings (Figure 2). A solid black stripe is evident along the outer side of the yellowish hind legs. Males are about 24-28 mm long and the females may grow up to 40 mm in length.

Overwinter as eggs that hatch in late May to early June. Nymphs feed for 5-6 weeks. Adults appear in the last 2 weeks of July and lay eggs in pods inserted into soil.

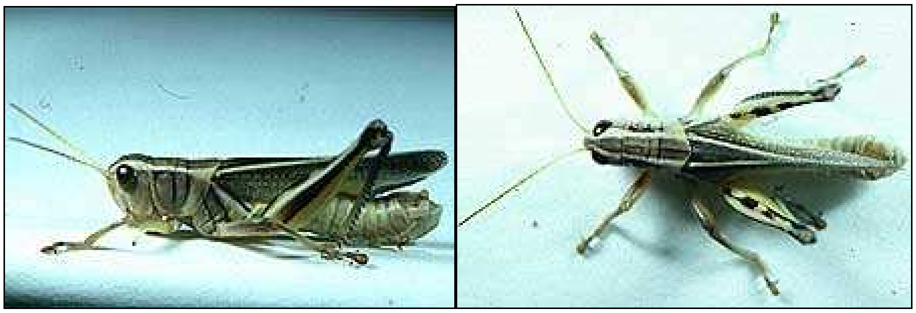


Figure 2: ***M. bivittatus***

Melanoplus packardii **(Scudder): Packard's Grasshopper [Orthoptera, Acrididae]**
Feed on both broadleaf plants and grasses, and is reported to prefer legumes. Adults are grayish, brownish or yellowish brown above and yellowish below (Figure 3). A diffuse dark stripe extends from the top of the head over the top of the pronotum. Forewings are grayish brown, usually with a few small spots. Hind femora are yellowish with a dark stripe along the upper edge. Males are 22-23 mm long, females 26-37 mm.

Eggs commence hatching in midspring. Nymphal development ranges from 47 to 63 days. **Adults are found from July to September.**

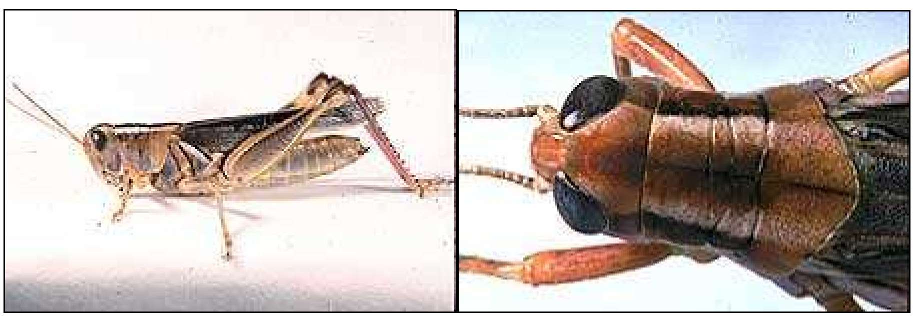


Figure 3: ***M. packardii***

Camnula pellucida **(Scudder): Clearwinged Grasshopper [Orthoptera, Acrididae]**
Clearwinged grasshopper inhabits grassy meadows, often in hilly or mountainous areas. Its tendency to aggregate when densities are high can lead to significant damage to pastures, grains and canola crops.
Adult is a yellowish or grayish brown grasshopper with transparent hind wings (Figure 4). Forewings bear dark round or oval blotches. Median ridge on the pronotum is slightly elevated and notched. Lateral lobes of the pronotum are marked with black. Males are 20-25 mm long; females are 25-31 mm.
Nymphal development ranges from 26-40 days. Because nymphs of the clearwinged grasshopper develop faster than those of the twostriped, adults of the clearwinged may appear first.
Eggs are laid in clutches of 10-38 enclosed in pods.

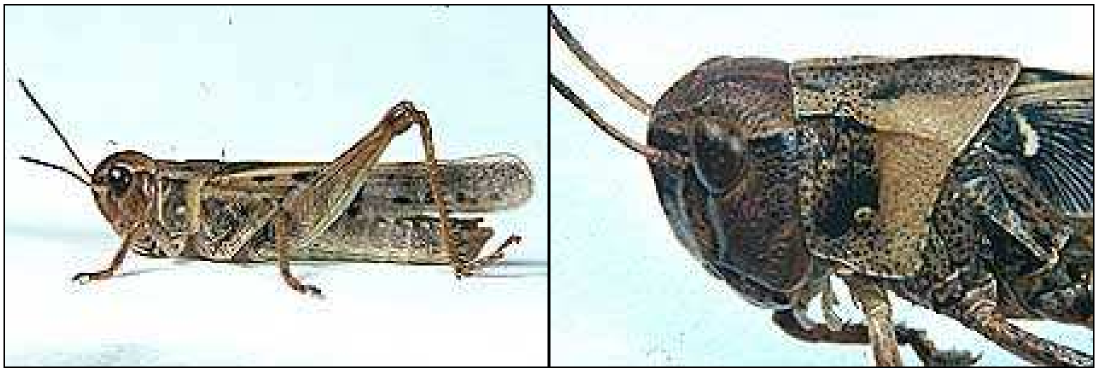


Figure 4: ***C. pellucida***

Monitoring

Timing:

Counting adult grasshopper densities in an area in late summer (when grasshoppers are laying eggs) helps estimate the number of eggs overwintering and forecast the risk of grasshoppers being at problem levels in the next growing season.

To count fully winged adult grasshoppers, sites should be surveyed between **August 1st and September 1st**

Location:

50 m of the field and the roadside (Figure 5). Sample enough locations so that you have a good representation of the grasshopper populations in your area. If possible, sample at least five locations in your district.

Estimate the Density:

At each location, the surveyor will estimate the average number of grasshoppers encountered while walking through the field and along the roadside. Preferably, the average is calculated from visual observations while walking 10 (long) X 1 (wide) m sampling areas and repeating this 10 times (field and roadside). When populations of grasshoppers appear similar or terrain becomes an obstacle the sample areas can be reduced to half as long as a reasonable estimate is made for each site.

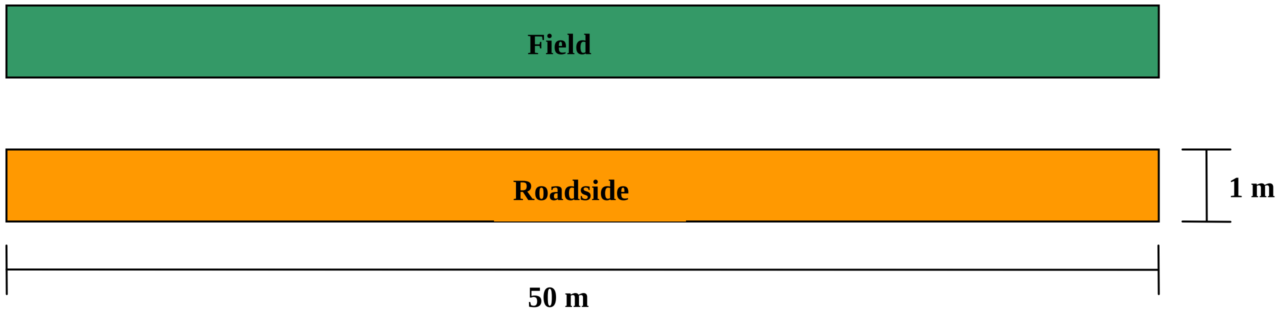


Figure 5. The survey area for each location for both the roadside and field areas.

Measure off a distance of 50 m on the level road surface and mark both starting and finishing points using markers or specific posts on the field margin (Figure 5).These points should be visible from both field and roadside. Starting at one end in either the field or the roadside and walk toward the other end of the 50 m making some disturbance with your feet to encourage any grasshoppers to jump. Grasshoppers that jump/fly through the field of view within a one meter width in front of the observer are counted. A meter stick can be carried as a visual tool to give perspective for a one meter width. However, after a few stops one can often visualize the necessary width and a meter stick may not be required. Also, a hand-held counter can be useful in counting while the observer counts off the required distance. At the end point the total number of grasshoppers is divided by 50 to give an average per meter. For 100 m, repeat this procedure.

Sweep net Sampling:
To monitor different species, use a sweep net to take five sweeps in each location and put in a clear plastic bag to observe the contents of the net for an estimate of species. Enter species percentages **only** if you are sure of the identification.

Damage Rating:

0-2 / m^2 = None to Very Light	8-12 / m^2 = Moderate (action threshold)*
2-4 / m^2 = Very Light	12-24 / m^2 = Severe
4-8 / m^2 = Light	> 24 / m^2=Very Severe

* **> 2 per m^2 can cause losses in lentils at flowering and podding stages**.

Leafhoppers: Vectors of Aster Yellow Disease Monitoring Protocol

Host Plants: Aster yellows can infect a wide variety of crops, including canola, alfalfa, flax, sunflower, echinacea, caraway, coriander, carrot, pea, ornamental plants, and weeds and, to a lesser extent, cereals.

Aster yellow Disease: Aster yellow is caused by a phytoplasma, a plant pathogenic micro-organism which inhabits the phloem of infected plants. The disease is carried from plant to plant by sap-sucking leafhoppers.

The pathogen can overwinter in crops and weeds providing a disease source for the next year, but most infections are carried north from the United States by migrating leafhoppers. In western Canada, the primary vector of the aster yellow phytoplasma is the aster leafhopper, also known as the six-spotted leafhopper *Macrosteles fascifrons* (Stal) (Figure 1). Several other leafhopper species might also be involved in the transmission of aster yellow.

Aster Leafhopper Adult: Adults of aster leafhopper are 3.5 to 4 mm long, light green to yellowish-green in color, and have six black spots arranged in three rows on the front of the head. Their wings held roof-like over the abdomen.

Figure 1: Adult

Aster leafhopper Nymphs: Leafhoppers undergo a series of nymphal stages before reaching adulthood. Nymphs resemble a wingless adult but are much smaller, ranging in size from 0.6mm to 3mm.

Adults and nymphs are sucking insects with piercing mouthparts. **Migratory leafhoppers follow the air currents up from the south-eastern and central United States and onto the Canadian prairies from mid May to mid-June,** but this may vary depending on the prevailing winds. Leafhoppers tend to take flight only when the air temperature exceeds 15℃. Cooler temperatures or r ain will delay their migration. Due to the leafhoppers' poor flying ability, aster yellow tends to proliferate in patches along the edge of a field.

There have been reports of adult aster leafhoppers appearing in the spring, suggesting that they may be able to overwinter as adults. While this has not been confirmed, it could help explain the significant increase in aster yellows in recent years. In Saskatchewan, leafhopper populations increase quickly and remain relatively high all summer.

Monitoring

The objectives of the survey are to establish a checklist of the leafhopper species present in canola crops, to monitor their population and to identify the insect species that carry phytoplasma. The study will provide information essential for the development of technologies for early-warning systems and pest management strategies.
Aster yellows symptoms can be confused with injury caused by nutrient deficiencies, some herbicides, drought or other environmental stresses. For example, nitrogen deficiency in echinacea will usually result in the yellowing of older leaves. The rest of the plant will appear normal.

In canola, purpling is caused by anthocyanin production as a result of stress. Although aster yellows can cause purpling, a purple plant does not necessarily indicate an aster yellows infection.

Percentage of symptomatic plants observed in canola crops is usually less than 1%, but have reached 5-12% in canola crops in some years.

When to sample:

Three sampling periods if possible: **rosette (stage 3.2), early flowering (stage 4.1) and late flowering (4.3-4.4), the two first ones being the most important.**

Insect sampling consists of taking sweeps with a 37 cm diameter sweep net at five locations along a transect within each field: 0 m (grass at the edge of the field), 5 m, 10 m, 20 m and 50 m into the canola field away from the edge. The transect can start from any edge of a field and go straight in the middle of the field (avoid field corner). At each of the five locations along the transect, take 20 sweeps (180 ^{o}) and place all the insects in a plastic bag and label the bag (date, land location and stage of canola development), 1 bag per location (or 5 bags per field).

Lygus: Various Species
Monitoring Protocol

Host Plants:

A wide range of hosts including alfalfa, canola, lentils, potato, strawberries, flax, vegetable crops, fruit trees and weeds such as stinkweed, wild mustard and lamb's-quarters.

Identification, Life Cycle and Damage:

Adult: In western Canada, four species *Lygus lineolaris* (tarnish plant bug), *L. borealis, L. elisus* and *L. keltoni* have been observed. Tarnsh plant bug is the most economically important lygus bugs on the prairies (Figure 1). Adults are about 5 mm long and 2.5 mm wide. They vary in color from pale green to reddish brown and have a distinct triangle or "V" shaped mark on the back. Adult lygus bugs overwinter under litter, debris or plant cover in shelterbeds, headlands and field margins. In the spring adults become active and feed on early-growing plants. They mate and move to crops for feeding and egg laying.

They start laying eggs mid May in southern prairies and in mid June in the Peace river region. Eggs are inserted individually into the stems and leaves of host plants. Egg laying usually lasts 3 weeks but may continue for up to 7 weeks.

Figure 1: ***L. lineolaris*** Adults- 1-5 days

Eggs: Eggs are slightly curved and approximately 1 mm long.

Nymphs: There are five nymphal instars. Young nymphs are light green and wingless (Figure 2). Older nymphs develop black dots on the top of the thorax and abdomen. Wing buds are evident in the fourth and fifth instars. Hot dry weather favors build up of lygus populations. There are two generations per year in the southern prairies, but only one in the northern areas.

Figure 2: Nymphs- 1-5 days

Lygus bugs have piercing-sucking mouthparts and physically damage the plant by puncturing the tissue and sucking plant juices. The plants also react to the toxic saliva that the insects inject when they feed. Lygus bug infestations can cause alfalfa to have short stem internodes, excessive branching, and small, distorted leaves. They feed on buds and blossoms and cause them to drop. They also puncture seed pods and feed on the developing seeds causing them to turn brown and shrivel.

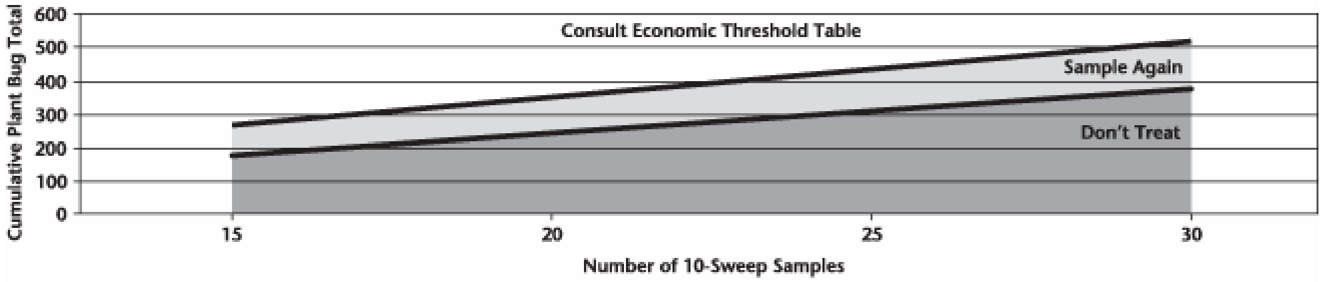
Monitoring

Begin monitoring canola when it bolts and continue until seeds within the pods are firm. Since adults can move into canola from alfalfa, check lygus bug numbers in canola when nearby alfalfa crops are cut.

Start monitoring at the bud stage. Sample the crop for lygus bugs on a sunny day when the temperature is above 20℃ and the crop canopy i s dry. With a standard insect net of 38 cm diameter, take ten 180° sweeps. Count the num ber of lygus in the net.

Repeat the sampling in another 14 locations. Samples can be taken along or near the field margins. Calculate the cumulative total number of lygus and then consult the sequential sampling chart (Figure 3). If the total number is below the lower threshold line, no treatment is needed. If the total is below the upper threshold line, take more samples.

Figure 3. Sequential Sampling for Lygus Bug at Late Flowering Stage



If the total is on or above the upper threshold line, calculate the average number of lygus per 10-sweep sample and consult the economic threshold table.

Table 3. Lygus Bug Economic Threshold

Application Costs		**Number of Lygus Bugs at Different Canola Crop Stages**						
$/ha	**$/ac**	**Bud**	**End of Flowering**			**Pod Ripening**		
22	8.90		14	12	10	20	17	15
24	9.70		16	13	11	22	18	16
26	10.50	No economic threshold available	17	14	12	24	20	17
28	11.35		18	15	13	25	22	19
30	12.15		19	16	14	27	23	20
32	12.95		21	17	15	29	25	21
Canola Price								
$/tonne			220	260	300	220	260	300
$/bu			5.00	5.90	6.80	5.00	5.90	6.80

Economic Threshold

The economic threshold for lygus bugs in canola covers the end of the flowering and the early pod ripening stages. Once the seeds have ripened to yellow or brown, the cost of controlling lygus bugs may exceed the damage they will cause prior to harvest, so insecticide application is not warranted.

Consider the estimated cost of spraying and expected return prior to making a decision to treat a crop. **For example, if an application will cost $26/ha ($10.50/ac) and the expected return is $260/tonne ($5.90/bu), the threshold level is an average of 14 bugs per 10-sweep sample. An economic threshold for lygus bugs in canola at the bud stage has not been established.**

If soil moisture levels and rainfall are high at flowering, plants likely will be able to compensate for damage caused by lygus bug populations well above economic thresholds and control may not be necessary. If the plants are under moisture stress during this time they are unable to compensate for most of the feeding injury. Spray using the economic thresholds above.

Wheat Midge
Monitoring Protocol

<u>Host Plants:</u> All wheat varieties are currently susceptible to wheat midge, but some are more seriously affected than others. Although the midge also attacks other members of the grass family, including barley, couch grass, intermediate wheat grass and rye, infestations on these plants are usually not serious enough to warrant control.

<u>Identification, Life Cycle and Damage:</u>

Adult: The female midge is a tiny, fragile fly about 3 mm long with a salmon pink body (Figure 1). The male is smaller. The head is light brown with two large black eyes. Legs are light brown and antennae are dark brown. Wings are dusky and fringed with hairs. **Adult midges emerge from the pupal stage in the soil over a 5-6 week period, from mid-June to mid-July.** This is about the time when the wheat heads are emerging from the sheath and beginning to flower.

During the day, the midge remains within the humid crop canopy. During warm (>15℃), calm (wind speed is less than 10 km/h) evenings, the female midges lay eggs on the wheat kernels singly or in groups of 3-5. Egg laying takes place just prior to or at anthesis, over their 4 or 5 day lifespan.

Eggs: The tiny, orange-coloured eggs are barely visible to the human eye. Plants are most vulnerable to attack if the eggs are laid during the time the heads are about one-half emerged from the boot to half-flowering.

Larvae: Eggs hatch in 5-7 days and the larvae move to the surface of the developing kernels and feed for 2-3 weeks. The newly hatched larvae are white; the mature larvae are oval-shaped and are orange-red in colour (Figure 2). Larval feeding will cause the kernels to shrivel. A few kernels may be aborted entirely. Others will not fully develop and will be so small and light. Damage will not be visible unless the developing kernels are inspected. The mature larvae remain in the wheat head, enclosed in a transparent skin, until activated by rain or damp weather conditions. Then, these larvae drop to the soil surface, burrow down into the soil (up to 10 cm down) and pass the winter in a resting stage enclosed in cocoons which are smaller than canola seeds (Figure 3).

Pupa: In the following spring, if the soil is moist enough, they pupate near the surface. Overwintering larvae may remain dormant until conditions are favourable for development in the following spring or years later. The adult midges emerge about 2 weeks later.

Figure 1: Adult **Figure 2:** Larva

Figure 3: Canola seeds (top) and wheat midge cocoons (bottom)

<u>Monitoring</u>

Field Inspection for Adults:

Inspections should be carried out in the evening (preferably after 8:30 pm.) when the female midges are most active. On warm (at least 15ºC), calm evenings, the midge can be observed in the field, laying their eggs on the wheat heads. Midge populations can be estimated by counting the number of adults present on 4 or 5 wheat heads. Inspect the field daily in at least 3 or 4 locations during the evening.

Sampling of Soil Cores for Extraction of Midge Cocoons

After the wheat has been harvested, a metal tube, with an inside diameter of 2.54 cm, is inserted into the soil to a depth of 15 cm and the resulting core, which has a surface area of 5.06 cm^2, is the basic sampling unit. Five cores will be taken at random from field at three locations within the field. Cores will be placed in individual plastic bags and stored at 2^oC until they could be processed. The cores will be processed by wet sieving as described by Doane *et al*. (1987), and cocoons, larvae, and pupae counted. The larvae will be dissected to determine if they are parasitized.

Reference: Doane, J.F., O. Olfert & M.K. Mukerji. 1986. Extraction precision of sieving and brine floatation for removal of wheat midge, *Sitodiplosis mosellana* (Diptera: Cecidomyiidae), cocoons and larvae from soil. J. Economic Entomology 80: 268-271.

Economic Thresholds:

To maintain optimum grade: **1 adult midge per 8 to 10 wheat heads during the susceptible stage.**

For yield only: **1 adult midge per 4 to 5 heads**. At this level of infestation, wheat yields will be reduced by approximately 15% if the midge is not controlled.

Inspect the developing kernels for the presence of larvae and the larval damage.

Wheat Stem Sawfly
Monitoring Protocol

<u>Host Plants:</u> Wild grasses are the primary host plants. Spring wheat and rye are the main cereals attacked.

Plant age is important to egg-laying females. Plants that have not reached the stem elongation stage are not acceptable to females. Similarly, plants in the boot stage are immune to sawfly attack.

<u>Identification, Life Cycle and Damage:</u>

Adult: The adult sawfly is wasp-like, slender, and about 8-13 mm long. The body colour is shiny-black, with 3 yellow bands on the abdomen (Figure 1). They have smoke-colored wings and yellow legs. **The adult emerges in June and can usually be found in wheat fields until mid-July.** Males emerge first; females emerge later. Adults have a habit of resting on the stems with head downward. During this time, the female inserts an egg into the elongating internode of a wheat stem. They use their saw like ovipositor to cut a slit in the plant to lay eggs.

When the weather is rainy in the fall or spring, the numbers of large, head-bearing stems of native grasses are adequate for sawfly populations. But when there is a drought, the numbers of grass stems suitable for attack are few and the sawfly concentrates its attack on wheat instead of on grasses.

Eggs: Eggs are crescent-shaped, glassy, milky white and about 1 mm long. The eggs will hatch in 5-8 days.

Larva: The mature larvae are dull-white, about 13-14 mm long, worm like and have a well-defined, brown head. The larva will assume an S-shape when extracted from the stem. Larvae feed within the stem until the plant is nearly mature (Figure 2). It then girdles the stem at ground level, plugs the pith cavity, and overwinters in the lower part of the stem (stub) enclosed in a long thin cocoon.

The sawfly larva bores down inside the stem and makes a discoloured tunnel from about the top joint to the root. Sometimes, however, eggs are laid above the top node of the plant and tunnelling by larvae destroys sufficient vascular tissue so that the head turns white.

Larvae feed within the stem reduce both yield (5 to 15 percent decrease in total seed weight) and quality of grain (from reduced protein and kernel weight). Drought conditions can reduce infestations in the following year by killing plants that have larvae inside them. Drought in the spring can cause overwintered larvae to re-enter diapause but the influence of this on population size is not clear.

Pupa: Overwintered larvae pupate in the spring. The pupal stage begins in mid- to late May and lasts about 10 days. Wheat stem sawfly has one generation per year.

Figure 1: Adult

Figure 2: Larva

Monitoring

Sweep Net Counts for Adults:

Use a sweep net to sample for wheat stem sawfly. **When sweep net samples average 2 female sawflies per 10 sweeps, you can expect about 12% cut stems; 4 females per 10 sweeps cause about 23% cut stems.**

Determine the **percent of females found**. The female sawfly will have a distinct ovipositor used for inserting the eggs into the stem.

Site Selection for Damage symptoms:

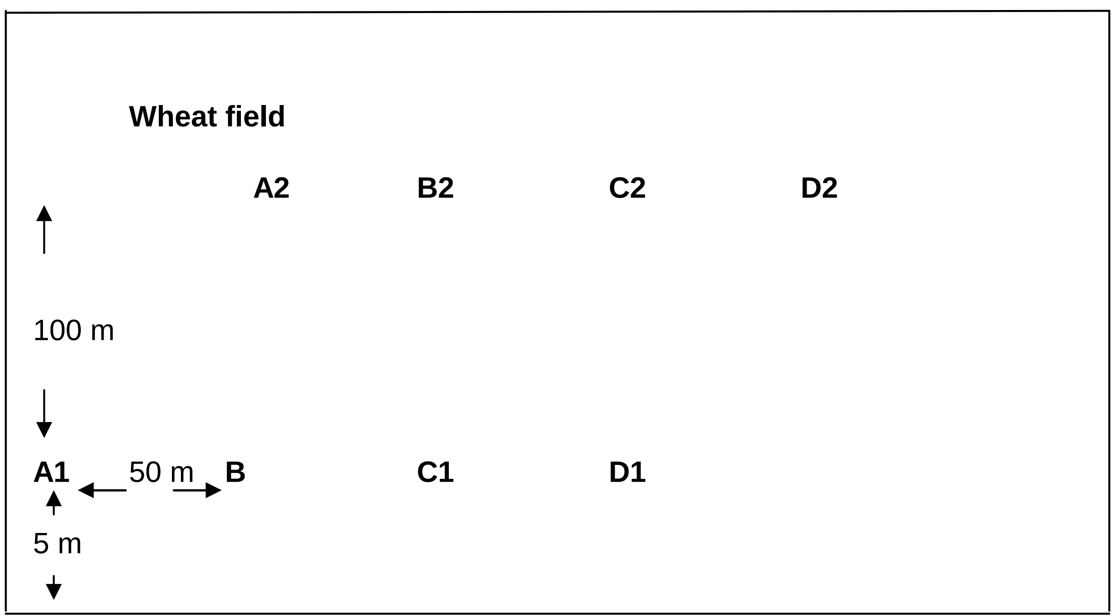
The greatest losses occur around the margins of fields. During warm, sunny, windless weather, especially after rain, the sawflies disperse widely. Their attack is otherwise concentrated near the field margins**. Monitor prior to harvest**. Stems that contain a sawfly larva usually develop a reddish brown band below the second or third node. Determine **percentage of plants cut by sawfly larvae per square metre**. Control methods are required **if 10 to 15 per cent of the crop in the previous year was cut by sawfly.**

Procedure:

Walk about 50 m along field edge (or stubble crop interface), 5 m into the crop

Randomly place 1 m stick parallel along one row. Count **the number of long stems** cut by swather or combine and the **number of short stubs** cut by sawfly along the meter row. It is easier to find the stubs cut by the sawfly by feeling them with fingers. They are very close to the ground surface and are about ¾ inch long. On the same spot, count the number of heads on the ground between two rows along a 5 m stretch (5 paces OK). Repeat sampling about 100 m toward the middle of the field perpendicular to the edge (see the figure attached).

For fields not seeded in rows, use I m square quadrat to count the heads and one of the sides to count the stems harvested and those cut by the sawfly.



APPENDIX B – Degree Day Modeling

What do these degree-day maps mean?

Background
Because insects are cold-blooded, temperature plays a major role in their growth and development. There is a threshold temperature, or BASE, below which it is too cool for further insect development. Degree-days (or heat units) are a unit combining time and temperature; one degree-day is 24 hours with the temperature one degree above the base. Base temperatures are specific for different insect species although 5^oC and 10^oC are common to a number of insect models. For some species, researchers have been able to model the number of degree-days required to complete each stage of an insect's life cycle.

Weather Parameters
The weather parameter that is most widely used to calculate degree-days is air temperature. However, more sophisticated insect growth models may also incorporate solar radiation, soil temperatures, relative humidity and etc. to provide more accurate predictions. In the case of models that use air temperatures only, it should be remembered that the degree-day calculations are only estimates of the influence of temperature on the different life stages of the insects. The insect life stage of interest may be in the soil or on the soil, in the shade or in the sun, and may be under other biological stress like poor nutrition or disease. So, depending on these varying conditions, the calculations based on air temperature may over-estimate or under-estimate the actual rate of development.

Calculation Methods
The accumulation of degree-days for the different insect growth models presented by the Tri-Provincial Insect Monitoring group begins on **April 1** each year (Biofix date). The method used to calculate degree-days is the **Modified Sine Wave** as described by Allen 1976[1]. The degree-days are calculated weekly for each available weather station and these point data are then used to generate surface maps of degree-day accumulations.

Insect Growth Models
The details of degree-day models used in our forecast maps for wheat midge, bertha armyworm and grasshoppers are as follows:

Wheat Midge

- Base temperature is 5^oC (Elliott et al. 2009)
- Male midge begin emerging at about 650 degree-days reach 50% emergence by 765 degree-days and 90% emergence by 850 degree-days.
- Female midge emerge later; 10% emergence at 660, 50% emergence by 800 and 90% emergence by 880 degree-days.

Bertha Armyworm

- Base temperature is 7^{o}C (Bailey 1976[2])
- We need to know when Bertha will finish pupation and begin to emerge and fly. As a result, the base 7 map is expressed, as % of the heat requirements needed to finish pupation 100%.

Grasshoppers

- Base temperature is 10^{o}C (Gage and Mukerji, 1976[3])
- Degree-days needed for 50% of the population to reach stage

Hatch – 325 degree-days
Nymphs
  Second instar – 414 degree-days
  Third instar - 501 degree-days
  Fourth instar - 614 degree-days
  Fifth instar - 801 degree-days
Adult - 936 degree-days

- Below are a few things to note regarding estimates of grasshopper development. Grasshopper populations are usually made up of 4-5 economically important species. Each species has slightly different behavior and feeding habits which will influence their overall growth response within a given temperature range. In addition, studies have shown that grasshoppers can adjust their internal body temperature above air temperature by basking in direct sun. They also move to thermo-regulate internal temperature in and out of sun depending on temperature and wind speed. (Lactin and Johnson, 1996 & 1997[4]). As a result, we would expect estimates based on air temperature alone to accumulate degree-days too slowly in spring when air temperatures are low and solar radiation is relatively important in warming nymphs. Conversely, in the heat of the summer when air temperatures are higher relative to solar radiation, this method may over estimate growth rates somewhat. Unfortunately, solar radiation data are only available in a sub-set of the total number of weather stations that do measure air temperature.

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[1] Allen, Jon C., 1976. A Modified Sine Wave Method for Calculating Degree Days. Environ Entomol. 5: 388 - 396

[2] Elliott, R.H., Mann, L., Olfert, O. (2009) Calendar and degree-day requirements for emergence of adult wheat midge, *Sitodiplosis mosellana* (Gehin) (Diptera: Cecidomyiidae) in Saskatchewan, Canada. Crop Protection 28: 588–594

[2] Bailey, Clyde G., 1976. Temperature Effects on Non-Diapause Development in *Mamestra configurata* (Lepidoptera: Noctuidae). Can. Ent. 108: 1339 – 1344.

[3] Mukerji, M.K and Stuart Gage, 1978.

[4] Lactin, Derek J. and Dan L. Johnson. 1997. Response of body temperature to solar radiation in restrained nymphal migratory grasshoppers (Orthoptera: Acridae): influences or orientation and body size. Physiological Entomology 22: 131-139.