Crop Profile for Collards in Arizona

Prepared: February, 2001

Family: Brassicaceae (Cruciferae)

Scientific name: *Brassica oleracea* var. *viridis* L (Brassica oleracea var. acephala DC. in part)

Edible portions: leaves, consumed cooked.

Use: fresh vegetable, greens, potherb

Alternate names: Cow Cabbage, Spring Heading Cabbage, Tall Kale, Tree Kale, and Winter Greens.

General Production Information

- In 1997, Arizona ranked 13th in the United States for collards production, contributing 1% of the nations collard production[2].
- 153 acres of collards was harvested in Arizona in 1997[2].
- Approximately 168,000 cartons of collards were harvested in Arizona in 1997.
- In 1997, Collards production had an approximate value of 899 thousand dollars in Arizona.
- The majority of collards production occurs in Maricopa County, but there is a small amount that occurs in Yuma and Cochise Counties.
- Land preparation and growing expenses for collards are approximately $1.45/carton[5].
- Harvest and post harvest expenses for collards are approximately $3.30/carton[5].

Cultural Practices

General Information: In the state of Arizona, collards are grown during the fall and winter. Planting of collards begins in September and is usually completed by the middle of November[7]. In Arizona, temperatures during the collard growing season may range from 30°F to 90°F[7]. Collards grow best in cooler weather but can survive a very wide range of temperature[4]. In fact, the flavor of the leaves is enhanced when the plant experiences freezing temperatures[4]. However, a long period (8-10 weeks) of temperatures below 40°F can induce flowering[3]. In Arizona, collards are grown on soils that range from a dry loam to a sandy loam, with a pH of 7.5-8.0[7]. Collards grow best on well-drained soil[4].

Cultivars/Varieties[6]: The most common variety of collards grown in Arizona is 'Vates'. This variety is an old standard that has been grown for many years. It is open-pollinated and is good for bunching.

Production Practices[3, 7]: Prior to planting, the field is deeply tilled, disced, land planed and then the beds are formed. The field is then pre-irrigated. A preplant herbicide may be applied prior to bed formation. If a pre-plant fungicide, such as mefenoxam, is utilized it is usually applied after bed formation but prior to planting.

Collards are directly seeded ¼ to ½ an inch deep into 40" beds. There are four rows per bed, and plants are spaced 1 to 2 inches apart within the row.

During the course of production, the field is cultivated two or three times. The field is also side dressed with fertilizer two or three times, depending on necessity. Little of the collard plant is left after harvest to replenish soil fertility; thus collards are referred to as 'heavy feeders' and require more fertilizer than other crops require. An adequate supply of nitrogen is especially important for leaf growth. Furrow irrigation is used to supply collards with a consistent water supply.

Harvesting Procedures: Collards require six to eight weeks, from time of seeding, to mature[4]. Harvesting usually begins the end of October and is complete by the beginning of January[8]. The entire collards plant can be harvested, by hand, at ground level. It is also possible to harvest only the mature leaves and allow the rest of the plant to continue producing leaves[1]. The leaves are then trimmed, washed and tied into bunched in the field[8]. Collards are packed with 1 or 2 dozen bunches per wax carton[8]. Leaves stored at 32°F and a relative humidity of 95-100% have a storage life of 10 to 14 days. To meet Arizona standards, all collards must be fresh, fairly tender, fairly clean, free from decay and free from serious damage[9]. All collards must be of one variety. No more than 5%, by weight, in any container or lot may have any one defect. No more than 10%, by count, can fail to meet Arizona standards[9].

Insects and Mites

(7, 10, 11, 12, 13, 14, 15, 16, 17, 18)

Hymenoptera

Harvester Ant (*Pogomyrmex rugosus*)

Ants are not frequently a pest of Arizona crops; however, when they do occur in a field they can be insidious. The harvester ant is primarily a pest during stand establishment. They eat the seedlings and carry the planted seeds and seedlings back to their nests. When there are ants in a field, typically there is no vegetation surrounding the ant hill. Ants generally do not cause damage to the mature collards plant. Ants can also be a pest to people working in the field.

Sampling and Treatment Thresholds: University of Arizona experts suggest that a field should be treated at the first signs of damage[15].

Biological Control: There are no effective methods for the biological control of ants.

Chemical Control: Hydramethylnon is often used to control harvester ant populations, by placing it around the anthill. Worker ants will carry the poisoned bait back to their nest and distribute it to the other ants and the queen. Hydramethylnon, however, can only be used on bare ground, outside borders and ditch banks. Carbaryl baits can be used within the crop field to control ant populations.

Cultural Control: Surrounding the field with a water-filled ditch can help control ant migration into the field. This method, however, has little value if the ants are already in the field.

Post-Harvest Control: There are no effective methods for the post harvest control of ants.

Alternative Control: Rotenone is an alternative method used by some growers to control ant populations. Another method is to pour boiling water that contains a citrus extract down the anthill to kill populations inside.

Coleoptera

Striped Flea Beetle (*Phyllotreta striolata*)
Potato Flea Beetle (*Epitrix cucumeris*)
Western Black Flea Beetle (*Phyllotreta pusilla*)
Western Striped Flea Beetle (*Phyllotreta ramosa*)

The color of flea beetles varies between species, but all species have a hard body and large hind legs. When flea beetles are disturbed, their large hind legs allow them to jump great distances.

In Arizona, flea beetles are particularly damaging to cole crops. The female flea beetle lays her eggs in the soil and on leaves of the collards plant. Depending on the species, the larvae feed on the leaves or the roots of the collards plant. The adult beetles will also feed on the collards plant, chewing small holes and pits into the underside of leaves. These insects are the most damaging during stand establishment. Even a small population can stunt or kill a stand of seedlings. Any damage to the collards' leaves will cause an economic loss.

Sampling and Treatment Thresholds: Flea beetles often migrate from surrounding production areas and Sudan grass. Fields should be monitored weekly for flea beetles and damage. According to University of Arizona guidelines, collards should be treated prior to the formation of the harvested leaves when there is 1 beetle per 50 plants[14]. After leaf formation the crop should be treated when there is 1 flea beetle per 25 plants[14].

Biological Control: There are no natural predators or parasites that can effectively control flea beetle populations.

Chemical Control: Methomyl, diazinon and pyrethroids are commonly used for the control of flea beetles. Methomyl is foliar applied; diazinon and pyrethroids can be foliar-applied or chemigated. Chlorpyrifos also has some activity against flea beetles. Diazinon and pyrethroids applied by chemigation have the added benefit of targeting crickets, grasshoppers and lepidopterous larvae.

Cultural Control: It is important to control volunteer plants and weeds, in and around the field, which could act as a host for flea beetles. Crop rotation is important; however, flea beetles have a wide range of hosts so not all crops are suitable for rotation. Collards fields should be disked immediately following final harvest. It is also important that Sudan grass is plowed under within a week of the final harvest, as this crop often harbors flea beetles.

Post-Harvest Control: There are no effective methods for the post-harvest control of flea beetles.

Alternative Control: Some growers use rotenone dust and pyrethrins to control flea beetles. Alternative control of these pests, however, is very difficult.

Darkling Beetle (*Blapstinus* sp.)

Rove Beetle (*Staphylinids* sp.)

Darkling beetles are dull black-brown in color. They are often confused with predaceous ground beetles, which are also black-brown but are shiny and lack clubbed antennae. It should be noted that the predaceous ground beetle is a beneficial because it feeds on lepidopterous larvae and other insects.

Rove beetles are a ¼" in length, or smaller, have a shiny, dark black-brown body and very short elytra that cover the wings. These beetles are frequently confused with winged ants and termites.

Darkling and rove beetles are most damaging during seedling establishment, digging planted seeds out of the soil. They will also feed on collards seedlings, girdling plants at the soil surface. Sometimes these beetles feed on the leaves of older plants. Any damage to the marketable leaves of collards will cause economic loss.

Sampling and Treatment Thresholds: Nighttime is the best time to monitor a field for darkling beetles; this is when they are the most active. During the day they tend to hide in the soil or debris. Flea beetles and rove beetles often migrate from nearby cotton and alfalfa fields or weedy areas. According to University of Arizona guidelines a collards field should be treated when beetle populations are high or there is a threat of migration into the field[14].

Biological Control: There are no effective methods for biologically controlling rove and darkling beetles.

Chemical Control: Placing baits around the perimeter of the field will provide some control when beetles migrate into the field. Methomyl, diazinon, chlorpyrifos and pyrethroids are routinely used to control rove beetle and darkling beetle populations. Diazinon and pyrethroids can be chemigated through the sprinkler system or foliar applied. These two active ingredients will also help control cricket, grasshopper and lepidopterous larvae populations.

Cultural Control: It is important to control weeds in the field, and surrounding the field, that can act as hosts for darkling and rove beetles. Ditches filled with water around the field's perimeter can deter beetle migration into the field. This control method, however, will have little value if the beetles are already in the field. Fields should be deeply plowed to reduce soil organic matter and beetle reproduction.

Post-Harvest Control: There are no post-harvest control methods for rove beetles or darkling beetles.

Alternative Control: Some growers use rotenone or neem oil to control darkling and rove beetles.

Orthoptera

Cricket (*Gryllus* sp.)

Cricket populations build up in cotton fields, Sudan grass and desert flora. At the end of the summer, crickets move from these fields into collards fields. Fields that use over-head sprinkler irrigation encourage inhabitance by creating an ideal environment for crickets. Female crickets often lay their eggs in damp soil.

Crickets are rarely a problem in Arizona but dense populations are capable of destroying an entire crop. Crickets are ½ to 1" in length, and brown-black in color. Crickets commonly feed at night; during the day crickets hide in the soil, weeds, ditches and under irrigation pipes. Crickets attack collards seedlings as they emerge from the soil. If cricket populations are large enough, they can completely decimate an entire crop.

Sampling and Treatment Thresholds: Crickets are difficult to monitor for during the day, as they tend to hide. One can check underneath irrigation pipes; however, a visual inspection of damage is usually sufficient to give an estimate of cricket activity. Fields planted near cotton or Sudan grass should be closely monitored. The University of Arizona suggests that a field should be treated when cricket damage is high or there is a threat of cricket migration into the field[14].

Biological Control: There are no effective methods for biologically controlling cricket populations.

Chemical Control: Baits such as carbaryl, can be used to control cricket populations. Baits are usually placed at the field borders to target crickets migrating into the field. Methomyl, diazinon and pyrethroids are the most commonly used treatments for controlling cricket populations. These insecticides can be ground applied or applied by chemigation. Spraying, rather than using baits, has the added benefit of also targeting lepidopterous pests.

Cultural Control: Fields should be disked immediately following harvest, this will help control cricket populations.

Post-Harvest Control: There are no effective methods for the post-harvest control of crickets.

Alternative Control: Some growers use rotenone to control cricket populations.

Spur-Throated Grasshopper (*Schistocerca* sp.)
Desert (Migratory) Grasshopper (*Melanoplus sanguinipes*)

In Arizona, grasshoppers are usually not a threat to collards stands. Occasionally, sometimes after a heavy rain, the grasshopper population can 'explode'. In these years grasshoppers move from the desert into produce fields and can decimate entire crops. Due to their ability to fly, it is difficult to prevent the migration of grasshoppers into a field. There have been such outbreaks in previous years in Arizona; however, they are rare. Grasshoppers are foliage feeders and will chew holes into leaves. Shippers will not accept any damage to collards leaves. In most years, grasshopper populations are so small their damage is insignificant.

Sampling and Treatment Thresholds: University of Arizona experts suggest that fields should be treated as soon as grasshoppers begin to cause damage to the crop[15].

Biological Control: A predaceous protozoon, *Nosema locustae*, can sometimes be used to control grasshopper populations.

Chemical Control: If grasshopper populations are large, chemical control is usually the only option, however chemical control of these insects can be difficult. Pyrethroids, such as lambdacyhalothrin have been occasionally used in the past. Carbaryl can also be used to control grasshoppers. Carbaryl can be sprayed, chemigated or applied as baits.

Cultural Control: Due to their ability to fly, it is difficult to prevent the migration of grasshoppers into a field. If grasshopper populations are decimating a field, replanting is often the only option.

Post-Harvest Control: There are no effective methods for the post-harvest control of grasshoppers.

Alternative Control: Some growers use rotenone to control grasshopper populations.

Diptera

Leafminers
(*Liriomyza* sp.)

Leafminers are not currently a large threat to collards Production in Arizona, but there is potential for leafminers to be quite destructive to collards production.

Adult leafminers are small, shiny, black flies with a yellow triangular marking on the thorax. The adult female leafminer oviposits her eggs within the leaf tissue. Male and female flies feed at these puncture sites. The larvae hatch inside the leaf and feed on the mesophyll tissue. The larvae do not emerge from within the leaf until they pupate. Leafminers usually pupate in the soil, although on occasion they will pupate on the leaf surface. When conditions are favorable, leafminers can complete a life cycle as quickly as 3 weeks.

As larvae feed on the mesophyll tissue, they create extensive tunneling within the leaf. The width of these tunnels increases as the larvae grow. These mines cause direct damage by decreasing photosynthesis; as well, the puncture wounds provide an entryway for pathogenic infection. The disfiguration of the leaves renders the collards unmarketable. Leafminers that pupate between the leaves of collards will contaminate the harvested plant. These bodies may also die and rot, providing a medium for the growth of pathogens. Leafminers, however, are often only found feeding on the cotyledons of collards.

Sampling and Treatment Thresholds: It is important that the crop is monitored regularly for leaf mines, larvae and adult flies. The cotyledons and first true leaves are the first to be mined. Mining is more visible on the undersurface of the leaf; thus both leaf surfaces must be examined. Presence of leafminer parasites and parasitized mines should also be determined. Yellow sticky traps are a good method for measuring leafminer migration into a field, as well as, determining which species are present. It is important to accurately identify which species are present, because insecticide resistance has been documented for *Liriomyza trifolii*.

The University of Arizona recommends the following treatment thresholds. Prior to the formation of the harvested leaves, collards should be treated when populations have reached 1 active mine per leaf[14]. After leaf formation, treatment should occur when populations reach 1 mine per leaf per 25 collards plants[14].

Biological Control: *Diglyphus* and *Chrysocharis*
genera of parasitic wasp are sometimes utilized to control leafminer populations. Insecticides used to control noxious pests should be used with care because they can eliminate parasitic wasps causing a secondary leafminer outbreak.

Chemical Control: Diazinon is commonly used to control *L. sativae* adults but is ineffective against *L. trifolii*. Spinosad is used for the control of the adults and larvae of both *L. sativae* and *L. trifolii*. Spinosad is the only available chemistry that effectively controls *L. trifolii*. Insecticide resistance has been noted in *L. trifolii* populations, thus there is a need for a diversity of insecticides to allow resistance management.

Cultural Control: It is best to avoid planting near cotton, alfalfa and other host fields, because leafminers will migrate from these fields into collards fields. A field that has a leafminer infestation should be disked immediately following harvest to kill pupating larvae.

Post-Harvest Control: There are no effective methods for the post-harvest control of leafminers.

Alternative Control: Some growers use insecticidal soaps to control leafminer populations.

Lepidoptera

Lepidopterous complex = diamondback moth, loopers, beet armyworm, corn earworm, tobacco budworm and imported cabbageworm.

Black Cutworm (*Agrotis ipsilon*)
Variegated Cutworm (*Peridroma saucia*)
Granulate Cutworm (*Agrotis subterranea*)

The threat of cutworms in Arizona is sporadic, and appears to vary in response to environmental conditions, such as warm weather. Cutworm populations are heaviest during the fall. The adult moth has gray-brown fore wings with irregular markings; the hindwings are lighter in color. The female moth lays her eggs on the leaves and stem near the soil surface.

Seedlings are the most significantly impacted by cutworm attack. Newly hatched larvae feed on the leaves temporarily, but then drop to the soil surface and burrow underground. The larvae emerge at night and feed on the collards plant. The cutworm attacks collards by cutting the stem at, or just below, the soil surface. A single cutworm is capable of damaging several plants in one evening and a large population can destroy an entire collards stand. When cutworms have been active, one might observe several wilted or collapsed plants in a row. A stand that has recently been thinned is especially sensitive to cutworm attack. Sometimes a cutworm will bore into a collards plant from below. Cutworms frequently occur in fields that were previously planted with alfalfa or pasture.

Sampling and Treatment Thresholds: Prior to planting, the field, field borders and adjoining fields should be monitored for cutworms. Pheromone traps can be used to monitor the presence of cutworms in a field. Once seedlings have emerged, fields should be scouted twice a week. If an area of several wilted or cut off plants is discovered, the surrounding soil should be dug into and searched for cutworms. Cutworms are nocturnal; therefore it is best to scout for them on the soil surface during the evening. Cutworms are often not noticed until crop damage has become severe. University of Arizona experts suggest treating a field as soon as soon as stand loss begins[14].

Biological Control: There are some natural enemies to the cutworm, however they do not provide adequate control.

Chemical Control: Baits can be used to control cutworms but are more effective when used prior to collards emergence. These baits should be placed in the areas where cutworms have been found in previous years. Cutworms often occur at the field borders or in isolated areas within the field. Sometimes spot and edge treatments are sufficient to control cutworm populations. Spinosad, chlorpyrifos, methomyl and pyrethroids are the most commonly used chemistries for controlling cutworm populations. Cutworms, however, are often controlled when the crop is sprayed for stand-establishment pests. The larvae usually do not get an opportunity to establish a population.

Cultural Control: Fields that are in close proximity to alfalfa fields are especially prone to cutworm infestation, and should be carefully monitored. Cutworms tend to reoccur in the same area of a field and in the same fields. It is important to control weeds that can act as hosts to cutworms, in the field and surrounding the field. The field should be plowed a minimum of two weeks prior to planting, in order to kill cutworms, hosts and food sources.

Post-Harvest Control: There are no effective methods for the post-harvest control of cutworms.

Alternative Control: Some growers use *Bacillus thuringiensis*
(Bt) for the control of cutworms. It is best to spray Bt in the dark because it is UV light and heat sensitive. Spraying at night will give the longest period of efficacy.

Saltmarsh Caterpillar (*Estigmene acrea*)

Saltmarsh caterpillar populations are heaviest in the fall. These larvae are more common in cotton, alfalfa, bean and sugarbeet fields and are not normally a cole crop pest. The larvae, however, will migrate from surrounding host fields.

The adult saltmarsh caterpillar moth has white forewings that are covered with black spots; its hindwings are yellow. The female moth lays groups of 20 or more eggs on the leaf surface. The young larvae are yellow-brown in color and covered in long, black and red hairs. Older larvae sometimes develop yellow stripes down the sides of their bodies. These caterpillars are often referred to as 'wooly bear caterpillars'.

The saltmarsh caterpillars feed on seedlings and can skeletonize the leaves of older plants. The larvae often feed in groups on older plants. If populations are high, they can decimate an entire seedling stand. Any damage to the leaves of collards results in an unmarketable plant.

Sampling and Treatment Thresholds: According to University of Arizona experts, fields should be treated at the first signs of damage[15].

Biological Control: There are no effective methods for the biological control of saltmarsh caterpillars.

Chemical Control: Field edges should be sprayed when saltmarsh caterpillars begin to migrate into the collards field. Methomyl, spinosad, tebufenozide, chlorpyrifos and pyrethroids are often used for controlling saltmarsh caterpillars. Methomyl, pyrethroids and chlorpyrifos are all foliar-applied contact insecticides. Spinosad is a translaminar insecticide that must be consumed or tread upon to kill the larvae. Tebufenozide is an insect stomach poison that must be consumed to be effective.

Cultural Control: The simplest way to control saltmarsh caterpillars is to prevent their migration into a field. Monitoring surrounding cotton and alfalfa fields prior to collards emergence will help assess the degree of risk for the crop. Saltmarsh caterpillars do not like to cross physical barriers. A six-inch high aluminum foil strip or irrigation pipes that the larvae cannot crawl under will provide a suitable barrier to the field. These barriers can also be used to herd the larvae into cups of oil. A ditch of water containing oil or detergent that surrounds the perimeter of the field can also be used as a barrier. Barriers work well to exclude saltmarsh caterpillars from the field, but will have no useful value if the larvae have already infested the field.

Post-Harvest Control: There are no effective methods for the post-harvest control of saltmarsh caterpillars.

Alternative Control: *Bacillus thuringiensis*
may be used to control saltmarsh caterpillars. One important consideration when using *B. thuringiensis* is its tendency to break down when exposed to UV light and heat. Usually it is sprayed at night to allow the longest period of efficacy.

Diamondback Moth (*Plutella xylostella*)

Diamondback moths are an important pest of collards and can cause significant damage. Diamondback moth populations peak in March and April and again in June through August.

The adult diamondback moth is small, slender and gray-brown in color. The name 'diamondback' is derived from the appearance of three diamonds when the male species folds its wings. The female moth lays small eggs on the underside of the leaf. Typically the eggs are laid separately but occasionally can be found in groups of two or three. The larvae are about a 1/3 of an inch long, pale yellow-green and covered with fine bristles. A v-shape is formed by the spreading prolegs on the last segment of the caterpillar. When startled, the larvae will writhe around or quickly drop from the leaf on a silken line. If conditions are favorable, this moth can have four to six generations a year.

The larvae attack all stages of plant growth but their damage is most significant during the seedling stage and at harvest. Larvae attack the growing points on young plants, stunting growth and decreasing yield. The larvae will chew small holes, mostly on the underside of mature leaves, on mature plants. The larvae of the diamondback moth penetrate collards heads and feed on the plant's growing point. Collards with feeding damage or contaminated by the larvae are unmarketable.

Sampling and Treatment Thresholds: Fields should be monitored during the seedling stage, crop thinning and prior to heading. Particular attention should be given if an adjacent field has recently been harvested or disked, as the larvae will migrate from such fields. According to University of Arizona guidelines, collards should be treated prior to the formation of the harvested leaves when there is 1 larva per 50 plants[14]. Once the collards head has formed, the crop can tolerate 1 larva per 100 plants[14].

All other larvae in the lepidopterous complex should be included in this count.

Biological Control: The ichneumonid wasp (*Diadegma insularis*) will commonly parasitize *Plutella* cocoons. *Trichogramma pretiosum* is a less common parasite that attacks diamondback moth eggs. Lacewing larvae and ladybug larvae (syn: ant lions) can also be used to control small diamondback larvae. Care must be used when spraying insecticides as they can harm populations of beneficial insects. These beneficial insects, however, usually will not provide complete control of diamondback moth populations.

Chemical Control: Methomyl, spinosad and pyrethroids are the most frequently used chemistries for the control of diamondback moths. *Plutella* resistance to insecticides has been reported and is a concern in collards production.

Cultural Control: Fields should be disked immediately following harvest in order to kill larvae and pupae in the soil and prevent moth migration to adjacent crops.

Post-Harvest Control: There are no effective methods for the post-harvest control of diamondback moths.

Alternative Control: *Bacillus thuringiensis* (Bt) can be used to control diamondback moth larvae. A consideration when using *B. thuringiensis* is its tendency to break down when exposed to UV light and heat. Spraying at night will allow the longest period of efficacy. Diatomaceous earth can be used to control diamondback larvae. Neem oil soap, neem emulsion, and rotenone are less effective choices for the control of larvae.

Cabbage Looper (*Trichoplusia ni*)
Alfalfa Looper (*Autographa californica*)

Loopers are a major pest in central and southwestern deserts of Arizona. They are present all year, but their populations are highest in the fall when winter vegetables are grown.

Cabbage loopers and alfalfa loopers are very similar in appearance, which makes it difficult to differentiate between the two species. The front wings of the adult looper are mottled gray-brown in color with a silver figure-eight in the middle of the wing; the hindwings are yellow.

The female moth lays dome-shaped eggs solitarily on the lower surface of older leaves. The larvae are bright green with a white stripe running along both sides of its body. The looper moves by arching its back in a characteristic looping motion, which is also the source of its name. Loopers can have from 3 to 5 generations in one year.

Looper populations are usually highest in the fall and can cause extensive damage to collards. Loopers will attack all stages of plant growth. These larvae feed on the lower leaf surface, chewing ragged holes into the leaf. Excessive feeding on seedlings can stunt growth or even kill plants. Collards that have been damaged by looper feeding or that are contaminated with larvae or larvae frass are unmarketable.

Sampling and Treatment Thresholds: Once collards have germinated, fields should be monitored twice a week. The lower leaf surface should be checked for larvae and eggs, especially on damaged leaves. When populations are noted to be increasing, fields should be monitored more frequently. Pheromone traps are useful for measuring the migration of moths into crop fields. The presence of parasitized and virus-killed loopers should also be noted. The following are the University of Arizona suggested treatment thresholds. Prior to the formation of the harvested leaves, collards should be treated when populations have reached 1 larva per 50 plants[14]. After leaf formation, collards can tolerate 1 larva per 100 plants[14]. All other larvae in the lepidopterous complex should be included in this count.

Biological Control: There are several species of parasitic wasps, as well as, the tachinid fly (*Voria ruralis*) that will aid in the control of the looper. Care must be taken with insecticide treatment, as it can decrease populations of beneficial insects. Nuclear polyhedrosis virus is a naturally occurring virus that can assist in the control of loopers when conditions are favorable.

Chemical Control: Spinosad, tebufenozide, chlorpyrifos and pyrethroids are the most commonly used chemistries for controlling looper populations. All are foliar applied insecticides.

Cultural Control: Weeds growing within the field or surrounding the field should be controlled because they can act as hosts for loopers and other lepidopterous insects. Weeds on ditch banks and adjacent fields should be monitored for eggs and larvae during seeding. Fields should be plowed immediately following harvest to kill larvae and remove any host material.

Post-Harvest Control: There are no methods for the post-harvest control of loopers.

Alternative Control: *Bacillus thuringiensis*
(Bt) can be used to control looper populations, but is the most effective if applied when eggs are hatching and larvae small. One concern when applying *B. thuringiensis*
is its tendency to break down when exposed to UV light and heat. Spraying at night will allow the longest period of efficacy. Bt will also control other lepidopterous insects, with the exception of beet armyworms, and will not affect beneficial predators and parasites. Diatomaceous earth, neem oil soap, neem emulsion and rotenone are other methods for the alternative control of cabbage loopers.

Beet Armyworm (*Spodoptera exigua*)

The beet armyworm is a key pest that affects collards Production in Arizona. Armyworm populations are heaviest during the months of July through November; the larvae attack all stages of plant growth. In the fall, beet armyworms often migrate from surrounding cotton and alfalfa fields to vegetable crops. Armyworms also feed on weeds including; redroot pigweed (*Amaranthus* sp.), lambsquarters (*Chenopodium album*) and nettleleaf goosefoot (*Chenopodium murale*).

The forewings of the adult moth are gray-brown in color with a pale spot on the mid-front margin; the hindwings are white with a dark anterior margin. The female moth lays clumps of light green eggs on the lower leaf surface. The eggs are covered with white scales from the female moth's body, giving the eggs a cottony appearance. Prior to hatching, the eggs darken. The emergent larvae are olive green and are nearly hairless, which distinguishes them from other lepidopterous larvae that attack cole crops. The larvae have a broad stripe on each side of the body and light-colored stripes on the back. A black dot is located above the second true leg and a white dot at the center of each spiracle. The mature larvae pupate in the soil.

Young larvae feed in groups near their hatching site. As the beet armyworm feeds, it spins a web over its feeding site. Mature armyworms become more migratory and move to new plants. Many armyworms will die while traveling between plants. Armyworms can skeletonize leaves and feed on the midrib; they also consume entire seedlings. A single armyworm can attack several plants. Collards leaves that have been damaged by armyworm feeding or are contaminated by larvae are unmarketable.

Sampling and Treatment Thresholds: Weeds surrounding the field should be monitored for larvae and eggs prior to crop emergence. If population levels are high in surrounding weeds, the crop should be monitored very carefully following emergence. Pheromone traps can be used to help monitor for the presence of beet armyworms in a field. After germination, fields should be monitored twice a week. University of Arizona experts suggest treating collards prior to the formation of the harvested leaves when populations reach 1 larva per 50 plants[14]. Once the leaves have formed, collards can tolerate 1 larva per 100 plants[14]. All other larvae in the lepidopterous complex should be included in this count.

Biological Control: There are viral pathogens, parasitic wasps and predators that attack the beet armyworm. These beneficials, however, are unable to completely control armyworm populations. Caution must be used when spraying insecticides as they can harm beneficial insects.

Chemical Control: Spinosad, chlorpyrifos, tebufenozide and pyrethroids are routinely used for the control of armyworms. The best time to spray with an insecticide is when the larvae are hatching; this allows maximum control of the population. This also provides the opportunity to determine the degree of predator activity and dispersal deaths. Insecticides are more effective when applied at dusk or dawn; this is the time when armyworms are the most active. It is important to practice sound resistance management practices by alternating chemistries.

Cultural Control: Weeds growing within and surrounding the field should be controlled, as armyworms can build up in these areas. Fields should be disked immediately following harvest to kill any larvae pupating in the soil.

Post-Harvest Control: There are no effective methods for the post-harvest control of beet armyworms.

Alternative Control: Some growers use diatomaceous earth, neem oil soap, neem emulsion or rotenone for the control of beet armyworms. *Bacillus thuringiensis* is registered for controlling beet armyworms but does not provide adequate control.

Corn Earworm (Bollworm) (*Helicoverpa zea*)
Tobacco Budworm (*Heliothis virescens*)

The tobacco budworm and corn earworm occur throughout Arizona but are the most prevalent in central and western parts of the state. Budworm and earworm populations peak during the fall. These larvae attack all stages of plant growth.

The adult corn earworm moth has mottled gray-brown forewings; the hindwings are white with dark spots. The forewings of the tobacco budworm moth are light olive-green with three thin, dark bands; the hindwings are white with a red-brown border. The female moth lays white eggs separately on the plant's leaves. Twenty-four hours after they are laid, the eggs develop a dark band around the top and prior to hatching the eggs darken in color. The emergent larvae of these two species can be a variety of colors and develop stripes down the length of their body. It is difficult to differentiate between the larvae of these two species until they are older. Older larvae can be distinguished by comparing the spines at the base of the abdominal tubercles and by the presence of a tooth in the mandible.

Both species of larvae are cannibalistic, eating larvae of their own species and of other lepidopterous species, thus they tend to feed alone. Budworms and earworms are capable of killing entire stands of seedlings. In older plants, the larvae chew holes into the leaves and gouge the midrib. They may also feed on the growing point of the plant, often killing the growing tip. Once the larvae are within the head they are difficult to control with insecticides. Damaged collards leaves are unmarketable.

Sampling and Treatment Thresholds: Field monitoring should begin immediately following seed germination. Pheromone traps can be used to monitor for the presence of tobacco budworms and corn earworms. Earworms and budworms migrate from corn and cotton fields, thus it is important to carefully monitor field edges that border these fields. If eggs are discovered, it should be determined if they have hatched, are about to hatch or have been parasitized. The collards should also be checked for larvae and feeding damage. It is important to correctly identify which larvae are present, as resistance in tobacco budworms has been reported. University of Arizona guidelines suggest that before the formation of the harvested leaves, the crop requires treatment when populations reach 1 larva per 50 plants[14]. After leaf formation the crop can tolerate 1 larva per 100 plants[14]. All other larvae in the lepidopterous complex should be included in this count.

Biological Control: Some parasites and predators of earworms and budworms include; *Trichogramma* sp. (egg parasite), *Hyposoter exiguae* (larval parasite), *Orius* sp. (minute pirate bug) and *Geocoris* sp. (bigeyed bugs). These enemies are often able to reduce earworm and budworm populations. Care must be taken with insecticide treatment, as it can decrease the populations of beneficial insects. Nuclear polyhedrosis virus, a naturally occurring pathogen, also helps control populations.

Chemical Control: Insecticide treatment is more effective at peak hatching, when larvae are still young. Eggs darken just prior to hatching, which gives a good indication when to prepare to spray. This also allows the opportunity to check for the presence of predators and parasites. The best time to treat for tobacco budworms and corn earworms is mid-afternoon when the larvae are the most active. Spinosad, chlorpyrifos and pyrethroids are the most commonly used chemistries for controlling earworms and budworms.

Cultural Control: Delaying planting until after cotton defoliation will decrease larvae migration into collards fields. Due to market demands, however, it is not always possible to delay planting. Fields that are planted next to cotton fields require careful monitoring. Fields should disked following harvest to kill any larvae pupating in the soil.

Post-Harvest Control: There are no methods for the post-harvest control of corn earworms or tobacco budworms.

Alternative Control: Methods for the alternative control of budworms and earworms include; diatomaceous earth, neem oil soap, neem emulsion and rotenone.

Imported Cabbageworm (*Pieris rapae*)

The imported cabbageworm is not a common pest in Arizona, but damage caused by this pest has been recorded. The adult cabbageworm moth, called the cabbage butterfly, has white-yellow wings with black spots on the upper surface. The female moth lays rocket-shaped eggs on the lower leaf surface. The larvae are green in color with a faint yellow or orange stripe down its back and broken stripes down the sides of its body. The larvae's body is covered with numerous hairs giving the larvae a velvety appearance.

The imported cabbageworm chews large, irregular-shaped holes into the leaves. When young plants are attacked, the larvae can stunt or kill the plants. The larvae feed for 2 to 3 weeks and then attach themselves to the stem or leaf on the plant or a near by object to pupate. The presence of the larvae, larvae frass or pupae within the collards head or damage to the leaves will render the plant unmarketable.

Sampling and Treatment Thresholds: The field should be randomly checked for areas of damaged plants. Cabbage loopers, however, cause the same sort of damage as the cabbageworm. Thus it is important to also check for eggs, larvae and moths to positively identify which larvae species is causing the damage. Experts at the University of Arizona suggest treating collards prior to the formation of the harvested leaves when there is 1 larva per 50 plants[14]. Once the collards leaves have formed, the crop can tolerate 4 larvae per 25 plants[14]. All other larvae in the lepidopterous complex should be included in this count.

Biological Control: There are many natural enemies to the imported cabbageworm including; *Pteromalus puparum*, *Apanteles glomeratus*, *Microplitis plutell*a and the tachinid fly (*Voria ruralis*). There are also some viral and bacterial Diseases that will attack cabbageworms. Insecticides should be sprayed with caution as they can harm beneficial insects.

Chemical Control: Spinosad, chlorpyrifos and pyrethroids, are the most commonly used methods for controlling imported cabbageworms.

Cultural Control: Weeds growing within the field and surrounding the field can act as hosts to cabbageworms and thus must be controlled. Fields should be plowed after harvest to eliminate any larvae that may be pupating in the soil. Sanitation of equipment is important to prevent the contamination of uninfected fields.

Post-Harvest Control: There are no methods available for the post-harvest control of imported cabbageworms.

Alternative Control: *Bacillus thuringiensis*
(Bt) can be used to control cabbageworms and will not harm beneficial predaceous and parasitic insects. Bt is most effective when sprayed on young larvae. A concern when spraying Bt is its tendency to break down when exposed to UV light and heat. Spraying at night will allow for a longer period of efficacy.

1999 Insecticide Use for Controlling Lepidoptera Larvae on Arizona Grown Collards

							(# of reports)			
Active Ingredient	Label Min.*	Avg. Rate*	Label Max.*	# of Acres	% of Acres Treated	# of Reports**	By Air	AW	CL	D
Bacillus thuringiensis	0.05	0.14	1.05	84	0	8	8	5	2	1
Cypermethrin	0.05	0.10	0.1	202.8	0	9	9	6	6	1
Diazinon (**OP**)	0.25	0.48	4	12.8	0	1	1	1	1	(
Dimethoate (**OP**)	0.25	0.25	0.25	12.8	0	1	1	1	1	(
Imidacloprid	0.16	0.05	0.38	194	0	9	9	4	4	2
Methomyl (**carbamate**)	0.45	0.77	0.9	53	0	8	8	8	0	(
Permethrin	0.5	0.14	0.1	31.5	0	5	3	3	1	(
Pyrethrins	0.01	0.01	0.01	222.8	0	12	12	7	7	1
Spinosad	0.023	0.08	0.156	268	0	15	15	12	5	1

AW armyworm

CL looper

DM diamondback moth

L loopers

OP organophosphate

Note: the percentage of acres treated is listed as 0, because there are no data available as to how many acres of collards were grown in Arizona.

*Application rates are pounds of active ingredient (AI) per acre. Average rate is an average of field level rates from the ADA 1080 reports using a NAS conversion table to determine the pounds of AI in pesticide products. Maximum and Minimum rates come from product labels.

**the number of reports is the number of unique 1080 forms received with indicated AI. 1080s with multiple AIs are counted for each AI. Acres for multiple AI mixes are separately counted for each AI. % of acres treated is AI acre total divided by planted acres. Only previous year's planted acres are available.

***Up to four target pests are recorded and multiple AI applications are common. No mechanism in the 1080 forms presently exists to link specific AIs to specific target pests. For this reason, all AI/pest counts do not necessarily reflect intended efficacy.

Homoptera

APHIDS (syn: "plant lice")

Green Peach Aphid (*Myzus persicae*)
Potato Aphid (*Macrosiphum euphorbiae*)
Turnip Aphid (*Lipaphis erysimim*)
Cabbage Aphid (*Brevicoryne brassicae*)

Currently aphids do not pose a large economic threat to collards production but have the potential to become a greater issue in the future. Aphid populations peak during the months of November and December and again during February and March. Populations consist entirely of asexual reproducing females that produce live young; this allows the population to increase rapidly. When conditions are ideal, aphids have as many as 21 generations in one year. When populations become too large or food is scarce, aphids produce winged offspring that are capable of migrating to new hosts.

There are four different species of aphid that can potentially be pests to collards: green peach aphids, potato aphids, turnip aphids and cabbage aphids. These aphids may or may not have wings. Green peach aphids are light green, red or pink in color. They are found feeding on the lower surface of mature leaves and will quickly colonize younger leaves as the population increases. Potato aphids have a similar appearance to green peach aphids but are larger and form small colonies on the lower surface of new leaves. The cabbage aphid is gray-green and covered with a waxy ‘bloom’ giving the insect a gray-white appearance. Some refer to this aphid as the ‘gray aphid’. Cabbage aphids colonize the young leaves of collards. Cabbage aphids are the most common species of aphid found on collards. The turnip aphid is similar in appearance to the cabbage aphid but is not covered with a waxy ‘bloom’. These aphids form small colonies on new leaves.

Aphid feeding can cause collards leaves to distort and curl and can deplete the plant’s phloem sap. Extreme aphid feeding can deplete a plant of enough phloem sap to reduce the plant’s vigor or even kill the plant. As an aphid feeds it excretes phloem sap ("honeydew") onto the plant’s surface. This provides an ideal environment for sooty mold infection, which inhibits photosynthesis. Another concern is the viruses that green peach aphids can transmit such as mosaic viruses. Aphids are also damaging as a contaminant. The presence of aphids in collards or aphid damage to the leaves will render the leaves unmarketable.

Sampling and Treatment Thresholds: To control aphid infestations, it is essential to monitor fields frequently and prevent the growth of large populations. These pests migrate into crop fields and reproduce rapidly, quickly infecting a crop. Beginning in January, fields should be monitored no less than twice a week. Yellow waterpan traps are useful for measuring aphid movement into the field. Aphids usually appear first at the upwind field border and those borders that are adjacent to fields of cruciferous weeds and crops. In infested fields, aphids tend to occur in clusters within the field, thus it is important to randomly sample the field. The following are the University of Arizona's suggested treatment thresholds. Prior to the formation of the marketed leaves, treatment should begin when populations reach 1 aphid per 10 plants[14]. After leaf formation, collards should be treated when aphid colonization begins[14].

Biological Control: Parasitoids and predators that attack aphids are available; however, they are usually unable to completely control aphid populations. Lady beetle larvae (syn: ant lions), lacewing larvae, syriphid fly larvae; aphid parasites are some of the insects used to control aphids. Spraying of insecticides should be performed with caution as it can eliminate beneficial insects. These beneficial insects, however, can also become contaminants of collards heads.

Chemical Control: A pre-plant application of imidacloprid is the most common method used to control aphids. This insecticide has the added benefit of long-term residual control. However, this prophylactic approach to control is expensive and is applied with the assumption that the crop will receive aphid pressure. Many growers will choose to wait and apply a foliar insecticide if required. When foliar insecticides are used, the timing of application is critical. Endosulfan, dimethoate and imidacloprid are the most commonly used foliar-applied treatments. The initial treatment should occur once aphids begin to migrate into a crop field. To ensure that the harvested collards are not contaminated with aphids, it might be necessary to use repeated applications. Aphids often hide within the collards' leaves making insecticide contact difficult. If aphids only occur at the field borders or in isolated areas, border or spot applications may be sufficient to control populations. Insecticide chemistries should be alternated for good resistance management.

Cultural Control: Aphids tend to build up in weeds, particularly cruciferous weeds and sowthistle (*Sonchus asper*), therefore it is important to control weeds in the field and surrounding the field. Fields should be plowed under immediately following harvest, to eliminate any crop refuse that could host aphids.

Post-Harvest Control: There are no methods for the post-harvest control of aphids.

Alternative Control: Some growers use; insecticidal soaps, neem oil soap, neem emulsion, pyrethrins, rotenone dust, plant growth activators, elemental sulfur, garlic spray and diatomaceous earth to control aphid populations.

WHITEFLIES

Sweetpotato Whitefly (*Bemisia tabaci*)
Silverleaf Whitefly (*Bemisia argentifolii*)

Whiteflies do not pose a large threat to collards Production in Arizona but are capable of causing damage. Whiteflies are also capable of transmitting viruses.

The adult whitefly is 1/16" in length and has a white powder covering its body and wings. The female whitefly lays small, oval, yellow eggs on the undersurface of young leaves. The eggs darken in color prior to hatching. The emergent nymph travels about the plant until it finds a desirable minor vein to feed from. The nymph does not move from this vein until it is ready to pupate. Whiteflies can have numerous generations in one year.

Whitefly infestations are usually the heaviest during the fall. Colonization of the crop can begin immediately following germination with whiteflies feeding on the cotyledons. Whiteflies migrate from cotton, melon and squash fields, as well as, from weed hosts. Collards planted downwind from these plants are particularly susceptible.

Whitefly feeding removes essential salts, vitamins and amino acids required by the collards plant for proper growth. This feeding results in; reduced plant vigor and can delay harvest if not controlled at an early stage. Whitefly feeding can also cause the leaves to acquire a silvery appearance. As with aphids, the phloem sap that whiteflies excrete onto the collards' surface creates an ideal environment for sooty mold infection. Whiteflies also contaminate the harvested collards leaves, making it unmarketable.

Sampling and Treatment Thresholds: The best way to prevent a whitefly infestation is to inhibit initial colonization. Whitefly counts should be performed early in the morning when the insects are the least active. Once whiteflies become active they are difficult to count. During the mid-morning, fields should be monitored for swarms of migrating whiteflies. Experts at the University of Arizona suggest that if a soil-applied insecticide is not used, collards should be treated when populations reach 5 adults per leaf[14].

Biological Control: Parasitoid wasps (*Eretmocerus* sp.) can be used to control whitefly populations, however they only parasitize immature whiteflies. Lacewing larvae and ladybug larvae (syn: ant lions) are also used for the control of whiteflies. These insects are very sensitive to pyrethroids and other insecticides, thus it is important to determine the severity of pest pressure and the activity of beneficial insects before spraying.

Chemical Control: If the crop is planted in August or September when populations are at their greatest a soil-applied prophylactic insecticide, such as imidacloprid, is often applied. If collards are planted after whitefly populations have declined, foliar-applied insecticides can be used as necessary. Imidacloprid, endosulfan and dimethoate are the most commonly used foliar insecticides. Tank-mixing insecticides helps control whiteflies, as well as, preventing the development of insecticide resistance. When spraying it is important to achieve complete crop coverage, this will provide the best control of whiteflies. There is a strong dependence on imidacloprid to control whiteflies; this creates concerns of product resistance. As well, whitefly resistance to organophosphates and pyrethroids has been noted in the past, thus resistance management is important.

Cultural Control: Whitefly populations are most active in early September and tend to migrate from defoliated and harvested cotton. Delaying planting until populations have begun to decrease and temperatures are lower will help manage whitefly infestation. Delaying planting, however, is not always a feasible option. Whiteflies build up in weeds, especially cheeseweed (*Malva parviflora*), thus it is important to control weeds in the field and surrounding the field. Crop debris should be plowed under immediately following harvest to prevent whitefly build up and migration to other fields.

Post-Harvest Control: There are no methods for the post-harvest control of whiteflies.

Alternative Control: Some growers use; neem oil soap, neem emulsion, pyrethrins, insecticidal soaps, rotenone, elemental sulfur, garlic spray and diatomaceous earth to control whiteflies.

Thysanoptera

THRIPS

Western Flower Thrips (*Frankliniella occidentalis*)
Onion Thrips (*Thrips tabaci*)

Thrips are present all year, but their populations increase in the early fall and late spring. Thrips spread from mustard, alfalfa, onion and wheat fields, surrounding weedy areas and unirrigated pastures. In the spring, thrips pose a great threat to the production of collards.

Thrips species are small (1/20-1/25 in.), slender and pale yellow-brown in color. The two species are similar in appearance, which can make it difficult to distinguish between them. It is important, however, to identify which species of thrips are present because western flower thrips are more difficult to control. Consulting a specialist is best if one is unsure. Female thrips lay small, white, bean-shaped eggs within the plant tissue. The hatched nymphs are similar in appearance to the adults, but smaller in size and lack wings. Thrips will pupate in the soil, or in the leaf litter below the plant.

Thrips feeding wrinkles and deforms leaves and stunts growth. Feeding can also cause brown scars on the collards leaves. Extreme damage causes leaves to dry and fall off the plant. Black dust, the thrips feces, on the leaves distinguishes this damage from windburn or sand burn. Thrips present in harvested collards are considered a contaminant and leaves damaged by thrips feeding are not marketable.

Sampling and Treatment Thresholds: Sticky traps are a good monitor of thrips migration into a field. When inspecting for thrips, the folded plant tissue must be carefully examined, as this is where thrips prefer to hide. It is estimated that for every 3 to 5 thrips observed there are three times as many that are undiscovered. University of Arizona experts suggest treating collards prior to the formation of the harvestable leaves when populations reach 1 thrips per 10 plants[14]. After leaf formation, the crop should be treated when the population reaches 1 thrips per 25 plants[14].

Biological Control: Lacewing larvae, ladybug larvae (syn: ant lions) and the minute pirate bug can be used to provide control of thrips. Insecticides must be sprayed with care as they can harm these beneficial insects.

Chemical Control: Treatment should begin when thrips populations are still low and when tissue scarring begins. For more effective control, applications should be made during the afternoon because this is when thrips are the most active. Studies have shown that even the most effective insecticides do not decrease thrips populations; they are merely able to maintain the population size. This is important to consider when an application date is being chosen. The number of applications a crop stand requires will vary according to the residual effect of the chemical and the rate of thrips movement into the crop field. The size of the plant and the temperature will also effect the degree of control. The more mature a plant is the more folds and crevices it has for thrips to hide in and avoid insecticide contact.

Pyrethroids will not control thrips nymphs but will suppress the adults. Pyrethroids should only be used in a tank mix to prevent chemistry tolerance in thrips. Dimethoate, spinosad and methomyl will provide control for nymphs but not adults. Currently there are no insecticides that provide complete control of thrips.

Cultural Control: Cultural Practices do not effectively control thrips because thrips will rapidly migrate from surrounding vegetation.

Post-Harvest Control: There are no methods for the post-harvest control of thrips.

Alternative Control: Some growers use pyrethrins and elemental sulfur to control thrips.

Other Contaminents (syn: 'Trash Bugs')

False Chinch Bug (*Nysius raphanus*) (Hemiptera)
Lygus Bug (*Lygus hesperus*) (Hemiptera)
Three-Cornered Alfalfa Hopper (*Sissistilus festinus*) (Homoptera)
Potato Leafhopper (*Empoasca fabae*) (Homoptera)

The false chinch bug is gray-brown with a narrow, 1/8" long body and protruding eyes. False chinch bugs tend to build up in cruciferous weeds.

The lygus bug varies in color from pale green to yellow-brown with red-brown or black markings. This insect is ¼" long and has a flat back with a triangular marking in the center. These insects are commonly found in cotton, safflower and alfalfa fields, as well as, on weed hosts.

The three-cornered alfalfa hopper has a ¼"long, light-green wedge-shaped body. The potato hopper has an elongated body and varies from light green to light brown in color. Both species have well-developed hind legs, allowing them to move quickly. These pests are common in alfalfa and legume fields as well as weed hosts. Leafhoppers are not commonly found in collards fields.

These contaminants normally do not cause direct damage to collards; they are more of concern as a contaminant. Populations of these insects often increase when the growing season experiences high rainfall and desert vegetation and cruciferous weeds flourish. These insects also build up when collards is planted near alfalfa or cotton.

Sampling and Treatment Thresholds: Experts at the University of Arizona suggest that collards should be treated prior to the formation of the harvested leaves when populations reach 10 contaminant insects per 50 plants[14]. Once the leaves have formed, collards should be treated when populations reach 1 contaminant insect per 25 plants[14].

Biological Control: There are no methods for the biological control of contaminant insects.

Chemical Control: Since these insects generally do not cause physical damage to collards, chemical control is not normally required until head formation begins. Growers typically spray as close to harvest as possible to ensure the collards are not contaminated. Dimethoate, methomyl, chlorpyrifos, diazinon and pyrethroids such as cypermethrin are the most commonly used insecticides for controlling contaminant insects.

Cultural Control: It is important to control weeds that can harbor contaminants, in the field and surrounding the field. Alfalfa should not be cut until the collards field has been harvested, this will prevent insect migration into the collards field.

Post-Harvest Control: There are no methods for the post-harvest control of contaminant insects.

Alternative Control: Some growers use neem oil, garlic spray, rotenone and pyrethrins to control contaminant insects.

1999 Insecticide Usage on Collards Grown in Arizona

							(# of reports)								
Active Ingredient	Label Min.*	Avg. Rate*	Label Max.*	# of Acres	% of Acres	# of Reports**	By Air	Aph.	LM	Lep.	SE	Thp.	WF	U
CENTRAL														
Bacillus thuringiensis	0.05	0.14	1.05	84	0	8	8	3	0	13	2	0	0	
Cypermethrin	0.05	0.10	0.1	202.8	0	9	9	3	0	16	5	2	3	
Diazinon (**OP**)	0.25	0.48	4	12.8	0	1	1	0	0	2	1	0	0	
Dimethoate (**OP**)	0.25	0.25	0.25	12.8	0	1	1	0	0	2	1	0	0	
Imidacloprid	0.16	0.05	0.38	198	0	10	10	6	0	13	4	0	4	
Methomyl (**carbamate**)	0.45	0.77	0.9	53	0	8	8	0	0	14	0	0	1	
Permethrin	0.5	0.13	0.1	12	0	2	2	0	0	4	0	0	0	
Pyrethrins	0.01	0.01	0.01	222.8	0	12	12	5	0	19	6	2	3	
Spinosad	0.023	0.08	0.156	268	0	15	15	3	1	28	5	1	3	
WESTERN														
Imidacloprid	0.16	0.05	0.38	8.2	0	1	1	1	0	0	0	1	0	
Permethrin	0.05	0.15	0.1	19.5	0	3	1	0	0	3	0	0	0	
EASTERN														
Esfenvalerate	0.02	0.044	0.05	30	0	1	1	0	0	0	0	0	0	

Aph. Aphids

LM Leafminer

Lep. Lepidoptera larvae

SE Stand Establishment Insects = ants, crickets, flea beetles, darkling beetles, grasshoppers

Thp. Thrips

WF Whitefly

OP Organophosphate

Note: the percentage of acres treated is listed as 0, because there are no data available as to how many acres of collards were grown in Arizona.

*Application rates are pounds of active ingredient (AI) per acre. Average rate is an average of field level rates from the ADA 1080 reports using a NAS conversion table to determine the pounds of AI in pesticide products. Maximum and Minimum rates come from product labels.

**the number of reports is the number of unique 1080 forms received with indicated AI. 1080s with multiple AIs are counted for each AI. Acres for multiple AI mixes are separately counted for each AI. % of acres treated is AI acre total divided by planted acres. Only previous year's planted acres are available.

***Up to four target pests are recorded and multiple AI applications are common. No mechanism in the 1080 forms presently exists to link specific AIs to specific target pests. For this reason, all AI/pest counts do not necessarily reflect intended efficacy.

Diseases

Fungal Diseases

(3, 7, 10, 11, 13, 19, 20, 24, 25)

Damping-Off (*Pythium* sp., *Rhizoctonia solani*)

In Arizona, damping-off is occasionally observed in collards fields. Damping-off is a soil borne fungus that attacks germinated seedlings that have not yet emerged or have just emerged. Cool, wet weather promotes infection by most *Pythium* species, where as cool to moderate weather promotes *Rhizoctonia* infection. Fields that have poor drainage, compacted soil and/or high green organic matter are the most susceptible to damping-off. The damping-off fungi will not affect plants that have reached the three to four-leaf stage.

Damage usually occurs at soil level, leaving lesions in the stem tissue. The tissue becomes dark and withered, the weak support causes the seedling to collapse and die. *Pythium* can also attack the seedling's roots, causing them to turn brown and rotten.

Biological Control: *Gliocladium virens* GL-21 is the only biological method available for controlling *Pythium* and *Rhizoctonia* induced damping-off. *G. virens* is a fungus that antagonizes *Pythium* and *Rhizoctonia*. In the greenhouse *G. virens* provides good control of damping-off; in the field the control that *G. virens* provides is more variable.

Chemical Control: Metam sodium and metam-potassium are fumigants registered for use on *Pythium* and *Rhizoctonia*; however, they are a very costly method of control and generally not considered a viable option. Mefenoxam is the only other chemical method available for controlling *Pythium*-induced damping-off. This fungicide works best when used as a preventative treatment, being applied before disease becomes apparent. Typically mefenoxam is applied in a band over the seed row, either pre-plant incorporated or preemergence. There are no other chemistries registered in Arizona to treat for *Rhizoctonia*-induced damping-off of collards. There are also no registered seed treatments in Arizona for controlling damping-off of collards. Most growers, however, do not treat for damping-off as this disease is not currently a large threat to collards in Arizona.

Cultural Control: All residues from the previous crop should be plowed under and completely decomposed before planting collards. It is best to plant when the soil is warm, as this will speed germination and allow the crop to quickly reach a resistant stage of growth. Overhead or sprinkler irrigation are the best methods for promoting rapid germination. It is very important to manage water application and avoid over saturating the field. Fields should be properly drained and low spots should be eliminated to avoid water accumulation. When directly seeding it is important not to plant too deep as this will slow emergence, increasing the seedling's susceptibility to damping-off. It is important to avoid stressing the crop, as this will make it more susceptible to damping-off.

Post-Harvest Control: There are no effective post-harvest measures for the control of damping-off.

Alternative Control: Some growers spread compost on the soil to control pathogens.

Downy Mildew (*Peronospora parasitica*)

Of the potential fungal Diseases, downy mildew poses the largest threat to the production of collards in Arizona. Downy mildew thrives in cool, humid weather, such as that which is typical of the winter growing season in western Arizona. This weather promotes spore formation and spore dispersal, as well as, plant infection. When conditions are favorable, *Peronospora parasitica* can spread rapidly. The fungus also produces resting spores, which can survive in the soil or crop residue until the following season. *P. parasitica* is spread by; wind, rain, infected seed and infected transplants.

Plant infection begins with the growth of gray-white fungi on the lower leaf surface. Damage occurs on both leaf surfaces, beginning with chlorotic lesions that later turn purple and eventually brown. Young leaves sometimes dry and drop off, while older leaves generally remain on the plant and develop a papery texture. Downy mildew can decimate large numbers of seedlings. Severe infections of mature collards can result in decreased photosynthesis, stunted plants and reduced yield. Downy mildew is a systemic disease that results in darkened areas and/or black streaks in the stem. This damage to the stem and leaves causes the plant to be susceptible to secondary infections. Any damage to the collards leaves, renders the plant unmarketable.

Biological Control: There are no biological methods for controlling downy mildew.

Chemical Control: Fosetyl-aluminum, phosphorus acid and copper-based fungicides are the only chemistries available for controlling downy mildew. Fosetyl-aluminum and phosphorous acid are systemic treatments; copper products are contact fungicides. Downy mildew is best controlled when treatment is used as a preventative measure, rather than waiting for the onset of disease symptoms. If there is heavy rain, one can anticipate downy mildew. If environmental conditions remain favorable for disease development, multiple applications may be required. It is important to alternate fungicides or apply fungicide mixtures to ensure proper resistance management.

Cultural Control: Cruciferous weeds that can act as a host for downy mildew must be controlled. Fields should be plowed under following harvest to promote the decomposition of infected plant debris. It is important to rotate to a non-cole crop the subsequent year. Overhead irrigation should be avoided, as this aids in the spread of *P. parasitica*.

Post-Harvest Control: There are no methods for the post-harvest control of downy mildew.

Alternative Control: Some growers use milk and hydrogen peroxide to control downy mildew. Spreading compost on the soil is also used for the control pathogens.

Powdery Mildew (*Erysiphe cichoracearum*)

Powdery mildew can become a significant threat to collards production during the spring at the end of the growing season. *Erysiphe cichoracearum* thrives in dry, mild temperatures. The fungus prefers low levels of light, thus it tends to develop on lower leaves. The fungus produces wind-dispersed spores that are capable of traveling long distances. Disease development occurs on both leaf surfaces, beginning with small spots of white, powdery fungal growth. Often the older healthy leaves are the first to be infected. As disease progresses the leaves become covered with white, powdery spores. The leaves may become chlorotic and eventually brown; sometimes they become dried and curled. Powdery mildew infection stunts growth and reduces plant marketability.

Biological Control: There are no biological methods for controlling downy mildew.

Chemical Control: Potassium bicarbonate and sulfur are registered for controlling powdery mildew on collards grown in Arizona. For the best protection, fungicides should be applied before disease onset. Potassium bicarbonate and sulfur are both contact fungicides. For the best results with potassium bicarbonate, the fungicide should be applied frequently and good coverage is essential.

Cultural Control: There are no methods for the cultural control of powdery mildew.

Post-Harvest Control: There are no methods for the post-harvest control of downy mildew.

Alternative Control: Spreading compost on the soil is sometimes used to control pathogens.

Alternaria Leafspot (*Alternaria* sp.)

Alternaria leafspot, also known as alternaria blight, can cause economic damage to collards grown in Arizona. Typically this disease occurs during a wet winter during the early part of the growing season. Spores can survive in weed hosts and plant debris for long periods of time. The fungal spores are wind and rain dispersed and require free water for germination to occur.

Symptoms begin as a small dark spot on the leaf. As the disease progresses, concentric rings will develop around the spot creating a bulls-eye pattern. Eventually, velvety, brown spore-bearing growths develop within the spots. If left untreated, alternaria leafspot will eventually defoliate a plant. *Alternaria* spores can survive for long periods of time on plant debris or within infected seeds. Any damage to the collards leaves render it unmarketable.

Biological Control: There are no biological methods for controlling *Alternaria*.

Chemical Control: Copper fungicides are the only chemistires labeled for controlling *Alternaria* in Arizona. These fungicides are foliar applied and are most effective when applied as a protectant before the onset of disease. Copper is a contact fungicide.

Cultural Control: *Alternaria*

can be passed on to the next generation in infected seed, therefore it is important to be certain that seed is disease-free. *Alternaria* also persists in the soil; thus cole crops should not be planted more than once in four years to avoid disease carryover. It is important to clean equipment between uses in different fields, to prevent contamination of an uninfected field. Controlling cruciferous weeds that can act as a disease host will prevent the transmission of *Alternaria* from these weeds to collards.

<u>Post-Harvest Control</u>: There are no post-harvest methods for controlling *Alternaria*.

<u>Alternative Control</u>: Some growers spread compost on the soil to control pathogens. Neem oil is also registered for the control of *Alternaria*.

Bacterial Diseases

(7, 19, 20, 21, 22)

Bacterial Soft Rot (*Erwinia* sp.)

In Arizona bacterial soft rot is occasionally reported to occur on collards. Bacterial soft rot does occur in the field, but is more common during post-harvest storage. Infection often occurs on collards that is stored at warm temperatures, or if heat is allowed to accumulate in the storage containers. This disease is capable of destroying an entire lot of collards.

Open wounds on the plant provide an entry for the bacterium. A plant that was infected with downy mildew or has been damaged by freezing or insects is particularly susceptible to bacterial soft rot. The architecture of the collards head also contributes to bacterial infection. The crevices formed by leaves are capable of holding water, creating an ideal environment for bacterial growth. The initial sign of infection is water soaked spots on the plant. Once inside collards the bacterium spreads rapidly. The bacterium dissolves the middle lamella that holds cells together and causes the inner contents of the cell shrink. The infected portions of the plant can develop a brown color and the wet rot is accompanied by a foul odor.

Machinery, insects, rain, irrigation and humans spread *Erwinia*.

<u>Biological Control</u>: There are no available methods for the biological control of bacterial soft rot.

<u>Chemical Control</u>: There are no methods for the direct chemical control of *Erwinia*; however, insecticides can control the insects that damage collards leaving it susceptible to bacterial infection.

<u>Cultural Control</u>: Crops should be cultivated carefully to prevent damage to the plant that could provide an entryway for bacterial infection. It is important to control weeds in and around the field that could act as a host to *Erwinia*.

<u>Post-Harvest Control</u>: Collards should be handled carefully to avoid bruising and wounding that will leave the plant susceptible to infection. Plants must be thoroughly cleaned with a chlorine wash and stored at a low temperature, typically 40 °F. It is important to keep the storage facility free of soft rot bacteria by immediately destroying any infected plants and maintaining a clean facility.

<u>Alternative Control</u>: Some growers spread compost on the soil to control pathogens. There are no alternative control methods that can be utilized during post-harvest storage.

1999 Fungicide Usage on Collards Grown in Arizona

							(# of reports)		
Active Ingredient	Label Min.*	Avg. Rate*	Label Max.*	# of Acres	% of Acres Treated	# of Reports**	By Air	Downy Mildew	*Pythium*
CENTRAL									
Fosetyl-Al	1.6	2.43	4	10	0	1	1	1	0
WESTERN									
Mefenoxam	0.5	0.50	1	5	0	1	0	0	1
Neem Oil	0	0.96	0	13.5	0	1	1	0	0

Note: the percentage of acres treated is listed as 0, because there are no data available as to how many acres of collards were grown in Arizona.

*Application rates are pounds of active ingredient (AI) per acre. Average rate is an average of field level rates from the ADA 1080 reports using a NAS conversion table to determine the pounds of AI in pesticide products. Maximum and Minimum rates come from product labels.

**the number of reports is the number of unique 1080 forms received with indicated AI. 1080s with multiple AIs are counted for each AI. Acres for multiple AI mixes are separately counted for each AI. % of acres treated is AI acre total divided by planted acres. Only previous year's planted acres are available.

***Up to four target pests are recorded and multiple AI applications are common. No mechanism in the 1080 forms presently exists to link specific AIs to specific target pests. For this reason, all AI/pest counts do not necessarily reflect intended efficacy.

Viral Diseases

(10, 25, 26)

Generally speaking, viral Diseases are not a common occurrence in cole crops grown during Arizona's winters. Mosaic viruses such as cauliflower mosaic and turnip mosaic viruses can occur in collards stands but their occurrences are rare. These viral Diseases cause the collards's leaves to develop a yellow/light green/dark green mottled appearance. Necrotic areas can also develop. When infection is severe and occurs early in plant development, it can decrease plant vigor. Any discoloration of the leaves of collards will render the leaves unmarketable. Green peach aphids and whiteflies are both capable of transmitting viral Diseases.

Biological Control: There are no biological methods for directly controlling viruses, however biological methods can be utilized to control virus vectors e.g. aphids and whiteflies. Controlling virus vectors, however, is generally ineffective because it only requires a few insects to spread viral Diseases.

Chemical Control: Viruses cannot be chemically controlled. The insects that spread viruses, however, can be controlled e.g. aphids, whiteflies. This method of control, however, is inefficient because it only requires a few insects to spread viral disease.

Cultural Control: Only planting disease-free seed and resistant cultivars will help control viral infections. Controlling weeds that can serve as hosts for viral Diseases is crucial. It is also important to avoid stressing the plant, i.e.) supply an adequate amount of water and fertilization. All plant residues should be plowed into the soil and promote their decomposition.

Post-Harvest Control: There are no available methods for the post-harvest control of viruses.

Alternative Control: There are no available methods for the alternative control of viruses.

Nematodes

(Various species)
(10, 11, 13, 27)

Nematodes are not a major pest of collards in Arizona. Due to the cool soil temperature, nematodes are relatively inactive during the winter months that vegetable crops are grown. Cool soil temperatures also slow the nematode's life cycle. If collards are grown when weather is warmer nematodes can pose a threat.

The female nematode lays her eggs on the plant and soil. Larvae hatch from the eggs and pass through three larval stages before becoming sexually mature adults. The hatched larvae enter the roots, and travel between and through the cells to the differentiating vascular tissue and feed on the cellular contents of the roots.

Sampling and Treatment Thresholds: Scouting should begin far enough in advance of planting to allow a pre-plant treatment if an infestation is discovered. Nematode infestations will often occur in isolated areas within the field. Areas where plants show symptoms should be specifically checked but random sampling should also be performed. The threshold at which a field should be treated is undetermined; however, when populations occur in soils that are sandy, sandy loam, loamy sand, or when populations are large the field should be treated. If infestations are in localized areas, spot fumigation can be used to reduce cost.

Biological Control: Some growers use *Stienernema carpocapsae*, a species of parasitic nematode, to decrease nematode pest populations. This species of nematode does not directly attack nematodes but does compete with them. Some growers have had success decreasing nematode populations with this method, but the results are inconsistent. *Myrothecium verrucaria* has been used with some success. *M. verrucaria* can be applied pre-plant, at planting or post-planting, but should not be applied directly to the foliage and must be incorporated.

Chemical Control: Chemical applications to a field are incapable of eradicating a nematode population; they will only reduce the population. Nematodes, however, are rarely a large enough threat in collards fields to warrant the expense of a chemical treatment. If treatment is necessary, fumigants are often used. The soil, however, must be properly prepared by plowing under all crop residues and allowing it to completely decompose. Decomposition can take as long as a month, but additional plowing or disking will speed decay. If this is not done prior to fumigation, the fumigant cannot properly penetrate debris and large soil clods and cannot kill the nematodes. The field must be at 50% capacity and the soil temperature should range between 50-80°F for fumigation to be the most successful. The amount of time that must lapse between fumigation and planting varies depending on the product used, soil temperature, soil moisture and the species of nematode present.

1,3 –dichloropropene is a popular choice for nematode control because it is inexpensive and will also control some fungal Diseases. This chemical must be used 1 to 2 weeks prior to planting due to its phytotoxicity. Metam-sodium is a fumigant that is sometimes used to control nematode populations and has the added benefit of also controlling some species of weeds and some fungal Diseases. Metam-sodium, however, is considerably more expensive than 1,3 –dichloropropene and is phytotoxic. Tarping is sometimes used when applying metam-sodium to prevent gas escape from the soil.

Cultural Control: Rotation to non-susceptible crops will help reduce nematode populations. It is important when planting a non-susceptible crop to control weeds that can act as nematode host. Summer fallowing and disking the soil during this fallow period can be used to reduce nematode populations, but it is a costly method of control. Any equipment that is used in an infested field should be carefully cleaned before being used in another field. It is important that the collards receives the appropriate amount of fertilizer and water to reduce plant stress, thus reducing their susceptibility to nematodes.

Post-Harvest Control: There are no effective methods for the post-harvest control of nematodes.

Alternative Control: Chicken manure can be used to control nematode populations. The efficacy of other types of manure is questionable.

Vertebrate Pests

(10, 11)

Birds can be very destructive of crops. Horned larks, blackbirds, starlings, cowbirds, grackles, crowned sparrows, house sparrows and house finches frequently eat planted seeds and seedlings. Frightening devices (visual and acoustical), trapping, poisoned baits and roost control can be used to control birds. Pocket gophers can be destructive to collards by eating and damaging the roots when they dig their burrows. The mounds that gophers produce while digging their burrows can be damaging to agricultural equipment and can disrupt irrigation furrows. Some methods for controlling gophers include controlling food sources (weeds), fumigation, flooding, trapping and poisoning. Ground squirrels are known to damage irrigation ditches and canals, as well as, feed on collards seedlings. These pests can be controlled by fumigation, trapping and poisoning. It is best to poison squirrels in their burrows to prevent the poisoning of predatory birds. There are several species of mice that can be pests of vegetable crops; they can be controlled by weed control, repellents and occasionally with poisoning. Wood rats sometimes pose a threat to the crop and can be controlled by exclusion, repellents, trapping, shooting, toxic baits. Raptors, kestrels and burrowing owls are all helpful for the control of rodent populations. Rabbits that infest fields can cause damage to collards. Rabbits can be controlled by habitat manipulation, exclusion, trapping, predators (dogs, coyotes, bobcats, eagles, hawks etc), repellents and poisons. In Arizona cottontails are classified as a small game species and state laws must be observed to take this species. Jackrabbits are classified as nongame species, but a hunting license or depredation permit is required to take the species. Elk, whitetail deer and mule deer can cause severe grazing damage to vegetable crops. Deer and elk, however, are classified as game species and require special permits to remove them. Fencing can be used for deer control; frightening devices and repellents provide some control. Feral horses and burros also cause damage to collards, but are protected by Arizona State laws.

Abiotic Diseases

(10)

There are a number of Abiotic Diseases that collards can suffer from that can affect the crop yield and often have symptoms similar to those caused by pathogens or insect pests.

Although collards are relatively tolerant of cold temperatures the leaves of the head can experience cold injury. The outer leaves that are damaged, however, can be removed and have little effect on the crop value. Cold injury will also leave the plant susceptible to secondary infections.

Winds that are strong and carry sand can abrade the leaves and make them susceptible to secondary infections. When the leaves heal themselves, it results in thickened, discolored areas that can be misidentified as pathogen infection. Wind can also severely damage seedlings, pinching the stem and collapsing them.

High salt concentrations in the soil can also be injurious to collards. Symptoms include; stunted plants, thick, dark leaves, yellowing or burning at the leaf margin and roots that are orange in color and rough in appearance. Salt may also inhibit seed germination.

Nutrient deficiencies can cause collards damage resulting in stunted plants, chlorosis and leaf spotting. Nitrogen, phosphorus and molybdenum are the most common element deficiencies to cause injury. Soil and plant tissue should be sampled regularly to determine if deficiencies are present. It is usually not possible, however, to replenish an element after the stand is established.

Weeds

(3, 7, 10, 11, 13, 28, 29)

Weeds are a threat to the cultivation of any crop. They compete with the crop for sunlight, water and nutrients. Control of weeds, especially cruciferous weeds, is fundamental for pest management in a collards field. Weeds can host a variety of Diseases and pests that can be transmitted to collards. Weed control is most important during the first 30 days of plant establishment, after this period collards are better able to compete with weeds. As well, the canopy created by the collards stand shades the underlying soil and inhibits the germination of additional weed seeds. The planting date can also give collards the advantage. Fields planted when summer weeds are dying back, but before winter weeds have begun to germinate, have decreased weed competition. It is essential that weeds are destroyed before they flower and produce seed. One plant can produce 1000s of seeds.

Weeds present at harvest can contaminate harvested collards. Weeds present at harvest will slow down the harvesting crew who will be forced to search through the weeds for the desired crop.

The summer weeds are the weeds most commonly found in Arizona between the months of August and October. Common summer broadleaf weeds include; pigweed (*Amaranthus* sp.), purslane (*Portulaca oleracea*), lambsquarters (*Chenopodium album*) and groundcherry (*Physalis wrightii*). Common summer grasses include; barnyardgrass (*Echinochloa crusgalli*), cupgrass (*Eriochloa* sp.), junglerice (*Echinochloa colonum*) and sprangletop (*Leptochloa* sp.). The winter weeds are the weeds most commonly found in Arizona between the months of November and March. Common winter broadleaf weeds include; black mustard (*Brassica nigra*), wild radish *(Raphanus sativus*), shepherdspurse *(Capsella bursa-pastoris*), London rocket (*Sisymbrium i*rio), cheeseweed (*Malva parviflora*), sowthistle (*Sonchus oleraceus*), prickly lettuce (*Lactuca serriola*), knotweed (*Polygonum* sp.), annual yellow sweet clover (*Melilotus indicus*) and nettleleaf goosefoot (*Chenopodium murale*). Common winter grasses include; canarygrass (*Phalaris minor*), annual blue grass (*Poa annua*), wild oats (*Avena fatua*) and wild barley (*Hordeum* sp.).

A yearly record should be kept detailing what weed species are observed in each field. This is important because herbicides usually work best on germinating weeds. To choose the appropriate herbicide, one must know what weeds are present before they have germinated.

Biological Control: There are no effective methods available for the biological control of weeds.

Chemical Control: Chemical control of weeds is difficult as many of the weeds are in the same family as collards (Brassicaceae). It is challenging to adequately control weeds while ensuring crop safety. It is important to correctly identify the weed species, as different weeds have different chemical tolerances. Most postemergence herbicides do not have a wide range of weed control and are especially poor at controlling cruciferous weeds such as wild mustard and shepherd’s purse. Preemergence herbicides are more effective for the control of weeds in a crucifer crop field. Another option is to use a non-selective herbicide such as glyphosate to sanitize the field prior to collards emergence.

Trifluralin and bensulide are the most commonly used preemergence herbicides. Bensulide is usually sprayed behind the planter in a band over the seed row; however, it can also be broadcast spayed or chemigated. Irrigation is required to activate this chemistry; sprinkler irrigation is often utilized. This herbicide is effective against grass weeds and will also control some small-seeded broadleaf weeds. Trifluralin is usually sprayed prior to planting and must be mechanically incorporated. This herbicide is effective on grass weeds, and has efficacy against some small-seeded broadleaf weeds. Trifluralin usually gives better broadleaf weed control than bensulide. Oxyfluorfen is an effective preemergence broadleaf herbicide but has little effect on grass weeds. As well, oxyfluorfen is only registered for use on fallow beds and has a 120 day plant back restriction before collards can be seeded. Oxyfluorfen has little soil residual, thus it is not a practical choice for collards production. DCPA will control many of the small-seeded broadleaf and grass weeds. This is a surface applied, preemergence herbicide. Sethoxydim is the only available postemergence herbicide. This herbicide has good grass control but has no efficacy against broadleaf weeds. Pelargonic acid can be used for spot treatment on postemergence crops.

Herbicides can cause injury to collards if not applied correctly and carefully. Injury may result from; spray drift, residue in the soil from a previous crop, accidental double application to a row, using the wrong herbicide or using a rate that is too high. Herbicide injury can cause leaf spotting or yellowing that can be misidentified as pathogen injury or nutrient deficiency. Soil, water or plant tissue test can be used to identify herbicide injury.

Cultural Control: Collards should be encouraged to grow quickly and establish the stand, which will increase the ability of collards to out compete any weeds present in the field. Precise planting, a regular water supply and appropriate fertilization will help increase the ability of collards to compete with weeds.

Purchasing seed that is guaranteed to be weed-free will help prevent the introduction of new weed species into a field. It is also important to maintain field sanitation by always cleaning equipment used in one field before it is used in another and ensuring that any manure that is used is weed seed free. Contaminated irrigation water from canals, reservoirs and sumps can also spread weed seed. Irrigation ditches, field borders and any other uncropped area should be maintained weed-free.

A properly leveled field is important to prevent the build up water in isolated areas, especially when utilizing furrow irrigation. This water build up will promote the germination of weeds that are favored by wet conditions.

Delaying planting until the time when summer weeds are declining but before winter weeds begin to germinate will decrease the amount of weed competition. However, due to market demands this control method is not always feasible.

Another method used to control weeds is to till the field, form beds and irrigate prior to planting. This will encourage the germination of the weed seeds. The field can then be sprayed with a nonselective herbicide or rotary hoed to kill the weeds. After the weeds have been destroyed, the collards are planted. Disking will eliminate germinated weeds but will also expose new weed seed that may germinate and cause a second flush of weeds.

Cultivation and hoeing can be used to control weeds in a planted field but should be done with care due to the shallow root system of collards. Rows and beds must be carefully planted and the cultivation equipment must be carefully aligned. Fields should be disked after harvest to eliminate any weeds present and to prevent the weeds from flowering and spreading seed.

Solarization involves laying down clear plastic sheeting over a field, usually a minimum of 60 days prior to planting. This sheeting will super heat the soil, burning and killing weed seeds and germinating weed seedlings.

Rotating to a non-crucifer crop will allow the use of herbicides that are more effective for the control of crucifer weeds. Crop rotation also promotes different Cultural Practices and planting times that will aid in weed control.

Post-Harvest Control: There are no methods for the post-harvest control of weeds.

Alternative Control: There are no alternative methods available for controlling weeds.

1999 Herbicide Usage on Collards Grown in Arizona

							(# of reports)		
Active Ingredient	Label Min.*	Avg. Rate*	Label Max.*	# of Acres	% of Acres Treated	# of Reports**	By Air	Broadleaf	Unsp
CENTRAL									
Trifluralin	0.5	0.46	0.75	99.5	0	3	0	1	[illegible]
WESTERN									
Bensulide (**OP**)	5	1.00	6	4.49	0	1	0	0	

Pronamide	1	0.40	2	4.49	0	1	0	0	

Note: the percentage of acres treated is listed as 0, because there are no data available as to how many acres of collards were grown in Arizona.

Unspecified typically refers to weeds that were treated at the germination stage or seedling stage with a general weed control.

*Application rates are pounds of active ingredient (AI) per acre. Average rate is an average of field level rates from the ADA 1080 reports using a NAS conversion table to determine the pounds of AI in pesticide products. Maximum and minimum rates come from product labels.

**the number of reports is the number of unique 1080 forms received with indicated AI. 1080s with multiple AIs are counted for each AI. Acres for multiple AI mixes are separately counted for each AI. % of acres treated is AI acre total divided by planted acres. Only previous year's planted acres are available.

***Up to four target pests are recorded and multiple AI applications are common. No mechanism in the 1080 forms presently exists to link specific AIs to specific target pests. For this reason, all AI/pest counts do not necessarily reflect intended efficacy.

Arizona Pesticide Use Reporting

The state of Arizona mandates that records must be kept on all pesticide applications. Submission to the Arizona Department of Agriculture (ADA) of these pesticide use reports (form 1080) is mandated for all commercially applied pesticides, pesticides included on the Department of Environmental Quality Groundwater Protection List (GWPL) and section 18 pesticides.

Commercial applicators licensed through the state must submit Arizona Department of Agriculture Form 1080 Pesticide Use Reports for all applications. The use of commercial applicators varies across crops. Aerial application is always performed by commercial applicators.

The GWPL is a list of active ingredients determined by the Department of Environmental Quality to potentially threaten Arizona groundwater resources. Enforcement of this list is difficult. Strictly speaking, only specific types of soil application of GWPL active ingredients must be reported. Inclusion on the GWPL should indicate a higher level of reporting but without further research no useful distinctions can be drawn.

Section 18 active ingredients should have 100% reporting. There were no section 18s active in Arizona for collards in the 1999 growing season.

Voluntary reporting does take place. Anecdotal evidence indicates some producers submit records for all applications.

Reported pesticide usage provides a solid lower bound of acres treated and a mean application rate of reported applications. Relative magnitude of reported acres is useful for rough comparison but could reflect a bias among commercial applicators or differing reporting rates as a result of inclusion on the GWPL. Finally, while the quality of data from the ADA 1080 forms has improved dramatically in recent years, there is still the possibility of errors.

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