NATIONAL PARK SERVICE
IPM Information Package

CRICKETS AND GRASSHOPPERS

Final Report

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Submitted To:

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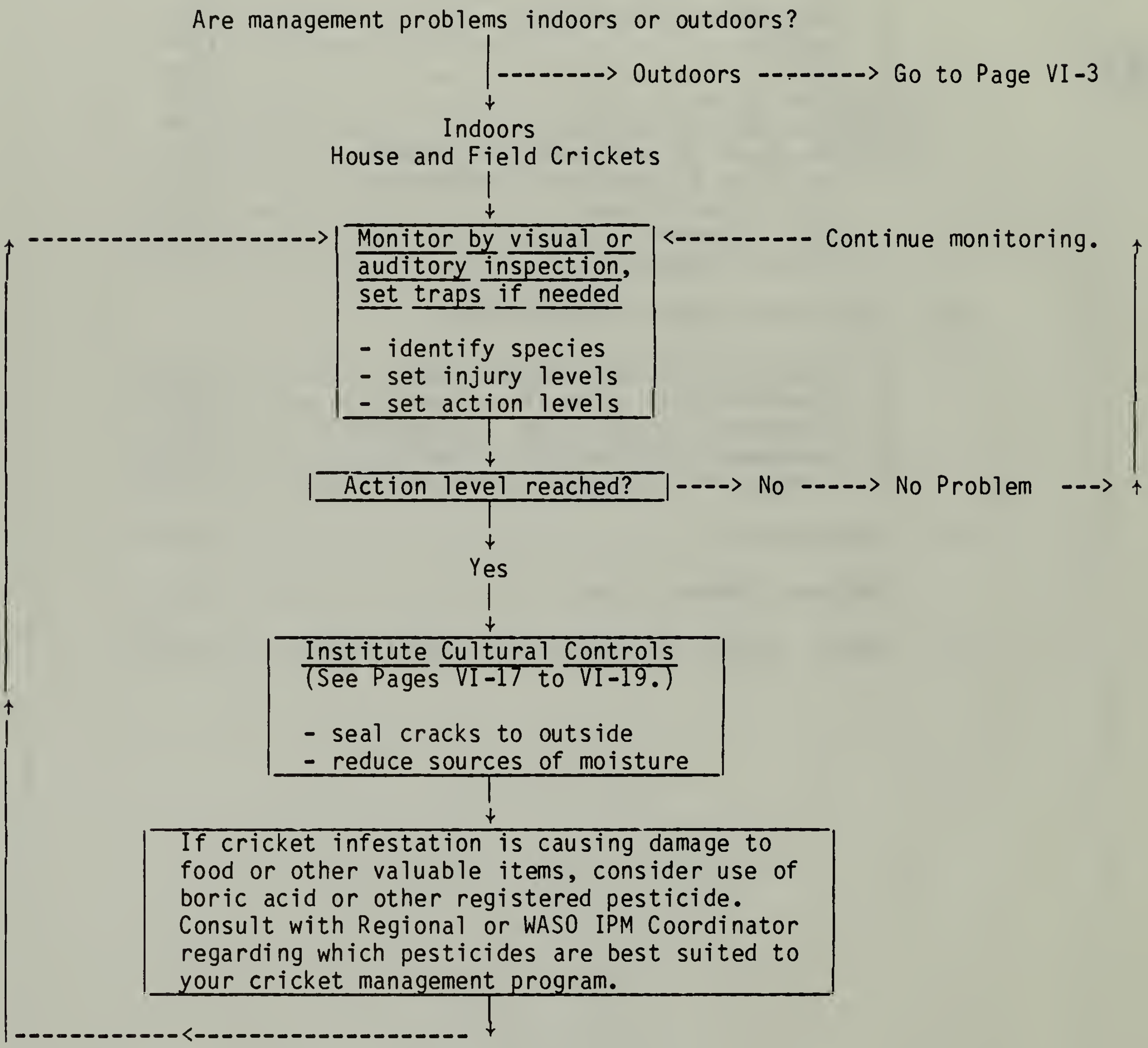
CONTENTS

I. CRICKET AND GRASSHOPPER IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



Are management problems indoors or outdoors?

Outdoors --------> Go to Page VI-3

Indoors
House and Field Crickets

Monitor by visual or auditory inspection, set traps if needed

- identify species
- set injury levels
- set action levels

Continue monitoring.

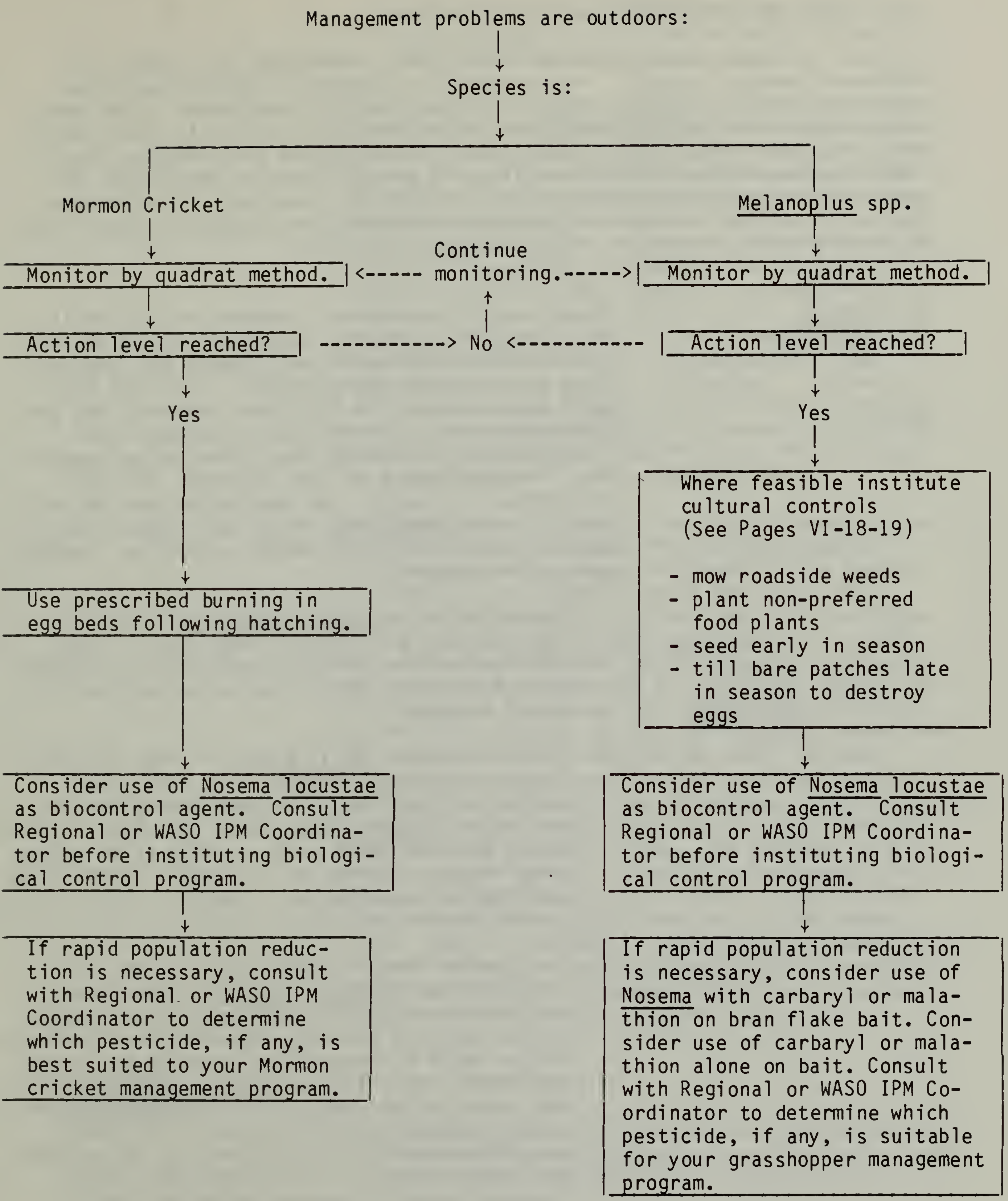
Action level reached? ----> No -----> No Problem

Yes

Institute Cultural Controls (See Pages VI-17 to VI-19.)

- seal cracks to outside
- reduce sources of moisture

If cricket infestation is causing damage to food or other valuable items, consider use of boric acid or other registered pesticide. Consult with Regional or WASO IPM Coordinator regarding which pesticides are best suited to your cricket management program.



Management problems are outdoors:

Species is:

Mormon Cricket

Monitor by quadrat method.

Action level reached?

Yes

Use prescribed burning in egg beds following hatching.

Consider use of <u>Nosema locustae</u> as biocontrol agent. Consult Regional or WASO IPM Coordinator before instituting biological control program.

If rapid population reduction is necessary, consult with Regional or WASO IPM Coordinator to determine which pesticide, if any, is best suited to your Mormon cricket management program.

Continue monitoring.

No

<u>Melanoplus</u> spp.

Monitor by quadrat method.

Action level reached?

Yes

Where feasible institute cultural controls (See Pages VI-18-19)

- mow roadside weeds
- plant non-preferred food plants
- seed early in season
- till bare patches late in season to destroy eggs

Consider use of <u>Nosema locustae</u> as biocontrol agent. Consult Regional or WASO IPM Coordinator before instituting biological control program.

If rapid population reduction is necessary, consider use of <u>Nosema</u> with carbaryl or malathion on bran flake bait. Consider use of carbaryl or malathion alone on bait. Consult with Regional or WASO IPM Coordinator to determine which pesticide, if any, is suitable for your grasshopper management program.

II. CRICKET AND GRASSHOPPER BIOLOGY AND ECOLOGY

Crickets and grasshoppers, members of the order Orthoptera, are common and widespread jumping insects. Many hundreds of species of crickets and grasshoppers occur in the United States. Although normally considered to be important components of natural ecosystems, a few species occasionally become serious pests, both indoors and in the field. Under certain environmental conditions huge swarms consisting of millions or even billions of individuals may appear, causing widespread destruction of crops and rangeland. This package describes the life histories and management of four groups that have been found to be of greatest concern in the National Park System.

1. Species Described:

A. House and Field Crickets - Members of the family Gryllidae, these are the common and widely distributed crickets with which most people are familiar. They have long slender antennae. The wings are well developed and lie flat over the back, but are bent down sharply at the sides. Male crickets chirp or sing by rubbing their front wings together. Females possess a long, straight, slender, cylindrical ovipositor, or egg-laying tube at the end of the abdomen. Both males and females have a pair of long cerci or filaments projecting from the end of the abdomen.

1. House Cricket - Acheta domesticus (L.) has a body about 5/8 - 7/8 inch long, and is light brownish yellow with dark markings on the head and thorax. The hind wings extend beyond the cerci at rest.

2. Field Crickets - Gryllus spp. - These are the large black field crickets with which most people are familiar. The males chirp both day and night. Gryllus spp. range from 5/8 - 1 inch long, and from solid black to pale straw color, with reddish or brownish coloration in the wings and legs of some species. The hind wings do not extend beyond the cerci at rest.

   The species known for many years as the black field cricket, Gryllus assimilis (Fabricius), has been shown to be a complex of five closely similar species that are separated most reliably on the basis of the calling songs of the males (Alexander 1957, 1962). Gryllus assimilis is now known as the Jamaican field cricket and the common name black field cricket is no longer used (Alexander 1957; Alexander and Walker 1962). Four other genera also are sometimes called field crickets (Alexander and Walker 1962).

Keys to southeastern species of Gryllus based on morphology are provided by Dakin and Hays (1970) and Nickle and Walker (1974).

B. Mormon Cricket - Anabrus simplex Haldeman is a shield-backed grasshopper of the family Tettigoniidae. Adults are large (1 - 2 3/8 inches) and dark shining brown to bluish black. These insects are flightless with the wings reduced to short stubs. The antennae are slender and as long as the body. The pronotum, or first segment of the thorax behind the head, is extended backwards as a shield that covers the rest of the thorax to the base of the abdomen. Males chirp by rubbing their stubby wings together. Female Mormon crickets have a flattened, upcurved ovipositor nearly as long as the body extending from the end of the abdomen. Two additional species, A. cerciata Caudell and A. longipes Caudell, are also referred to as Mormon crickets in some older literature. See Milne and Milne (1980) for photograph and further description.

C. Melanoplus spp. - This is a very large genus of the family Acrididae, or short-horned grasshoppers, that contains some of our most destructive species of range and crop land in North America. The damage is caused mainly by four species: the migratory grasshopper M. sanguinipes (Fabricius), the differential grasshopper M. differentialis (Thomas), the twostriped grasshopper M. bivittatus (Say), and the redlegged grasshopper M. femurrubrum (De Geer); another 8-10 species are also of economic importance. All of them can be recognized by their antennae which are much shorter than the length of the body, the short spine between the front legs, and the clear, colorless hind wings. They range in size from 3/4 to 1 1/2 inches long, and the females are larger than the males. They are yellowish to olive green or reddish-brown, with light stripes and/or dark red, brown, or black markings on the thorax, wings, legs, and abdomen. Males sing by rubbing their hind legs against the front wings. See Milne and Milne (1980) or Anonymous (1969) for illustrations and descriptions of several species.

2. Geographic Distribution:

A. House and Field Crickets -

1. House Cricket - This species was introduced to North America during the eighteenth century, and now is widely distributed in Canada and the United States.

2. Field Crickets - *Gryllus* spp. are widespread throughout North America, Central America, and northern South America. *Gryllus assimilis*, which is found only southern Florida in the U.S., might have been introduced there from the West Indies or a Central or South American Caribbean country where it also occurs (Alexander and Walker 1962). D. A. Nickle (personal communication) considers all species of *Gryllus* in the U.S. to be native. Some species in other genera have been introduced (Alexander and Walker 1962).

B. Mormon Cricket - Common from the Coast Range east to the northern and central Great Plains, and extending from Canada to Arizona. It also has been reported from Tennessee (Goodwin and Powders 1970).

C. *Melanoplus* spp. - Species of this genus occur throughout North America.

3. Habitat:

A. House and Field Crickets -

1. House Cricket - House crickets may be common in garbage dumps. Seeking warmth, they often enter houses as fall approaches. They are more frequently found indoors, where they may become established if food and moisture are available.

2. Field Crickets - These normally are found in open fields and along roadsides where they live in cracks in the soil and under litter. Different species have different habitat preferences. Like the house cricket, they may enter buildings in the fall seeking warmth, but do not become established there.

B. Mormon Cricket - This species occupies a wide variety of habitats, including sagebrush communities, mountain and desert shrub communities, and riparian communities (J.L. Kennedy, in litt.).

C. *Melanoplus* spp. - Most species are found in open grasslands, meadows, and cultivated fields.

4. Hosts:

A. House and Field Crickets - These crickets feed on juicy fruits and vegetables, flowers and developing seeds, and leaves, stems and roots of plants such as alfalfa and small grains. In houses, they may feed on wool, linen, fur, silk, nylon, rubber, and leather, as well as meat or meat products and dead insects. They also will eat paper or other items stained with grease or perspiration.

B. Mormon Cricket - Wakeland (1959) stated that Mormon crickets are omnivorous feeders, and that they eat almost any green vegetation. They are known to feed on more than 250 species of range plants and all cultivated crops they come in contact with (Wakeland and Parker 1952). However, Ueckert and Hansen (1970) report that forbs comprise 50% of the diet of Mormon cricket adults and late instar nymphs, while grasses, clubmoss, and grasslike plants (Carex and Juncus spp.) comprise 6%, 5%, and 2%, respectively. In addition, aphids and other small arthropods form 21% of the diet, and fungi 16%. Injured or dead Mormon crickets are readily eaten by healthy individuals (Wakeland 1959; J.L. Kennedy, in litt.).

C. Melanoplus spp. - Most species show a distinct preference for particular food plants, but, in general, the economically important species tend to be omnivorous or to prefer grasses over forbs (Hewitt and Onsager 1983). During outbreaks, however, they "...frequently consume every bit of green vegetation." (Comstock 1925).

5. Life Cycles:

A. House and Field Crickets -

1. House Cricket - The life cycle takes one year to complete in the field, but populations in houses may be active year round. Mated females deposit eggs singly in cracks and crevices in dark recesses. The number of eggs laid by a female varies directly with the temperature. Eggs hatch in 8-12 weeks. The nymphal stage lasts 30-33 weeks, with 9-11 molts. After mating, females wait up to 10 days before starting to lay eggs. Oviposition may continue for 5 weeks and females may live a further 19 days after egg-laying is finished.

2. Field Crickets - The life cycles of Gryllus spp. vary from species to species and with locality. Species in northern states normally have one generation per year, but field crickets in southern states may have as many as three. Mated females deposit eggs singly in the ground by inserting the ovipositor to a depth of 1/4 - 1 inch, preferrably in sandy

soil. Many eggs may be deposited in the same vicinity, with a total of 150-400 eggs laid by each female. Eggs are elongate-oval, slightly curved, and up to 1/8 inch long. They are light honey-yellow at first, turning cream colored as they develop. Egg-laying in species having one generation per year begins in August and continues into the fall. The eggs overwinter, but most adults and nymphs die as winter approaches. Hatching depends on the weather, but generally starts in April or May. In species having multiple generations per year, nymphs and adults may be active throughout the year. Nymphs pass through 8-10 instars before becoming adults: males normally pass through 8 instars, and females pass through 9. Complete nymphal development requires 80-90 days, with the males maturing before the females.

B. Mormon Cricket - There is one generation per year in most localities, but, at least at high elevations in the Big Horn Mountains, eggs may not hatch in the first year and thus the life cycle may require 2 years to complete (Cowan and Shipman 1940). Eggs are deposited singly in the soil just below the surface, but many eggs may be deposited in one place without the female completely withdrawing her ovipositor (Wakeland 1959). Deep, well-drained soils are preferred for egg-laying (J.L. Kennedy, in litt.). Each female deposits about 150 eggs. Egg-laying occurs in the summer and development begins soon afterward. Eggs are dark brown at first, becoming dull gray as the embryos mature. Hatching does not occur until the ground warms in the following spring. Hatching has been recorded as early as mid-January and as late as August 1, but normally extends from mid-April through May (J.L. Kennedy, in litt.). First instar nymphs range up to 1/4 inch in length. They are light tan initially, becoming black with white markings on the pronotum. Nymphs pass through 7 instars over a period of approximately 60 days. As they grow they may assume various colors, showing shades of green, red, and yellow.

C. Melanoplus spp. - Grasshoppers typically have a single generation per year, but some species, such as the migratory grasshopper, may have two or three generations per year in the southern parts of their ranges. Those having a single generation per year lay eggs in the fall by depositing them in clusters in the soil. The eggs are elongate oval, about 1/8 inch long, and are cream-colored. A glue-like secretion holds the eggs together and also binds soil particles to the eggs, producing a small case called an egg pod. The number of eggs per pod varies depending on the species: pods of the migratory grasshopper contain 15-25 eggs, those of the redlegged 25-30, and pods of the differential and twostriped 50-150. Each female lays as many as 800 eggs, depositing them in 5-40 pods. The eggs overwinter and hatch the following spring, usually beginning in late April in the southern states and in late May in the northern states. Hatching takes place over a period of several weeks. Nymphs pass through 5 instars, each instar lasting 7-10 days. Adults become sexually mature in 10 to 14 days after the last molt. Egg-laying occurs approximately 2 weeks after mating. Adults may live another 4-6 weeks after reproducing.

6. Seasonal Abundance:

A. House and Field Crickets -

1. House Cricket - Once established indoors, house crickets may be seen throughout the year.

2. Field Crickets - Gryllus spp. populations in the north decrease gradually from a peak following the spring hatching of eggs, but their presence becomes more noticeable as nymphs become larger later in the season and as adults appear and males begin calling. In populations having more than one generation per year, densities are greatest in late summer and fall.

B. Mormon Crickets - Populations are greatest in the spring immediately after egg hatch; however, the larger nymphs and adults increase steadily in numbers through spring and summer, reaching peaks from June through August.

C. Melanoplus spp. - Densities of larger grasshopper individuals increase steadily throughout the year from egg hatch in the spring to peak adult population density in August and September.

7. Responses to Environmental Factors:

A. House and Field Crickets -

1. House Cricket - These crickets are nocturnal and are attracted to lights and warmth. During the day they hide in dark cracks and crevices,

or under litter out of the light. Growth and development are faster at higher temperatures. Ghouri and McFarlane (1958) report an average of 728 eggs laid at 82°F and 1060 at 89°F. The rate of chirping is directly related to the ambient temperature. Highly favorable conditions in successive years may give rise to exceptionally large populations that eventually reach outbreak proportions. When this occurs, huge swarms of crickets begin migrating away from their breeding grounds toward new sources of food.

2. Field Crickets - As with house crickets, field crickets are nocturnal and are attracted to lights at night (Hutchins and Langston 1953; Howell and Hensley 1955). Field crickets grow faster and require fewer molts to mature at higher temperatures than at lower temperatures. The rate of chirping is directly related to ambient temperature. Outbreaks of field crickets usually occur after a rainfall which ends a period of drought (Hutchins and Langston 1953).

B. Mormon Crickets - Temperature affects hatching, growth, and vigor. In cold weather these insects seek protection in soil cracks, under rocks or in debris. Migration of nymphs during outbreaks takes place on days that are clear or partly cloudy, with air temperatures between 65-90°F, soil temperatures between 75-125°F, and winds less than about 20-25 mph.

Crickets roost in brush at night, beginning at dusk when the temperature drops below about 65°F. They leave their roosts in the morning between 7:30 and 8:00 a.m. when the temperature rises and begin feeding. About 10:30 to 11:00 a.m. the crickets begin migrating and during migration very little feeding takes place, unless a good quality food source is encountered (i.e., a bran bait). About 3:30 to 4:00 p.m. the crickets stop migrating and begin to feed, and continue to feed until dusk (BLM report quoted by J.L. Kennedy, in litt.).

C. Melanoplus spp. - Temperature affects grasshopper growth and life history in every stage of development. Timing of egg hatching depends on accumulated degree-days since laying, as do the rates of development of the nymphal instars (Gage et al. 1976). Outbreaks occur after several successive years of highly favorable conditions in which the weather is warm and not too wet in the growing season, and high quality food is abundant. The number of grasshoppers doubles from year to year at first, then triples or quadruples, resulting in outbreaks every 8-10 years (Pfadt 1978; J. Onsager, personal communication).

8. Impact of Crickets and Grasshoppers:

8.1 Direct Impact:

A. House and Field Crickets -

1. House Cricket - These crickets may cause damage by feeding on household items, such as silk, wool, or other fabrics, food left exposed, or paper, leather, rubber, or other goods.

2. Field Crickets - Indoors, field crickets have much the same impact as the house cricket. Outdoors, field crickets may damage garden plants or field crops by feeding on flowers and developing seeds. They frequently cut off the seeds of grain crops and let them fall to the ground uneaten. The entire plant may be destroyed in a heavy infestation, with leaves, stems, fruits, roots, or tubers eaten.

B. Mormon Crickets - Injury is caused by feeding on leaves and reproductive tissues of plants, reducing yield and reproductive potential. Preferred food plants may be completely devoured even under normal circumstances, and in an outbreak young plants of many species may be completely devoured, older plants defoliated, and the twigs of bushes and shrubs may be girdled (Wakeland 1959).

C. Melanoplus spp. - See 8.1.B. Even in non-outbreak years, grasshoppers destroy in excess of 20% of all available range vegetation (Hewitt and Onsager 1983).

8.2 Indirect Impact:

A. House and Field Crickets - These insects primarily are nuisance pests indoors. The incessant chirping of the males at night is particularly annoying to some people. In an outbreak, huge swarms may be attracted to window lights, street lamps, or other outdoor lighting. Streets may become slippery with crushed crickets. Food may be contaminated by crickets walking over or defecating on it. During outbreaks, cats may feed exclusively on crickets and become emaciated and subject to fits (Ebeling 1975). In many places in the Southeast, crickets are reared for fish bait to be sold locally or shipped to bait stores in other parts of the country. This also serves to distribute species outside their normal geographic range (Alexander and Walker 1962).

B. Mormon Cricket - Swarms of Mormon crickets crossing highways may make driving hazardous as roads become slippery with crushed crickets. Crickets may contaminate water supplies when they fall into wells or other water systems and decompose. Overgrazing may lead to increased erosion by wind and water.

Mormon cricket feeding may or may not result in competition with livestock for forage, depending on local circumstances. Cowan and Shipman (1947) concluded that such competition may occur with serious results in Nevada. However, in the vicinity of Dinosaur National Monument, heavy utilization of death camas may actually have a desirable impact on livestock growers (J.L. Kennedy, in litt.).

C. Melanoplus spp. - Overgrazing may lead to increased erosion. Grasshoppers can transmit plant diseases such as potato spindle tuber, turnip yellow mosaic, tobacco mosaic, and tobacco ringspot, and some species are vectors of parasites of birds such as poultry tapeworm.

9. Natural Enemies:

A. House and Field Crickets -

1. House Cricket - Predators include spiders, ground beetles, the American cockroach, and the conenose bug Rasahus thoracius Stal.

2. Field Crickets - Many organisms parasitize field crickets, including species of wasps, flies, nematodes, gordian worms, mites, and protozoans. Of these, Severin (1926) found that the parasitic wasp Ceratoteleia marlatti Ashmead destroys 20-50% of field cricket eggs each year in South Dakota, and the protozoan Gregarina (sp.?) reduces the vitality of infected crickets, shortens their life span, and limits production of females. Field crickets have been shown to be susceptible to infection by Nosema locustae Canning, a microsporidian (Henry and Oma 1981). Predators include: several species of spiders; a digger wasp, Chlorion cyaneum Dahlborn; and several species of birds. Spiders and birds were found to have significant impact on nymph and adult population densities (Severin 1926). Ebeling (1975)

reports that during outbreaks cats may feed on crickets to the exclusion of all other food.

B. Mormon Cricket - Mormon crickets have many parasites and predators. Parasites include wasps, gordian worms, nematodes, and Nosema locustae, an extremely promising biological control agent. Predators include sphecid wasps, ground beetles, robber flies, spiders, many species of rodents and birds, as well as, coyotes, skunks, and badgers. Groups of kestrels can be used as an aid in locating bands (J.L. Kennedy, in litt.). Wakeland (1959), who was unaware of N. locustae, concluded that while these parasites and predators serve to keep Mormon cricket populations in check under normal conditions, in outbreaks they serve little practical use. Historically, however, gulls are credited with stemming an outbreak that threatened the survival of pioneers in the vicinity of Salt Lake City in 1848.

C. Melanoplus spp. - Grasshoppers have many natural enemies. Eggs are parasitized by wasps of the genus Scelio, while flesh flies, tachinid flies, and tangleveined flies parasitize nymphs and adults. When humidity is high a fungal pathogen, Entomophthora grylli, can cause extensive epizootics. An extremely promising biocontrol agent is Nosema locustae Cannings, a microsporidian parasite fatal to grasshoppers and Mormon crickets.

Predators include many species of spiders, robber flies, predatory wasps, larvae of bee flies, blister beetles, and ground beetles. Rodents and other mammals feed on the eggs, nymphs, and adults, and birds may eat a large number of grasshopper nymphs and adults.

Although these organisms help to keep populations in check under normal conditions, and may even help end an outbreak, only the microsporidian and fungal parasites are considered to have much immediate potential for biological control.

1. <u>Population Monitoring Techniques:</u>

A. House and Field Crickets -

1. House Cricket - The presence of house crickets usually is first noticed by hearing the males singing at night. Thus, a monitoring program consists initially of locating singing crickets. Because of the nocturnal habits of crickets, this may be done most effectively by turning on the lights in a darkened room where crickets have been heard singing, or by searching in the dark with a flashlight. If the crickets can not be located in this manner, it will be necessary to move boxes or furniture, or look behind appliances. Because house crickets seek shelter in cracks and crevices behind baseboards, in loose fitting masonry, or in cabinets, and prefer warm areas near stoves, fireplaces, and furnaces it is most productive to search these areas first. Look for signs of cricket feeding damage in fabrics, food, or other items. Holes made by crickets can be distinguished from feeding damage caused by case-making or webbing-making moths or beetle larvae because the holes are large and there is never any silk associated with the damaged areas. A floor plan map of the infested room(s) may be needed to record data on cricket harborages and population levels.

Relative population size can be estimated by determining the number of crickets heard singing, or by visual counts. Females are attracted to singing males: therefore, for each singing male there may be assumed to be at least 2 and probably more females present (Ebeling 1975).

Alternatively, a simple pit-fall trap may be made from a 1-quart or larger wide-mouth jar. A piece of juicy fruit or other suitable food (see Section II.4.A, Page VI-6) is put in the bottom of the jar, and the trap is positioned upright in a corner or near a known or suspected cricket harborage. It may be necessary to apply a thin film of petroleum jelly around the inner neck of the jar to prevent the crickets from escaping. Pieces of wood, cardboard, or other material are attached to serve as ramps allowing the crickets to enter. The location of each trap is recorded on the appropriate room floor plan. Each trap is inspected and the number of crickets captured is recorded daily, and the trap emptied.

2. Field Crickets - These insects rarely, if ever, become established indoors, and are usually found close to their point of entry into a building. Auditory and visual monitoring as described for the house cricket are adequate in most situations. In cellars or infrequently used structures traps may be used if necessary. If crickets are entering a building, monitor the exterior by both auditory and visual methods. Crickets may be located during the day by disturbing their hiding places in grass or bushes, in wood piles, or under leaves or other items providing a dark protected hiding place on or near the ground. Pit-fall traps as described above for the house cricket survey may be buried in the ground up to the top of the jar, or provided with ramps for access as described above. The traps should be covered with a board or other material, leaving space for the crickets to crawl under. Traps should be inspected, the number of crickets recorded and the traps emptied daily.

B. Mormon Crickets - No completely satisfactory method is available to sample Mormon cricket populations. APHIS recommends the same quadrat method that is used for grasshoppers (see 1.C). NPS personnel in Dinosaur National Monument (J.L. Kennedy, in litt.) use circular hoops of 1 yd^2 or 0.1 yd^2 depending on the size and number of crickets to estimate population density. Twenty or more samples are averaged for each band. This technique is useful except at very low densities, in which case populations are recorded as 0-1/yd^2. Plot the location of all samples on a map and record the density, date, time of day, temperature, and the type, density and height of vegetation on a survey form, such as the one on Page 25. Record the location and extent of egg beds. These may be located by observing oviposition, and confirmed by taking soil samples and carefully sifting for eggs.

C. Melanoplus spp. - Grasshopper populations are monitored using a quadrat technique. A monitor walks in a straight line and counts the number of grasshoppers leaving a square foot of area (or 0.33 m^2 area) selected by the monitor well ahead of his approach. Eighteen counts are made 15-20 paces apart along the line of march. The total number from all 18 square foot (or 0.33 m^2) samples is computed and divided by two to determine the density per square yard (or meter) (Anonymous 1969; Anonymous 1981). Data are recorded on a survey form such as the one on Page 25. Also record the date, time of day, temperature, and the type, density, and height of vegetation present, and the economically important species encountered. Take notes on the relative proportions of different nymphal stages and adults, mating and oviposition activity, and presence of predators and parasites. Reliable maps are used to plot the location of each area surveyed, and the density of grasshoppers at each location recorded.

The timing of a survey will depend upon the management needs of a particular Park, and the history of grasshopper problems in the area. A survey of adult populations in August or September will help determine if there is a potential for damaging grasshopper densities the following season. Beginning in the spring, nymphal surveys in high risk areas identified the previous fall will allow park personnel to monitor populations that show the greatest potential for problems.

2. Threshold/Action Population Levels:

A. House and Field Crickets - Threshold and action levels for crickets suggested here are arbitrary, as there are no published guidelines that deal with nuisance crickets. If crickets are indoors and damage to food, fabric, or other items is discovered, action should be taken immediately. If there are no visible signs of damage, action levels will need to be determined by park personnel by correlating cricket densities with staff complaints.

If field crickets threaten gardens or other valuable plants outside, action levels must be determined by weighing the desirability of management measures against the aesthetic or other value of the threatened plants, taking into account such factors as the season and the stage of development of the plants. In general, if cricket densities of greater than 5 large individuals per pit-fall trap per night are encountered for 3 consecutive nights, and the plants are at a susceptible stage such as the start of fruit or seed set, management measures may be required.

B. Mormon Crickets - APHIS has set the threshold for Mormon crickets at 8 per square yard, but that figure is flexible and depends on the ability of the affected land to withstand damage (C. Bare, personal communication). Because they are natural components of park ecosystems, one criterion for use in the NPS might be to prevent undue economic impacts on adjacent land (J.L. Kennedy, in litt.).

C. Melanoplus spp. - The APHIS threshold for grasshoppers is 8 per square yard. However, recent research indicates that the APHIS action levels can be considerably refined by taking into account the value of the forage, the average amount eaten by the grasshoppers, the cost of management, and other factors (Onsager 1984). See also 2.B.

3. Management Alternatives-Nonchemical:

A. House and Field Crickets - Most cricket management problems can be solved through the use of cultural management methods. These are detailed in Carr (1982), and may be divided into exterior and interior controls:

1. Exterior - Reduce cricket harborage by keeping lawns mowed and gardens close to buildings weeded. Remove woodpiles stacked against buildings to at least 1 ft away and keep the space between clear of weeds and debris. A layer of ashes applied in a band around the base of the wood pile will help decrease its attractiveness to crickets. Keep shrubs and other harborages away from building entrances. Fill the space between the building foundation and the soil with gravel. Garbage cans should be raised off the ground on pallets or other supports and the space beneath them kept free of litter. If large populations of crickets are developing in garbage dumps or trash heaps, the dumps should be removed or buried. Outdoor lighting should be eliminated or reduced where feasible, or yellow "bug" lights used in place of white incandescent or fluorescent lights. Buildings should be inspected for openings near ground level that might allow crickets to enter. Weather strip doors and windows, especially window wells. Screens, and vents should be repaired if they are not tight fitting. Holes should be caulked or plastered. Corrective measures should be taken to repair clogged drainpipes or other problems which cause moisture buildup near the foundation.

2. Interior - Repair leaking pipes or other sources of moisture to deny the crickets water. Repair loose-fitting baseboards, seal cracks, and tighten the fit of cabinet doors. Doors of closets which have spaces at the bottom should be made tight fitting to deny crickets access to stored items. Clean up cellars and basements; remove trash, sweep and vacuum up debris, and maintain a high level of cleanliness. If crickets have damaged food, discard it, and in the future store food in sturdy containers with tightfitting lids.

No biocontrol agents are recommended for these insects. However, the microsporidian *Nosema locustae* has been found to infect field crickets, but not house crickets (Henry and Oma 1981).

B. Mormon Crickets - Prescribed fire in egg bed areas after hatching of Mormon crickets may be an effective cultural control in some localities, particularly since the egg bed areas are quite small and, once located, are easily definable (J.L. Kennedy, in litt.).

Nosema locustae has been found to cause death of Mormon crickets (Henry and Oma 1981), and is available in a commercial preparation from the following source:

Reuter Laboratories
14540 John Marshall Highway
Gainesville, Virginia 122065
Attn: Carter Marantette

(703) 754-4167

Consult with your Regional or WASO IPM Coordinator to determine the suitability of biological control measures for your Mormon cricket management program.

C. *Melanoplus* spp. - Various methods of cultural control may be used where appropriate in grasshopper management programs. Tilling the soil can bury eggs so deep that hatching will not occur, or it can bring the eggs to the surface where they are exposed to drying by the sun and wind, and to feeding by predators. Tillage can also make the earth unattractive for oviposition. Fall is the most effective time to attempt grasshopper management by tillage.

Early spring planting helps reduce the impact of grasshopper feeding later in the season, since the plants have had a longer time to mature.

Weedy field margins, roadsides and fence rows are favored grasshopper egg-laying sites. Plant perennial grasses, such as crested wheatgrass, in these areas.

Some varieties of sorghum, such as sorgo and kafir, are resistant to grasshopper attack after reaching over 8 inches in height. In localities where grasshopper outbreaks are severe, substitute these crops for more susceptible small grains.

More details on methods of cultural control are available in Anonymous (1977) and Pfadt (1978).

Nosema locustae is registered for use against grasshoppers. See 3.B for information on obtaining *N. locustae*. It should be applied when grasshoppers are young (i.e., 3rd to 4th instars), because it takes two or more weeks to take effect. Since grasshopper outbreaks tend to occur in 8 to 10 year cycles, a single, properly timed application of *N. locustae* could give up to 10 years of control (J. A. Onsager, personal communication). Research on the efficacy of *N. locustae* as a biocontrol agent for grasshoppers is carried out at the Rangeland Insects Laboratory of the USDA in Bozeman, Montana.

4. Management Alternatives-Chemical:

A. House and Field Crickets - Many chemicals are registered for use against crickets in buildings. The timing of chemical use is very important; chemicals are not needed until late summer, if at all. Several boric acid products, Dri-Die® (silca gel and fluosilicate), and Drione® (silica gel and pyrethrins) are registered for cricket management and may be used around stoves, furnaces, or other large appliances that are difficult to move, and may be blown into cracks or behind baseboards. Allethrin, chlorpyrifos, diazinon, pyrethrins, and resmethrin are also registered for application as sprays or dusts around baseboards or in cracks.

B. Mormon Cricket - Consult with Regional or WASO IPM Coordinator concerning chemical control of this species.

C. *Melanoplus* spp. - *Nosema locustae* may be used in combination with carbaryl on a bran flake bait

(Onsager et. al. 1980). Carbaryl and malathion are registered for use separately on bran bait. These should be applied when hatching of the target species is completed and before egg-laying begins.

Consult with your Regional or WASO IPM Coordinator to determine which chemical, if any, is suited to your cricket and grasshopper management program.

5. Summary of Management Recommendations: Keep records of all important infestations of any pests, and the management measures taken and their effectiveness.

A. House and Field Crickets -

a. Begin monitoring when crickets are seen or when males are heard singing. Use auditory and visual techniques to locate harborages; set up pitfall traps if necessary. If populations are large (i.e., 5 or more males singing per night per room, or 2 or more crickets captured per trap per night), look for signs of cricket feeding damage to food, fabrics, or other items.

b. If action levels are reached, undertake cultural controls to eliminate food and moisture sources indoors, and reduce harborages indoors and out. Seal cracks, fix loosefitting doors and windows, and reduce outside lighing or use "bug" lights.

c. If the situation warrants, consider use of a registered pesticide.

B. Mormon Cricket -

a. Monitor Mormon crickets beginning in the spring. Record densities, locations, stages of growth, feeding habits, mating and oviposition activity, predators and parasites, and presence or absence of endangered species in cricket habitats.

b. If Mormon crickets must be controlled, consider use of prescribed burns in egg bed areas after eggs hatch. Consider application of Nosema.

C. Melanoplus spp. -

a. Monitor grasshopper populations in areas where outbreaks have occurred in the past or where high densities have been noticed by park personnel. Estimate densities using quadrat method.

b. If grasshoppers have reached damaging levels in recent years, apply cultural control methods: mow weedy roadsides to reduce feeding grounds; plant forbs or other non-preferred plants in bare or eroded areas; till open areas where grasshopper eggs are buried in the soil. Consider use of *Nosema locustae* as potential long-term biocontrol agent.

c. If grasshoppers threaten endangered species, historically important areas, or an outbreak that would spill over onto non-park property consider application of *Nosema* in combination with carbaryl or malathion, or consider use of carbaryl or malathion alone on treated bran bait for immediate control.

Consult with your Regional or WASO IPM Coordinator to determine which pesticide, if any, is best suited to your cricket and grasshopper management program.

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V. PERSONAL COMMUNICATIONS

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VI. SAMPLE CRICKET AND GRASSHOPPER SURVEY FORM
(Adapted from Plant Protection and Quarantine Form 370.)

PARK

DATE

MONITOR(S)

Sq. ↓	STOP NUMBER														
	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15
1															
2															
3															
4															
5															
6															
7															
8															
9															
10															
11															
12															
13															
14															
15															
16															
17															
18															
TOT															
#/m^2															

Total grasshoppers from 18 squares divided by 2 = #/m^2 (or #/ft^2).

(OVER)

Sample Cricket and Grasshopper Survey Form
(back page)

Stop No.	Location (be specific)	Notes* (species, food plants, weather, temperature, time, habitat, etc.)
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		
11		
12		
13		
14		
15		

* Use additional sheets for notes where necessary.

NATIONAL PARK SERVICE
IPM Information Package

EXOTIC WEEDS I: KUDZU, SALTCEDAR, AND BRAZILIAN PEPPER

Final Report

16 November 1984

Submitted To:

William E. Currie
U.S. Environmental Protection Agency
Arlington, Virginia 22202

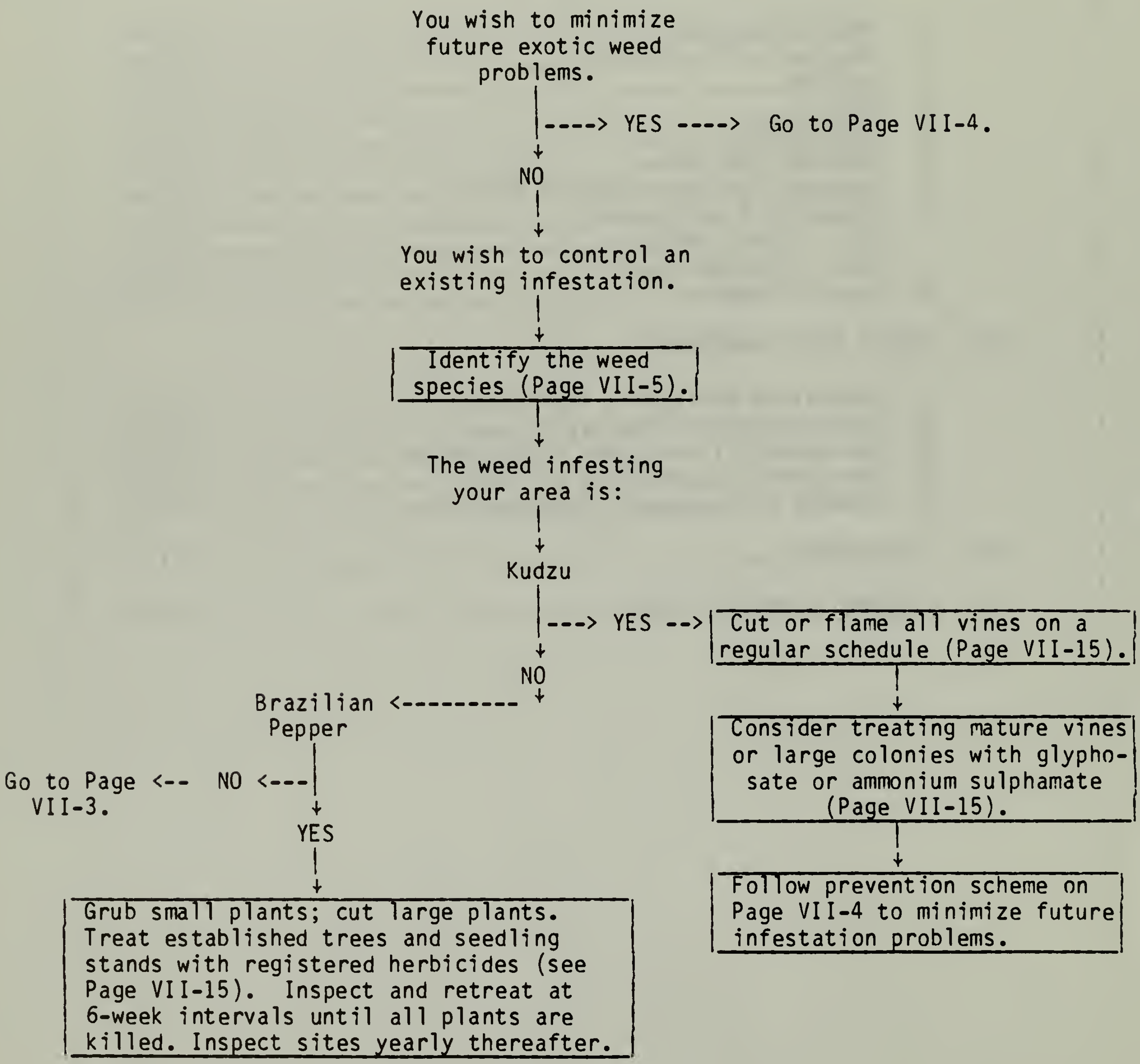
Submitted By:

Dynamac Corporation
11140 Rockville Pike
Rockville, Maryland 20852

CONTENTS

I. EXOTIC WEED IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



You wish to minimize future exotic weed problems.

YES Go to Page VII-4.

NO

You wish to control an existing infestation.

Identify the weed species (Page VII-5).

The weed infesting your area is:

Kudzu

YES

Cut or flame all vines on a regular schedule (Page VII-15).

Consider treating mature vines or large colonies with glyphosate or ammonium sulphamate (Page VII-15).

Follow prevention scheme on Page VII-4 to minimize future infestation problems.

NO

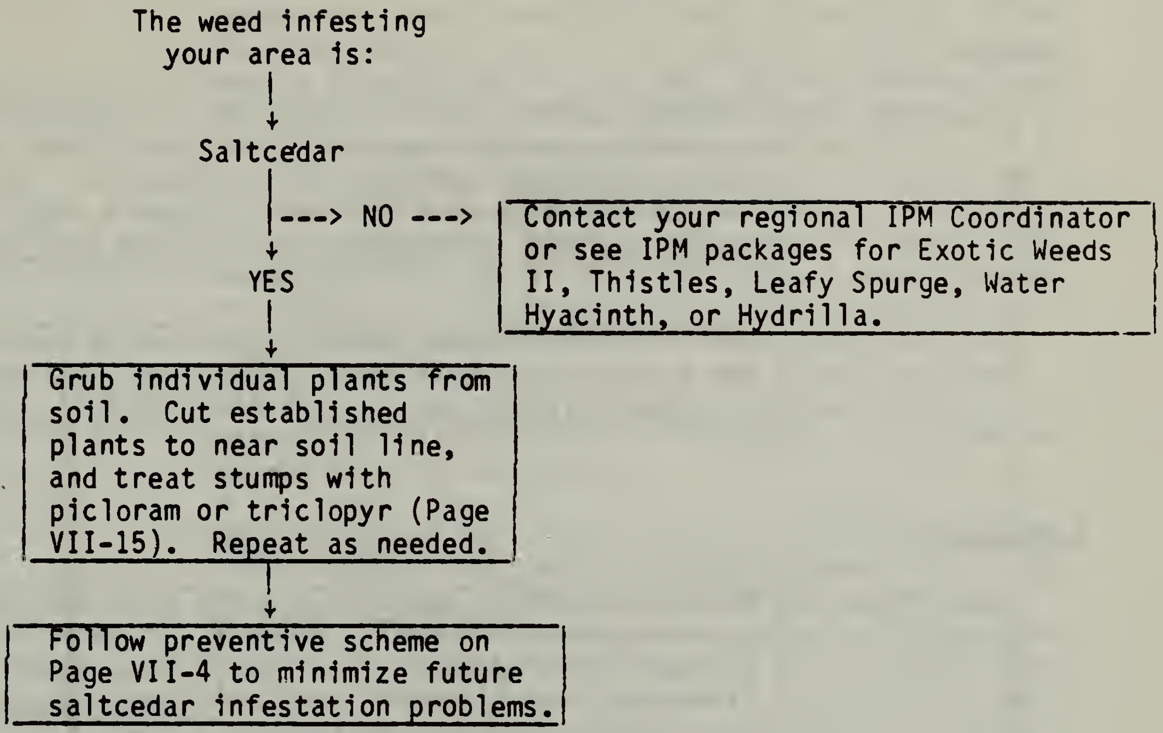
Brazilian Pepper

NO Go to Page VII-3.

YES

Grub small plants; cut large plants. Treat established trees and seedling stands with registered herbicides (see Page VII-15). Inspect and retreat at 6-week intervals until all plants are killed. Inspect sites yearly thereafter.

Continued from Page VII-2.



The weed infesting your area is:

Saltcedar

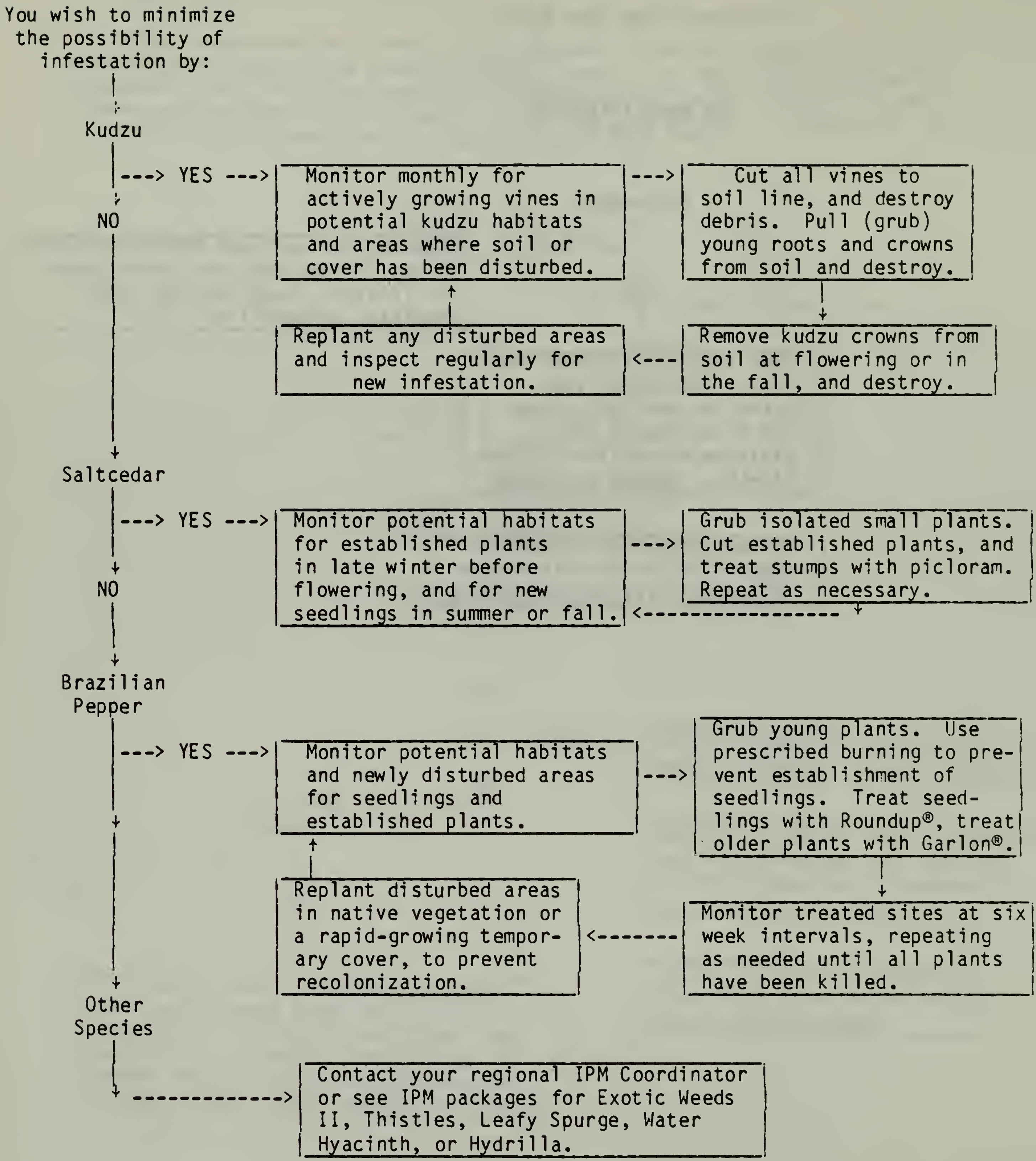
NO

Contact your regional IPM Coordinator or see IPM packages for Exotic Weeds II, Thistles, Leafy Spurge, Water Hyacinth, or Hydrilla.

YES

Grub individual plants from soil. Cut established plants to near soil line, and treat stumps with picloram or triclopyr (Page VII-15). Repeat as needed.

Follow preventive scheme on Page VII-4 to minimize future saltcedar infestation problems.



You wish to minimize the possibility of infestation by:

Kudzu

YES

Monitor monthly for actively growing vines in potential kudzu habitats and areas where soil or cover has been disturbed.

Cut all vines to soil line, and destroy debris. Pull (grub) young roots and crowns from soil and destroy.

Remove kudzu crowns from soil at flowering or in the fall, and destroy.

Replant any disturbed areas and inspect regularly for new infestation.

NO

Saltcedar

YES

Monitor potential habitats for established plants in late winter before flowering, and for new seedlings in summer or fall.

Grub isolated small plants. Cut established plants, and treat stumps with picloram. Repeat as necessary.

NO

Brazilian Pepper

YES

Monitor potential habitats and newly disturbed areas for seedlings and established plants.

Grub young plants. Use prescribed burning to prevent establishment of seedlings. Treat seedlings with Roundup®, treat older plants with Garlon®.

Monitor treated sites at six week intervals, repeating as needed until all plants have been killed.

Replant disturbed areas in native vegetation or a rapid-growing temporary cover, to prevent recolonization.

Other Species

Contact your regional IPM Coordinator or see IPM packages for Exotic Weeds II, Thistles, Leafy Spurge, Water Hyacinth, or Hydrilla.

II. EXOTIC WEED BIOLOGY AND ECOLOGY

1. Species Described:

A. Kudzu - Pueraria lobata (Willd.) Ohwi is a legume of the subfamily Fabaceae. It is a trailing or climbing semiwoody perennial vine reaching 32-100 ft in length. Young vines are covered with soft, fine hairs. First-year vines may reach 1/2" in diameter; old vines may reach a diameter of 4". As many as 30 vines may radiate from a single crown. Vines can grow up to 60 ft in a single growing season (and reportedly up to 1 ft per day). Vines may climb vertically as high as 50 ft, completely covering trees, buildings, or other supporting objects. During the growing season, vines are densely covered by foliage. Leaves are alternate and compound, with three broad leaflets up to 4" across, each leaflet entire or deeply 2-3 lobed and with hairy margins. Foliage drops after the first fall frost. The roots of kudzu are fleshy; the taproot may reach over 6 ft in length, 7" in diameter, and may weigh up to 400 lb.

Kudzu plants do not usually flower until their third year. Flowers are purple, fragrant, about 1/2" long, produced in long racemes, and resemble pea flowers in shape. They are produced in August and September. Flowers are followed by flattened, 2" long hairy pods which may contain 3-10 hard seeds. In the U.S., kudzu generally spreads by means of stolons (runners) and rhizomes. In addition, any vine contacting the soil will produce roots at nodes; these roots enlarge, forming new crowns. Vine cuttings and root divisions will also sprout. See Shurtleff and Aoyagi (1977) or other weed atlases for drawings of kudzu.

B. Saltcedar - Tamarix spp., especially T. ramosissima (Ledeb.), which is generally (but incorrectly) known as T. pentandra (Baum, 1978). Saltcedar is a deciduous shrub or small tree growing to 12-15 ft in height. Slender, long gray-green branches are spreading or upright, often forming dense thickets. Scalelike leaves are gray-green, alternately arranged, narrow, pointed, about 1/16" long, and overlap one another on the stems. The leaves often become encrusted with salt secretions. Branches take on a brown-purple color as they age. Bark is reddish-brown and smooth on young branches, becoming ridged and furrowed on older limbs. Large numbers of pink to white flowers, about 1/16" across, appear in a dense mass on 1/2-2" spikes at branch tips from March to September. Flowers are followed by greenish-yellow to pinkish-red capsules, 1/8-1/5" long, which split into 3-5 parts on maturity. Seeds are 1/25" long, with a tuft of fine hairs at one end. The number of seeds per capsule is not constant. See Baum (1978) or Parker (1972) for drawings of saltcedar.

C. Brazilian Pepper - Schinus terebrinthifolius (Raddi). This species is a member of the Anacardiaceae, and is closely related to poison ivy. It is a broad-topped, rapidly-growing tree reaching up to 40 ft tall, with a short trunk up to 40" thick. The trunk is usually hidden by a dense head of intertwining, contorted branches. Leaves are evergreen, pinnate, and have reddish midribs which may be winged. Each leaf bears 3-13 sessile, oblong or elliptical, finely toothed, glossy, resinous, aromatic 1-2" leaflets. These are dark green on the top, and lighter on the underside. Five-petaled, white, 1/8" flowers are borne in 6" sprays originating in leaf axils along the upper 32-43" of each stem. Male and female flowers are borne on separate trees. Flowering peaks in October in Florida. Blooms are followed by masses of round, single-seeded drupes, which change from green to bright red at maturity. The appearance of the fruit is responsible for the common names "Florida holly" and "Christmas berry." See Olmstead and Yates (1984) for photographs of Brazilian pepper.

2. Geographic Distribution:

A. Kudzu - A native of Asia, P. lobata was introduced into the U.S. at the Philadelphia Centennial Exposition in 1876. Beginning in 1933, farmers in the South were encouraged to plant kudzu to reduce soil erosion. By 1953, it had become such a weed problem that is was removed from the USDA's list of permissable cover plants. In 1970, the USDA began listing kudzu as a common weed in the South. Today, kudzu is common in Alabama, Georgia, Mississippi, Tennessee, the Carolinas, Kentucky, Virginia, Maryland, and west to Texas and Oklahoma (Edwards, 1982). The weed has also been reported in New York, Illinois, Iowa, Nebraska, and Washington (Shurtleff and Aoyagi, 1977).

B. Saltcedar - This plant is a native of Eurasia and Africa that was introduced into the U.S. as an ornamental shrub in the early 1800's, and has now spread throughout the intermoutain region of the western U.S. (Carman and Brotherson, 1982).

C. Brazilian Pepper - This weed was introduced from its native Brazil in 1898 by a USDA plant explorer (Morton, 1978). It was considered an ornamental shrub and was distributed by the USDA Plant Introduction Station in Miami, FL, to local plant enthusiasts. Since then, Brazilian pepper has spread over thousands of acres of land in South and Central Florida, the Florida Keys, the Hawaiian islands, southern Arizona, and southern California.

3. Habitat:

A. Kudzu - Kudzu grows well under a wide range of environmental conditions, although best growth is achieved where winters are mild (40-60° F), summer temperatures rise above 80°F, and rainfall is abundant (40" or more). Kudzu can grow in nearly any type of soil (e.g., acid soils, lime soils, lowlands with high water tables, and over heavy subsoil), and where winter soil temperatures remain above -25°F (which temperature kills roots). Forest edges or disturbed areas such as abandoned fields and roadsides are preferred habitats.

B. Saltcedar - This species occurs in moist rangeland and pastures, bottomlands, banks, and drainage washes of natural or artificial waterbodies, and other areas where seedlings may be exposed to extended periods of saturated soil conditions. Established plants have long roots with which they can tap deep water tables, and can survive in drought conditions. Saltcedar may survive in saline soils containing up to 15,000 ppm soluble salt.

C. Brazilian Pepper - This tree quickly colonizes disturbed areas. Seedlings are shade tolerant, and can tolerate moist or saturated soils. Established plants can tolerate extended drought or inundation (up to 6 months). Apparently, Brazilian pepper can tolerate Mediterranean, tropical, and desert climates.

4. Hosts:

Not applicable.

5. Life Cycles:

A. Kudzu - Kudzu is a perennial which rarely produces seeds in the U.S. (except on plants supported vertically on buildings, trees, or other supports [Shurtleff and Aoyagi, 1977]). Establishment of new plants is by rooting of vine nodes which come in contact with soil; these roots produce new crowns, and the connection to the mother crown dies within 1 year after rooting. Kudzu is deciduous; its leaves drop after the first frost, and new leaves are produced each spring.

B. Saltcedar - A deciduous perennial, this species annually produces seeds which are windborne to new locations. Flowers are pollinated by bees and other insects. Seedlings require extended periods of soil saturation for establishment. As seedlings become established, they develop long roots which are able to absorb water from deep below the soil surface.

C. Brazilian Pepper - This evergreen perennial produces large quantities of seeds each year. Seeds may be dispersed by birds or small mammals, or may germinate near the parent plant, producing dense spreading colonies.

6. Seasonal Abundance:

A. Kudzu - Kudzu foliage is present and vine growth occurs between early spring and the first frost. The vines are perennial, however, and are obvious year-round.

B. Saltcedar - Active growth occurs from early or mid-spring to fall, when leaves drop. Stems do not die back, forming perennial thickets which spread farther each year.

C. Brazilian Pepper - This species is evergreen, but becomes dormant during the winter months.

7. Responses to Environmental Factors:

A. Kudzu - Vines are intolerant of shade, and grow toward light. Large roots store water, allowing plants to survive in fairly dry climates (to 20" of rain per year). Growth is most rapid in acid to neutral soils (pH 4.5-7.0). P. lobata can reportedly grow in areas where winter temperatures reach -22°F; exposure to -25°F can kill roots.

B. Saltcedar - Seedlings require extended periods of saturated soil conditions for establishment; they cannot survive where water is scarce. Saltcedar can grow on soils with up to 15,000 ppm soluble salt. Established plants have among the highest known evapotranspiration rates of any desert phreatophytes (Carman and Brotherson, 1982), which may result in water depletion from the underlying soil.

C. Brazilian Pepper - Seedlings can tolerate low light levels, growing slowly until the overstory canopy is opened up. Trees can withstand extended drought, and up to 6 months of inundation. Large trees can withstand fires and high winds without suffering significant damage (Olmsted and Yates, 1984). Seedling survival is low on inundated ground.

8. Impact of Exotic Weeds:

8.1. Direct Impact:

A. Kudzu - Kudzu grows rapidly, choking out competing vegetation in sunny areas. Climbing vines may completely cover and shade out trees, and may cover and damage buildings, overhead wires, and other structures.

B. Saltcedar - Among the serious direct impacts of this species are the displacement of native range plants by its aggressive growth, the possibly serious depletion of ground water due to its rapid evapotranspiration rate, increased deposition of sediments in tamarisk-infested streams, and the blockage of streams and artificial water channels by dense clumps of saltcedar growth, which can promote flooding during periods of heavy rains.

C. Brazilian Pepper - Direct impacts include the displacement of native plants, not only because of this species' aggressive, rapid growth, but also because of allelopathic effects (toxic or inhibitory activity) of chemicals in vegetative plant parts and fruits.

Brazilian pepper is closely related to poison ivy, and can produce effects similar to that plant on humans and animals (Lloyd et al., 1977; Morton, 1978; Olmsted and Yates, 1984). Massive bird kills in Florida may have been caused by excessive feeding on Brazilian pepper berries.

8.2 Indirect Impact:

All of these species are aggressive growers, able to outcompete native plants which provide food and habitat for native animals. Replacement of the existing growth by these weeds results in a large-scale alteration of biotic communities and the potential elimination of certain species whose habitats are destroyed.

9. Natural Enemies:

A. Kudzu - In the U.S., kudzu vines may be attacked by a root knot nematode (*Meloidogyne* sp.), a "blackleg" fungus disease, a viral mosaic disease, and a rust fungus (Shurtleff and Aoyagi, 1977). These pests produce only minor injury, and are not known to kill kudzu plants.

B. Saltcedar - Watts et al. (1977) found only a few native insects that fed on saltcedar in New Mexico. These did little harm to the plants except under exceptional circumstances. Bugs, aphids, grasshoppers, beetles, and spider mites were among the organisms found. Watts et al. also reported two introduced insects, the leafhopper *Opsius stactogalus* and the scale *Chionaspis etrusca*, were found regularly on saltcedar. The leafhopper sometimes caused substantial damage. Baum (1978) compiled a list of insects and fungi which attack various species of *Tamarix* in Europe, Africa, and Asia, but found no records of enemies of *T. ramosissima*.

C. Brazilian Pepper - Goats can graze on foliage of this species without suffering ill effects (Morton, 1978). A witches' broom disease fungus, *Sphaeropsis tumefaciens* Hedges, attacks Brazilian pepper, but is also a pest of *Ilex opaca*, *Citrus* spp., and numerous ornamentals. The red-banded thrips (*Selenothrips rubrocinctus* Giard.) occasionally kills plants, but is also a pest of mango and cashew plantings.

III. EXOTIC WEED MANAGEMENT

1. Population Monitoring Techniques:

A. Kudzu - Regular monitoring of both developed and natural areas is required to determine the presence and extent of kudzu incursions. Since this species is a rapid grower and an aggressive competitor, these inspections should be conducted frequently (at least monthly) during the growing season.

In addition to inspecting areas for actively growing kudzu, monitors should also inspect disturbed areas, which can be rapidly colonized by the weed. All records of sightings of kudzu and of disturbed sites should be recorded, maintained, and updated at each inspection.

B. Saltcedar - Inspection of both developed and natural areas is necessary to determine the presence and extent of saltcedar incursions. One inspection should be made early in the growing season (before or at flowering), to identify mature plants and initiate control before seed can be set and distributed. Additional inspections should be made later in the growing season to identify seedlings developing from seed set in the current year. All records should be maintained and updated at each inspection.

C. Brazilian Pepper - Inspection of all likely habitats is required to determine the presence and extent of Brazilian Pepper incursions. At least one inspection per year should be made for the presence of established plants. Frequent inspections (i.e., at least monthly) should be made for the presence of disturbances in the normal plant cover (e.g., due to storms, alterations of water levels, fires, and human activities), since such sites can be rapidly colonized by this weed. All records should be maintained and updated at each inspection.

2. Threshold/Action Population Levels:

A. Kudzu - Since this weed is an adaptable, aggressive competitor which can rapidly overgrow native vegetation, the presence of any kudzu should trigger control activities. There is no acceptable population level (L.K. Thomas, Jr., personal communication).

B. Saltcedar - The presence of any saltcedar should trigger control activities, although it should be recognized that where stands are extensive, elimination is probably infeasible (P. Sanchez, personal communication).

C. Brazilian Pepper - The presence of any plants should trigger control activities, since this species is capable of displacing native vegetation.

3. <u>Management Alternatives-Nonchemical:</u>

A. Kudzu -

1. Cutting - Vines (including runners) are chopped just above ground level, and the pieces destroyed by burning or feeding to livestock. Early in the season, cutting is repeated at 2-week intervals, to weaken the crown and prevent resumption of photosynthesis. Later in the season, when the stored energy in the taproot has been reduced, the interval between cuttings can be extended (L.K. Thomas, Jr., personal communication). Cutting does not affect roots or crowns, which will regrow unless their supply of stored energy is depleted.

2. Flaming - A kerosene torch is played over the foliage, wilting the leaves, thus defoliating the plant. Flaming should be done according to the same schedule as cutting. Where all foliage can be reached, this method may be more effective than cutting. Like cutting, flaming does not affect the roots or crowns.

3. Burning - Destroys above-ground growth. Since kudzu vines usually will not burn during active growth (because of their high water content), vines may be flamed (see above) two or three days prior to burning. This causes the leaves to wilt and dry, providing fuel for the burning process.

4. Grubbing - This consists of mechanical removal and destruction of the entire plant, including the taproot. If all root tissue is removed, no regrowth can occur, so that repetition should not be necessary. However, this procedure can be destructive to the treated area. Removal of crowns only is more effective than cutting, but must be repeated, since remaining roots will resprout. Crown removal is most effective at flowering (when the plants are weakest) or in the fall. However, the crowns are difficult to find except in the spring, when the operation will be less effective.

5. Grazing - Kudzu is a favored food of goats and cows, which can provide useful levels of control. Where these can be accommodated in the park management plan, this technique can be effective.

B. Saltcedar -

1. Cutting - This process involves removal of all growth at ground level, but regrowth is not prevented.

2. Burning - This removes above-ground growth, but allows remaining roots and crowns to resprout.

3. Grubbing - Grubbing with a grubber blade, which is smaller than a root plow, and is used to remove smaller stands. This is less destructive than root plowing.

4. Root Pulling - Removal of the main portion of the root system and crown is labor and time intensive. Regrowth from incompletely-removed roots may occur.

5. Chaining - A chain, 360-400 ft long, and weighing 40-50 lb/ft, can be doubled and pulled between two crawler tractors. Chaining may uproot whole plants and/or may shear trunks at ground level. Drawbacks of chaining include the failure to remove all below-ground tissue, allowing regrowth; and the destructiveness of the procedure itself.

6. Root plowing - This process shears vegetation below the ground surface. The root plow kills medium to large shrubs by shearing below the crown, largely (but not completely) preventing regrowth. This technique is destructive to the environment but is widely used in areas where saltcedar coverage is nearly 100% (Gangstad, 1982).

7. Draglining - Drag lines are used to shear vegetation growing in water bodies or channel banks. It is not suitable for large vegetation.

8. Bulldozing - This shears plants at ground level, or uproots entire plants. Regrowth from sheared trunks can occur. This, too, is a destructive technique.

C. Brazilian Pepper -

1. Hand Removal - Seedlings or small saplings can be pulled from the soil. Pulled plants must be removed from their growing site and bagged or dried, to prevent resprouting.

2. Burning - Olmsted and Yates (1984) report that prescribed burning has kept a slash pine forest in Florida free of Brazilian Pepper seedlings.

3. Bulldozing - This technique has been used in the Everglades National Park (Olmsted and Yates, 1984).

4. <u>Management Alternatives-Chemical:</u>

A. Kudzu - Ammonium sulfamate and glyphosate are recommended for control of kudzu in the NPS. Bratton (1981) and Rosen (1982) report success controlling kudzu in two national parks using Roundup® (glyphosate) applied to the foliage two or three times each year.

B. Saltcedar - The following herbicides are registered for saltcedar control: 2,4-D; a mixture of dicamba + 2,4-D; and picloram. P. Sanchez (personal communication) reports that direct application of picloram to freshly cut stumps can provide effective control. The pesticide must be formulated in a nonevaporative (e.g., glycol) base, to prevent treated stumps from drying out before the pesticide has entered. Treatment should be repeated as necessary.

C. Brazilian Pepper - Non-woody seedlings can be treated with 2% Roundup® in water, as a foliar spray. Small woody saplings and established trees can be treated with 2% Garlon 4® (triclopyr butoxyethyl ester), applied as a spray to every major stem (complete coverage to runoff, at 12-15" above ground level). Treated sites should be monitored and surviving trees retreated at six week intervals following treatment, until regrowth no longer occurs.

Contact your regional IPM Coordinator to determine which, if any, herbicide is the best suited to your exotic weed management program.

5. Summary of Management Recommendations:

A. Kudzu - Regular cutting (or flaming, where applicable) may be sufficient to control most kudzu populations. Grubbing may control small infestations, if it will not result in too much destruction of other vegetation. Where it can be accommodated, grazing by goats may preclude the need for additional measures. For large overgrown areas, application of a recommended pesticide may be necessary.

B. Saltcedar - Individual plants can be grubbed from the soil. Cutting followed immediately by application of picloram to stump ends is the most effective means of controlling small stands of mature shrubs.

C. Brazilian Pepper - Small trees or individual seedlings can probably be mechanically pulled by workers wearing protective clothing. Prescribed burns may prevent establishment of seedlings in appropriate circumstances. Cutting and bulldozing may be useful against large trees and stands. Seedling stands and established trees may be treated with registered herbicides.

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V. PERSONAL COMMUNICATIONS

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NATIONAL PARK SERVICE
IPM Information Package

MOLES & POCKET GOPHERS

Final Report

30 September 1984

Submitted To:

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Arlington, Virginia 22202

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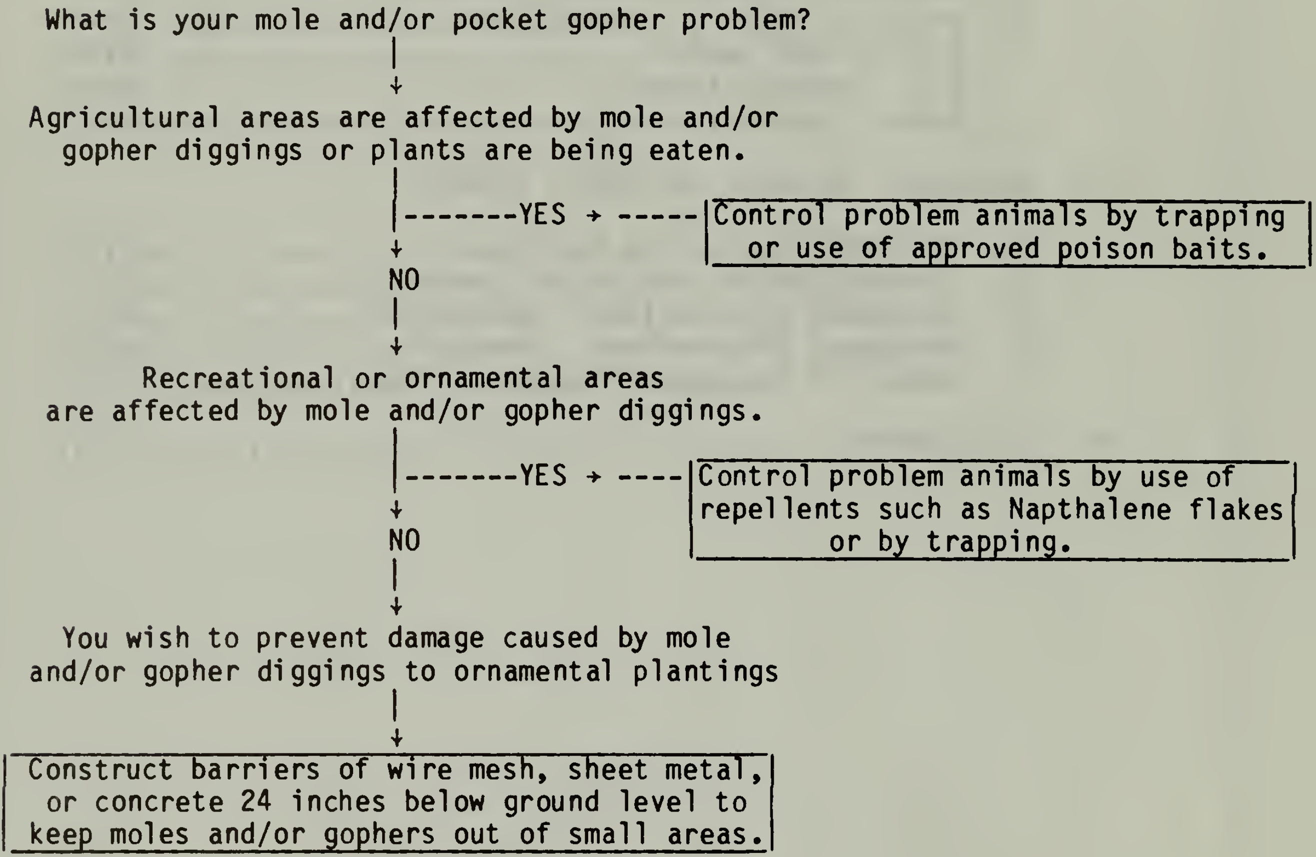
CONTENTS

I. MOLES AND POCKET GOPHERS IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



What is your mole and/or pocket gopher problem?

Agricultural areas are affected by mole and/or gopher diggings or plants are being eaten.

YES → Control problem animals by trapping or use of approved poison baits.

NO

Recreational or ornamental areas are affected by mole and/or gopher diggings.

YES → Control problem animals by use of repellents such as Napthalene flakes or by trapping.

NO

You wish to prevent damage caused by mole and/or gopher diggings to ornamental plantings

Construct barriers of wire mesh, sheet metal, or concrete 24 inches below ground level to keep moles and/or gophers out of small areas.

BIOLOGY AND ECOLOGY OF MOLES AND POCKET GOPHERS

Species Described:

1. Moles - Moles are Insectivores, related to shrews. They are members of the family Talpidae. Moles are burrowing mammals, rarely coming to the surface. Seven species in 5 genera occur in the U.S.; the most common species, and the one which is most often a pest, is the Eastern mole (*Scalopus aquaticus*). Most other species are local or uncommon.

   The adult Eastern mole is 4.5-6.5 inches in body length, with the tail an additional 1-1.5 inches. It weighs 2.5-5 ounces. The front feet are broader than long, with the palms facing outward for digging. The snout is pointed with the end naked, and the nostrils open upwards. The tail is naked. The eyes are pinhead size and covered with thin skin; there are no external ears. The fur has a silvery sheen; slate gray in the North, and brown to gold in the South and West. There are 6 mammae. The skull has 36 teeth.

   See Burt and Grossenheider (1964) for illustrations and detailed descriptions of North American moles.

2. Pocket Gophers - Pocket gophers are rodents, and comprise the family Geomyidae. There are fifteen species in three genera in the United States.

   Pocket gophers are burrowing mammals and are seldom seen on the surface. They are 4.75-9 inches in length, depending upon the species, and weigh up to a pound. They have external cheek pouches which are fur-lined and reversible, opening on either side of the mouth. The large, yellow incisors are always exposed in front of the mouth, even when it is closed. The front claws are large and curved for digging, the tail is short and sparsely haired or naked. The eyes and ears are small but functional. The fur is light brown to yellowish.

   See Burt and Grossenheider (1964), for illustrations and detailed descriptions of all species of pocket gophers.

2. Geographic Distribution:

1. Moles - Three species of moles occur in the East. The seven western species occur primarily on the Western slope of the Rockies. Moles are found throughout the U.S., except in the Rocky Mountain region and the Great Basin.

   The Eastern mole occurs from Massachusetts to Florida along the East Coast. It ranges as far west as eastern Colorado and Texas, north to Michigan and Wisconsin. It is not found in the mountains of West Virginia and Pennsylvania.

   See Burt and Grossenheider (1964) for detailed range maps.

2. Pocket Gophers - Pocket gophers occur throughout the Western and Southern portions of the U.S. One or more species occur in the following states: Washington, Oregon, California, Nevada, New Mexico, Arizona, Utah, Idaho, Montana, Wyoming, Colorado, Texas, Oklahoma, Kansas, Nebraska, South Dakota, North Dakota, Minnesota, Wisconsin, Iowa, Illinois, Indiana, Missouri, Arkansas, Louisiana, Alabama, Georgia, and Florida.

   See Burt and Grossenheider (1964) for detailed range maps for each species.

3. Habitat:

1. Moles - Moles prefer moist sandy loams. Meadows, fields, gardens, lawns, and golf courses are common habitats. Moles tend to avoid dry soils, and are seldom found in heavy clays, or stony or gravelly soils.

2. Pocket Gophers - Pocket gophers prefer slightly moist soils which are suitable for burrowing. Most species inhabit soils similar to those best suited for moles, but some western species are found in rocky soils in mountains.

4. Hosts:

1. Moles - Moles feed almost exclusively on earthworms, grubs, and soil inhabiting insects. Tunnel systems are used as traps; worms falling from the ceiling are captured and eaten. Moles will store prey for later consumption after immobilizing it with a bite. Moles also eat some plant material. Due to their high metabolic rates, moles eat 25% to 100% of their body weight in food every day.

2. Pocket Gophers - Gophers feed primarily on roots and tubers underground. Some surface vegetation is eaten after being pulled into the burrow. Gophers sometimes forage aboveground.

5. Life Cycles:

1. Moles - Moles are primarily solitary except during spring breeding season. After a gestation period of 6 weeks, 2-5 young are born in a grass-lined nest 18-24 inches below the surface. There is 1 litter per year. The young are naked at birth, independent after 1 month, and capable of breeding after 1 year. Moles are active year-round, day and night. Moles may live for several years.

2. Pocket gophers - Pocket gophers are solitary except during breeding season. They breed once per year in the northern part of their ranges and twice a year in the south. The plains pocket gopher (the most common species) breeds in April to July in the north, and twice between February to August in the south. The southeastern pocket gopher (the common eastern species), breeds in any month of the year, and usually has 2 litters of young. After a gestation period of 18-19 days, 1-3 young are born. The young are independent within 3 months, and are capable of breeding at one year of age. Pocket gophers are active day and night the year round, and are seldom seen aboveground.

6. Seasonal Abundance:

Populations of moles and gophers are highest just after the young are born and before natural mortality factors become prevalent.

7. Response to Environmental Factors:

1. Moles - The major environmental factors affecting populations of moles are soil type and associated availability of prey. Prey density is thought to account for the low densities of moles in most areas. A population of 2-5 moles per acre is considered high in most areas of North America, although high populations may not be injurious.

2. Gophers - Pocket gopher populations are affected by soil type and availability of preferred vegetation, primarily roots and tubers. There is some evidence for territoriality in males of some species. Males tend to have home ranges of approximately 2200 square feet, females have home ranges of about 1300 square feet. A density of 7-10 pocket gophers per acre is considered high.

8. Impact of Moles and Pocket Gophers:

8.1 Direct Impact:

1. Moles - The major impact of moles is the production of ridges of earth which are thrown up during tunneling. These ridges may have aesthetic impacts on lawns, golf courses, cemeteries, parks, and other ornamental areas. Tunneling does not damage the turf area.

2. Gophers - Pocket gophers are undesirable in fields and lawns primarily due to the mounds of earth thrown up during burrowing. These mounds may interfere with harvest of crops or recreational use of land. Gophers also eat plants and plant parts and are considered pests in agricultural areas, particularly alfalfa growing regions.

   Gophers have damaged irrigation canals and dikes by burrowing, and have damaged tree roots and lead-sheathed underground cables.

8.2 Indirect Impact:

Moles are considered beneficial in most circumstances due to their insectivorous diets, and the transport and aeration of soils caused by their tunneling activities.

Moles are often blamed for damage to plants caused by voles or other mice which may inhabit the tunnel systems.

Pocket gophers may be considered beneficial in many circumstances due to the transport and aeration of the soil during burrowing. It is estimated that each gopher transports over 2 tons of soil to the surface each year (Henderson, 1982).

9. Natural Enemies:

Moles and gophers are preyed upon by a wide variety of animals including snakes, weasels, coyotes, badgers, hawks, owls, dogs, and cats.

III. MANAGEMENT OF MOLES AND POCKET GOPHERS

1. Population Monitoring Techniques:

Moles and gophers are best monitored by noting ridges and mounds caused by burrowing. Control should be attempted only for those individuals which are directly interfering with activities.

1. Moles - Mole activity can be determined by the heaved ridges from near-surface tunnels, and by circular mounds of earth pushed to the surface.
2. Gophers - Pocket gopher activity can be identified by the presence of numerous large earth mounds. Gophers push earth up in lateral burrows which are 15 inches away from and at right angles to the main tunnels located 10 inches below the surface.

2. Threshold/ Action Population Levels:

Since naturally occuring population densities of moles and gophers are normally low, there are no established population thresholds. As noted above, control should be directed at individual animals, not on an area wide basis for entire populations.

3. Management Alternatives - Nonchemical:

1. Moles - Moles may be kept from small areas such as flower beds by placing sheet metal, concrete, or wire mesh barriers around the perimeter of areas to be protected. The barrier should extend downward for 2 feet to prevent moles from tunneling beneath it.

Populations of moles in turf areas may indicate high populations of insects (e.g. white grubs) which may be detrimental to turf. Monitoring of insect populations should be initiated before undertaking control of moles. See Turf Insects IPM Package for applicable techniques.

Traps in surface tunnels are effective control devices. Two types of traps, scissors and harpoon, are recommended for use in mole control. Several traps should be used. In large areas such as golf courses, 25-100 traps may be needed to reduce populations enough to offset immigration and reproduction.

Mousetraps can be used to trap moles. Tunnels are cut across, the trap is set perpendicular to the tunnel, with the trigger in the tunnel, or two traps are set back to back. A box may by placed over the hole to block light. It is not necessary to bait mousetraps used for mole control.

Pitfall traps may be used in circumstances where mechanical traps are not desirable. A large can is placed below the tunnel with the top of the can even with the bottom surface of the tunnel.

Traps should be placed where the surface tunnel is straight for several feet. Moles use straight tunnels more often than winding tunnels. To determine which tunnels are used most often, collapse a portion of the tunnel and check back the next day. Active tunnels will have been repaired.

The active burrow should be opened and the scissors trap placed in the tunnel with the jaws encircling the burrow. Harpoon traps are set straddling the burrow. Mark a map with all trap locations for retrieval. The opening should be covered with cardboard or wood. Check traps twice a day, if a trap is not sprung in 24 hours, move the location of the trap. If traps are set but no moles are captured, reset the trap further back in the tunnel, and reset the trigger.

See Henderson (1982) for details on mole management through trapping.

2. Gophers - Undergound fences can be used to protect tree plantings from gophers. A cylinder of 1 inch or smaller wire mesh 12 inches in diameter and 18 inches tall should be placed in the hole around the tree during planting. The top of the wire should be 1-2 inches under the soil surface to allow cultivation around the tree.

   Trapping pocket gophers is similar to mole trapping. There are three types of pocket gopher traps in use; the box trap, the spring trap, and the Macabee trap.

   Traps should be set near fresh gopher workings. To locate the tunnel, push the fresh mound aside and look for the earth plug where the gopher has filled a lateral tunnel. This can be determined by the subsoil, which is different from the surface soil in color. Dig down until the open burrow is reached (from 2-16 inches down). Set the trap far back in the tunnel. Loose soil can be used to partly cover the buried trap.

Attach a wire to the trap and anchor it to a stake. This will prevent a predator from dragging the gopher and trap away during the night.

Cover the entrance either partially or completely, and mark the site with flags for easier checking later. If a lateral tunnel is opened, set one trap. If a main tunnel is opened, set two traps back to back.

Pocket gophers travel their entire burrow system every few hours. Air coming into the system from the opening will also attract the gopher. As the gopher checks the burrow or tries to plug the opening, it will be caught in the trap. Traps should be checked every 4-8 hours.

Repeated misses by a spring trap, or a blocked burrow where the set traps have been buried, require adjustment in the trap or in trapping procedure.

4. <u>Management Alternatives - Chemical:</u>

1. Moles - Chemical control of moles has met with some success. Before considering chemical methods, consult with your regional IPM coordinator.

   Food source removal discourages moles and generally results in lower populations, but tunneling may increase before moles leave the area.

2. Pocket gophers - Pocket gophers have been controlled using poison baits such as chlorophacinone, or zinc phosphide on corn placed in burrows (Case, 1983). When soil conditions are right, a "burrow builder" which creates and baits artificial burrows intersecting the gopher's burrow system, has been used with some success (Henderson, 1982).

   Fumigants have been used in the past without much success due to the extent of the burrow systems, leakage into the soil, and plugs constructed within the system to keep out predators. Generally fumigants will not be recommeded for use on NPS lands.

   Consult your regional IPM coordinator to determine which pesticide, if any, is best suited to your IPM program.

5. Summary of Management Recommendations:

1. Protect small areas with barriers of sheet metal, screen mesh, or concrete.

2. Before trapping in turf areas, monitor turf insect populations which may be attracting moles. The presence of moles may indicate future turf insect problems.

3. Trap problem moles and gophers using approved traps in burrow systems.

4. Use approved poison baits to control individual problem animals.

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NATIONAL PARK SERVICE
IPM Information Package

Leaf Miners

Final Report

11 April 1985

Submitted To:

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Washington, D.C. 20240

Submitted By:

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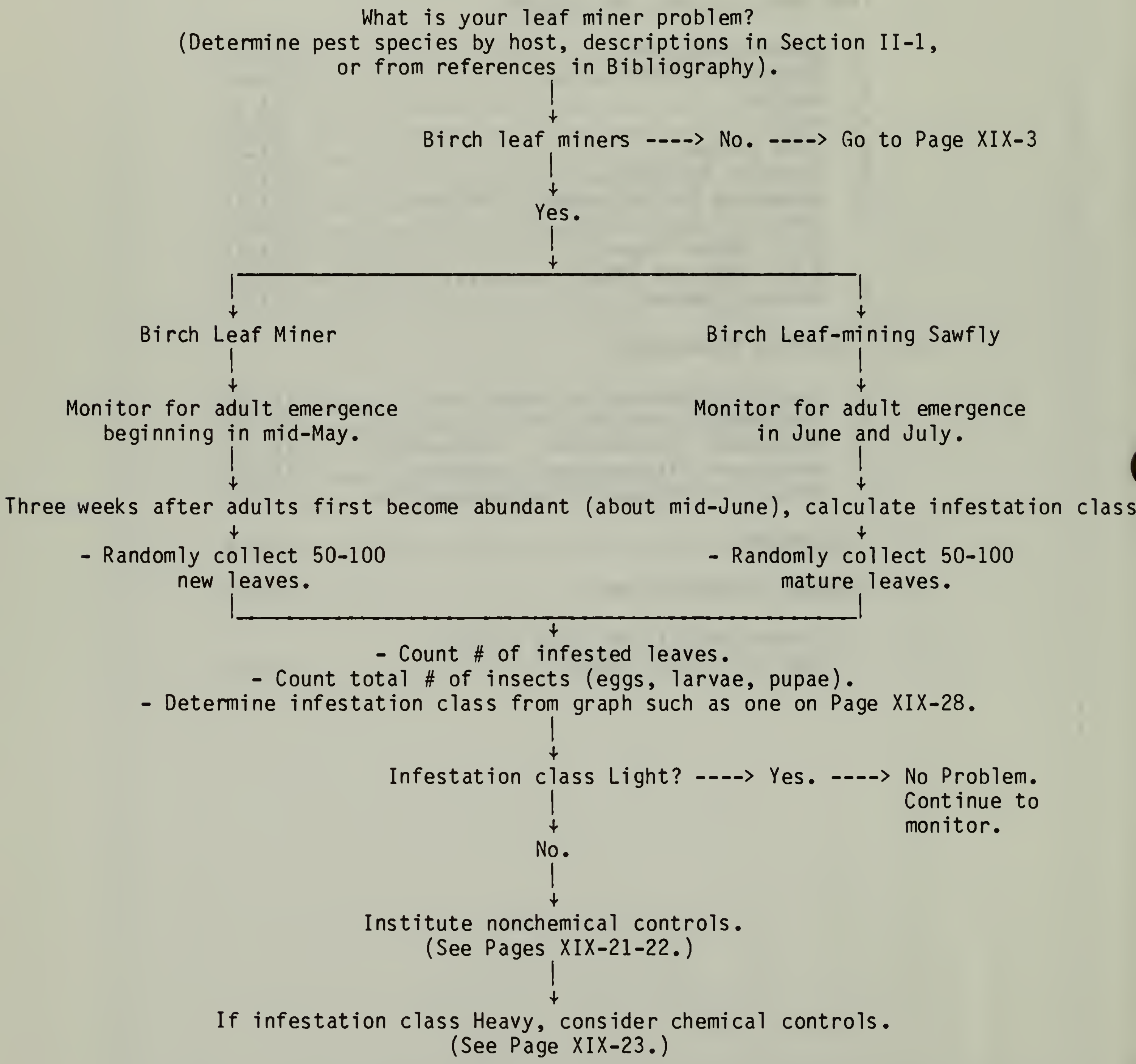
CONTENTS

I. LEAF MINER IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



What is your leaf miner problem?
(Determine pest species by host, descriptions in Section II-1, or from references in Bibliography).

Birch leaf miners ----> No. ----> Go to Page XIX-3

Yes.

Birch Leaf Miner

Monitor for adult emergence beginning in mid-May.

Birch Leaf-mining Sawfly

Monitor for adult emergence in June and July.

Three weeks after adults first become abundant (about mid-June), calculate infestation class

- Randomly collect 50-100 new leaves.

- Randomly collect 50-100 mature leaves.

- Count # of infested leaves.
- Count total # of insects (eggs, larvae, pupae).
- Determine infestation class from graph such as one on Page XIX-28.

Infestation class Light? ----> Yes. ----> No Problem. Continue to monitor.

No.

Institute nonchemical controls.
(See Pages XIX-21-22.)

If infestation class Heavy, consider chemical controls.
(See Page XIX-23.)

Boxwood Leaf Miner ----> No. ----> Go to Page XIX-4

↓

Monitor for adult emergence
in late April and May, when bush-honeysuckle (weigela) blooms.

↓

Four weeks after adults first become abundant, calculate infestation class.
- Randomly collect 50 new leaves.
- Count # of infested leaves.
- Count total # of insects (eggs, larvae).
- Determine infestation class from graph such as one on Page XIX-28.

↓

Infestation class Light? ----> Yes. ----> No Problem.
Continue to
monitor.

↓

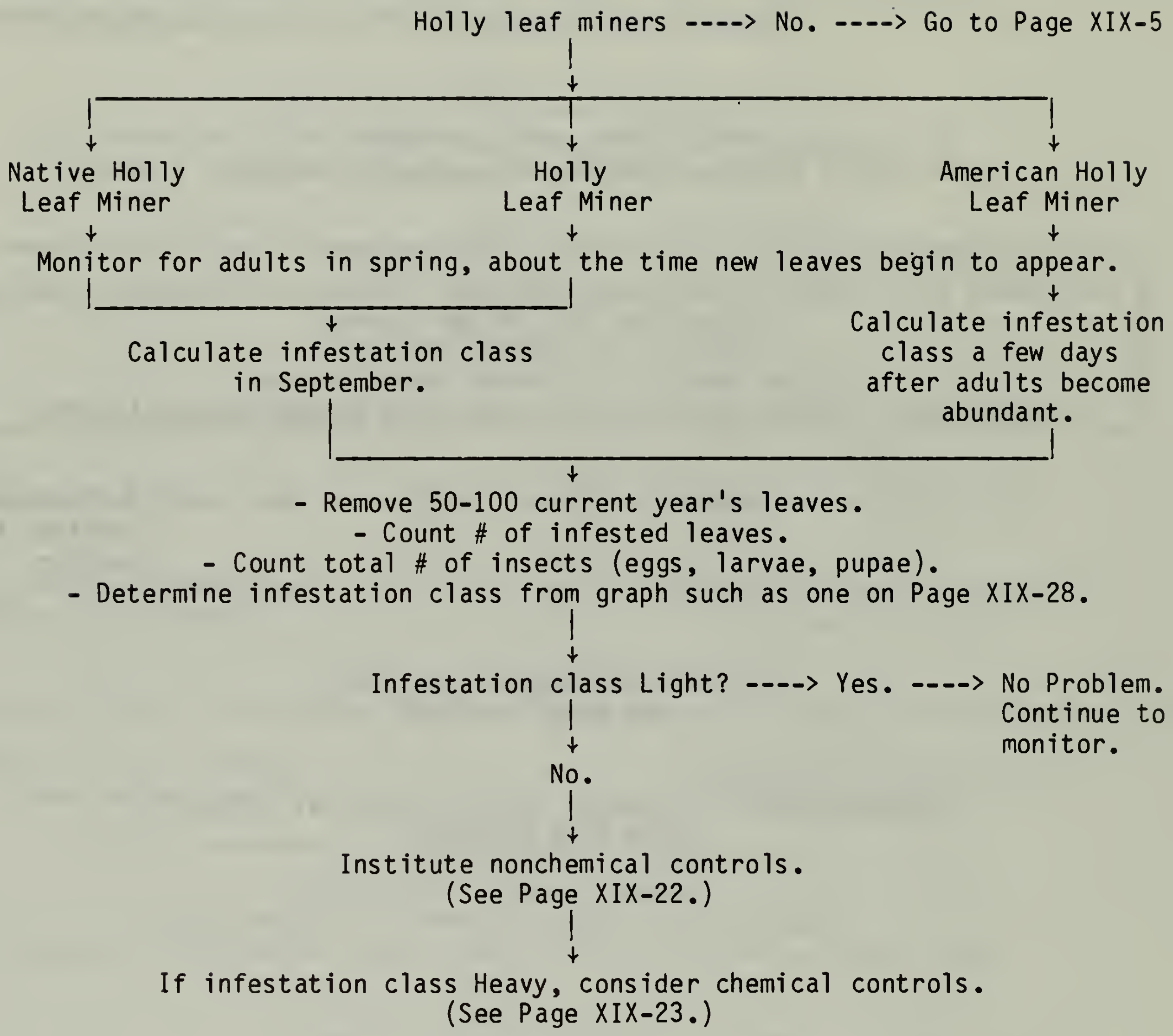
No.

↓

Institute nonchemical controls.
(See Page XIX-22.)

↓

If infestation class Heavy, consider chemical controls.
(See Page XIX-23.)



Holly leaf miners ----> No. ----> Go to Page XIX-5

Native Holly Leaf Miner

Holly Leaf Miner

American Holly Leaf Miner

Monitor for adults in spring, about the time new leaves begin to appear.

Calculate infestation class in September.

Calculate infestation class a few days after adults become abundant.

- Remove 50-100 current year's leaves.
- Count # of infested leaves.
- Count total # of insects (eggs, larvae, pupae).
- Determine infestation class from graph such as one on Page XIX-28.

Infestation class Light? ----> Yes. ----> No Problem. Continue to monitor.

No.

Institute nonchemical controls. (See Page XIX-22.)

If infestation class Heavy, consider chemical controls. (See Page XIX-23.)

Locust Leaf Miner ----> No. ---------------------------->

When damage first becomes apparent,
calculate infestation class.

- Randomly collect 5-10 leaves.
- Count # of infested leaflets.
- Count total # of insects (eggs, larvae, pupae).
- Multiply # eggs x 5, # larvae x 4.
- Determine infestation class from graph such as one on Page XIX-28.

Infestation class Light? ----> Yes. ----> No Problem.
Continue to
monitor.

No.

Institute nonchemical controls.
(See Page XIX-22.)

Lodgepole Needle Miner

Begin sequential sampling program on:
- June 15 of odd-numbered years for pupae.
- October 1 of odd-numbered years for 1st instar larvae.
- September 15 of even-numbered years for 4th instar larvae.

1) Use pole pruners to collect 5-internode tips from midcrown of trees.
2) Count number of live insects; add to previous total.
3) If cumulative total of live insects falls within infestation class on graph on Page XIX-29, discontinue sampling, otherwise return to step 1).

Infestation class light or medium. ----> No problem.
Continue to monitor.

Infestation class heavy. ----> Institute nonchemical controls.
(See Page XIX-22.)

Infestation class heavy. ----> Consider chemical controls.
(See Page XIX-23.)

II. LEAF MINER BIOLOGY AND ECOLOGY

Leaf miners are insects that live as larvae and sometimes as pupae inside the leaves of plants and feed on the soft tissues between the upper and lower surfaces. The spaces the larvae hollow out by feeding are called mines. Many species of insects, primarily in the orders Coleoptera (beetles), Lepidoptera (moths), Diptera (flies), and Hymenoptera (sawflies), mine the leaves of plants. This Information Package deals with selected species of concern to the National Park Service. Most of these leaf miners are more easily recognized on the basis of their host plant and the type of mine they make, than on the basis of structural features of the insects themselves.

1. Species Described:

A. Birch Leaf Miner (BLM) - *Fenusa pusilla* (Lepeletier), a sawfly of the family Tenthredinidae. The oval eggs are about 0.5 mm (1/50 inch) long. Full grown larvae are somewhat flattened, and about 6 mm (1/4 inch) long. They are yellowish-white in appearance, and, except for the first and last (5th) instars, they have distinctive black spots on the underside of the thorax and first abdominal segment. The legs are small and far apart. Larvae form a common blotch mine that contains frass (excrement). Pupae are about 4 mm (1/16 inch) long, and are white at first, changing to black as the adult develops. Adults are 3-5 mm (1/8 - 3/16 inch) in length, and black. Color photographs of larvae, mines, and adults are presented in Johnson and Lyon (1976).

Birch Leaf-mining Sawfly (BLS) - *Heterarthrus nemoratus* (Fallen) is similar in appearance to BLM. Eggs are oval and about 0.5 mm long. Newly hatched larvae are soft and tapering, with a broad, flat thorax. The body is whitish and the head, brownish. Tiny, useless legs project sideways from the thorax. Later-stage larvae are similar, but tend to be more yellowish and have darkened areas on the first thoracic segment. Sixth instar larvae measure 6.5-10 mm (1/4-3/8 inch) in length; the thorax is flattened and the abdomen is cylindrical. The seventh instar larvae do not feed and differ in appearance from the earlier feeding instars: the body is whitish in color, including most of the head, and is flattened throughout and measures 6-8.8 mm in length. Larvae form a single large blotch mine that does not contain frass. The pupa is 6 mm (1/4 inch) in length, and is entirely white at first, gradually developing the black coloration of the adult insect. Adult females also are about 6 mm long, and are black with fine pale yellow or whitish markings on the head, thorax, legs, and edges of the upper side of the abdomen. Males are unknown. Descriptions and illustrations of all stages, including mines and hibernacula, are provided by Peirson and Brower (1936).

B. Boxwood Leaf Miner (BXM) - Monarthropalpus buxi (Laboulbene) is a fly of the family Cecidomyiidae. Larvae are yellowish or white, and less than 3 mm (1/8 inch) long when full grown. The body is slightly flattened, widest at about 1/4 of the distance from the head, and tapered to the rear end. The last segment of the abdomen has two minute papillae, or processes. Larvae make common mines in the undersides of leaves marked by blister-like swellings of the leaf surface. Pupae are about the same size as mature larvae, widest at the front and tapered to the rear, and are reddish brown when mature. Adults are about 2.5 mm long, and look like tiny, frail, orange-red mosquitoes. Color photographs of all stages, including mines, are presented in Johnson and Lyon (1976) and Baker (1980).

C. Holly Leaf Miners - Phytomyza species are flies of the family Agromyzidae. Seven species are recorded mining leaves of holly (Ilex spp.) in North America (Kulp 1968). Three species are of concern to the NPS: Phytomyza ilicicola Loew, the Native Holly Leaf Miner (NHLM); P. ilicis Curtis, the Holly Leaf Miner (HLM); P. opacae Kulp, the American Holly Leaf Miner (AHLM). All of them are similar, but can be distinguished by host preferences and type of mine.

Eggs are white, tapering at both ends, but slightly blunted at one end, and are 0.25 mm in length. Larvae are typical fly maggots, shaped like an elongated cone with the mouth opening at the pointed end and the anal opening and breathing holes at the blunt end. They range from colorless to yellow to pale green in color. Mature larvae measure 0.9-3.9 mm (1/64-5/64 inch) in length. NHLM larvae form mines that begin near the edge of the leaf, are narrow linear at first and widen into irregular blotches as the larvae mature. HLM larvae form small mines in the midrib initially, then move into the blade of the leaf and form blotch mines. AHLM form linear mines in all instars, each yellow-orange mine traversing a leaf 2-3 times. Puparia are pale white or yellow at first, becoming reddish brown or black as they mature. They range from 1.6-3.0 mm (1/16-1/8 inch) in length. The adults measure 1.4-2.8 mm in length, with the females slightly larger than the males of the same species. They are grayblack in color, and the abdomen is tapered and blunt at the end. Detailed descriptions of all stages, illustrations of the mines, and keys to adults are given in Kulp (1968).

D. Locust Leaf Miner (LLM) - Xenochalepus dorsalis (Thunberg). This is a small leaf-mining leaf beetle (Chrysomelidae: Hispinae). Full grown larvae are about 9 mm (3/16 inch) long. The head and legs are black, as are large areas of the top and underside of the first thoracic segment and the tip of the abdomen; the rest of the body is yellowish white. Larvae are somewhat flattened and about the same width throughout. Abdominal segments 1-8 are cone-shaped on each side, with darkened protuberances at the tips of the cones of segments 2-8. Larvae form irregular blotch mines. The adults are about 6 mm long and slightly flattened. Their overall appearance is wedge-shaped, with a small head and pronotum and the wing covers (elytra) widening posteriorly and bluntly rounded apically. The head, antennae, and legs are black; the pronotum is orange, and the wing covers are orange except for a streak of black along the inner margin of each elytron. Each wing cover has 10 rows of indentations and three raised longitudinal ridges. A color photograph of larval mines and adults is given in Johnson and Lyon (1976), and descriptions of adults, pupae, and larvae are available in Needham et al. (1928) (as Chalepus) and Baker (1972) (as Odontota).

E. Lodgepole Needleminer (LNM) - Coleotechnites milleri (Busck) is a moth of the family Gelechiidae. Eggs are lemon yellow, ovoid, and 0.2-0.3 mm long. Larvae are usually a fairly uniform lemon yellow, but shades of orange, pink, and red also occur; the head and top of the prothorax are brown to black. Full grown larvae are about 7 mm (just over 1/4 inch) in length. Mined needles are recognized by the small hole near the base made by the larva and the yellowish discoloration. Pupae are about 6 mm (1/4 inch) long, and darken to jet black as they mature. The face of the adults is white and the rest of the head, the thorax, and the front wings are light gray mottled by irregular darkened areas; the hind wings are dusky gray. The male abdomen is silvery gray and has hairy claspers at the end; the female abdomen is cigar-shaped, and is light gray at first, turning yellowish as the eggs mature. Body length ranges from 4 to 4.5 mm (less than 3/8 inch), and wingspread is about 12 mm (1/2 inch). Descriptions and photographs of all life stages are presented in Struble (1972).

2. Geographic Distribution:

A. BLM - This species, introduced to North America from Europe, was discovered in Connecticut in 1923, and now is widespread throughout the Northeastern States and southeastern Canada.

BLS - Introduced to North America from Europe in the late 1800's and first discovered here in Nova Scotia in 1905, this species now is widely distributed in southeastern Canada and the Northeastern States.

B. BXM - Another pest introduced from Europe, this one first was reported in the U.S. in 1910, and now is found coast to coast wherever boxwood is grown.

C. NHLM - Connecticut to Ohio, south to Tennessee and Virginia.

HLM - Introduced from Europe with imported holly, this species now is found in British Columbia, Oregon, and Washington in the West, and where its host is grown in the East.

AHLM - From New Jersey to Washington, D. C.

D. LLM - Eastern North America as far west as Missouri.

E. LNM - Cascade Mountains of Washington and Oregon, and Sierra Nevada Mountains of California.

3\. *Habitat:* All of the species described in this Information Package live, as larvae, inside the leaves of their host plants. Adults rarely stray far from these plants. See *Hosts*, below.

4\. *Hosts:*

A. BLM and BLS - Most species of birch (*Betula* spp.); the BLS also occasionally attacks alder and hazlenut. Although they develop on most species of birch, they grow faster on gray birch, *Betula populifolia*, white birch, *B. papyrifera*, and European white birch, *B. alba*, than on other species. Females of BLM failed to oviposit in cage tests with a Korean birch, *B. davurica* (Fiori and Dolan 1984).

B. BXM - Boxwood, *Buxus sempevirens*, although English varieties are less susceptible than American varieties. Also *B. microphylla* and *B. harlandii*.

C. NHLM - American holly, *Ilex opaca*; also *I. cumulicola*, and one variety of English holly, *I. aquifolia* var. Shepherd.

HLM - English or Christmas holly, *I. aquifolia*.

AHLM - American holly; also *I. cumulicola* and some varieties of English holly.

D. LLM - Black locust, *Robinia pseudoacacia*; adults also feed on dogwood, elm, oak, beech, cherry, wisteria, hawthorn, and several herbaceous plants.

E. LNM - Lodgepole pine, *Pinus contorta murrayana*; occasionally on other pines and firs when larval densities are extremely high.

5\. *Life Cycles:*

A. BLM - There are 2-4 generations each year depending on the length of the growing season. Adults begin to appear about the middle of May, when the first leaves of gray birch are fairly well developed. The adults do not feed. Females deposit eggs singly in slits cut in the central area of newly developing leaves (never in older mature leaves), and prefer the lower reaches of the tree. Each female lays about 22 eggs per day for a period of several days. A female usually lays only a few eggs in a leaf, but several females may oviposit in the same leaf; as many as 63 eggs have been counted in one leaf, although the average is nine (Friend 1933). Eggs hatch in 6-10 days and the larvae begin to mine the leaf. At first, mines are separate and small but soon coalesce to form a single large blotch mine containing several larvae. Final instar larvae do not feed. Larvae mature in 10-15 days, cut a hole in the leaf and drop to the ground. They work their way through the leaf litter and humus, then burrow 1-2 inches into the soil and build a small silklined cell in which they pupate. Adults emerge after 2-3 weeks. Mature larvae of the final generation of the season overwinter in their underground cells, and pupate in the spring.

BLS - This species consists entirely of females and has a single generation per year. Adults emerge in June and July, and within a few hours they begin to lay eggs. Females prefer healthy mature leaves exposed to sun and air, and avoid young leaves and those in shaded and protected areas. Each female deposits a single egg in the tip of a leaf-tooth, most often in the apical 2/3 of the leaf. Females deposit from 22-67 eggs each, with egg laying lasting for about one week. Eggs hatch in 12 to 26 days, and after several hours the larvae begin to mine toward the center of the leaf. As the larvae develop they form a single large blotch mine in the leaf. Although 10 or more eggs may be deposited, rarely are more than five larvae able to mature in a single leaf, and most leaves support only two or three larvae. Larval development time varies, but generally lasts about 58 days. The 7th, nonfeeding instar makes its pupal chamber, or hibernaculum, within the mine near the center of the leaf. The hibernaculum is roughly circular and lens-shaped, and is formed from silk secreted by the larva. The silk hardens into a tough, waterproof, parchment-like substance, and the larva overwinters within its hibernaculum in the leaf on the ground. Pupation occurs during June and July, and the pupal stage lasts eight or nine days.

B. BXM - This species has a single generation per year. Adult flies emerge in late April and May during the time bush-honeysuckle (weigla) is in bloom (Baker 1980). Females lay eggs in new leaves, leaving conspicuous punctures. The females die within hours of completing egglaying, and the larvae hatch about three weeks later. A single leaf may contain a dozen or more larvae. Larvae overwinter inside the leaves, and resume feeding in the spring. Pupation occurs in April, and the pupal stage lasts about 10-14 days.

C. NHLM - This species has one generation per year. Adult flies begin to emerge in the spring after the plant has begun to produce new leaves. Adults of both sexes feed on sap flowing from holes made in the leaves by the ovipositor of the females. About 10 days after emerging, the females begin to lay eggs, depositing them in the soft tissues on the underside of young leaves. Larvae hatch soon thereafter. Because the larvae are so small, they frequently go unnoticed until late in the year when their mines are larger. Second instar larvae begin to appear in October, and third instars by December. Some pupae appear in January, but mostly larvae overwinter and pupate in the spring.

HLM - Very similar to the NHLM, however, the females oviposit in the midrib of the leaf, and the first instar larvae remain in the midrib until September, when they move into the blade of the leaf and mine there.

AHLM - The biology of this species is largely unknown, but there probably are several generations per year; larval development from the first appearance of a mine to the appearance of adults takes only a few days. Adults first appear in late May, and also have been captured as late as August.

D. LLM - This beetle has one generation each year throughout the northern portion of its range, and two generations each year from Ohio south. Adults emerge from hibernation in the spring as the new leaves begin to unfold. They feed on the underside of the leaflets, chewing holes in young leaflets and skeletonizing older ones. Eggs are laid in groups of 3-5 each, overlapped like shingles, glued together and partially covered with frass. The first larva to hatch from a group of eggs makes a small hole in the leaflet and enters it to begin feeding; the other larvae follow the first through this hole as they hatch. After feeding together on the mesophyll within this common mine for 2-4 days, they have eaten half or more of the leaflet. They then leave the leaflet and each larva searches out its own new leaflet and starts a new mine. As it matures, which takes about three weeks, each larva mines several leaflets. Pupation occurs within the final mine, and the pupal stage lasts 7-10 days. Adults emerge and begin feeding on the underside of leaflets until fall, when they seek sheltered places, such as in the leaf litter under the host tree or in crevices in the bark, in which to overwinter.

E. LNM - Two years are required for the LNM to complete its life cycle. Each new cycle begins in the summer of odd-numbered years. Adults begin emerging in July and continue emerging for about three weeks. Males usually begin emerging about 10 days before females. Oviposition begins about 24 hours after mating. Eggs are deposited in groups of 4-11 in the current year's growth in mines that have been vacated by third or fourth instar larvae, or in older, previously mined needles still firmly attached and green. Eggs hatch in 35-60 days, and the first instar larvae search out fresh green needles in which to overwinter. Rarely is there more than one larva per needle. Larvae resume feeding the next year and pass through the second instar and into the third within the original needle chosen by the first instar. Some third instar larvae leave the initial needle and migrate to new growth needles, while others continue to mine the first needle. All fourth instar larvae leave their needles and migrate to new growth needles. In trees with stunted needles as the result of heavy infestations, each larva may mine four or more needles. Fourth instar larvae overwinter, and resume feeding in the spring of the following (odd-numbered) year. Beginning in mid-April, the larvae molt to the fifth instar and mine one or more needles before pupating inside the final mine in mid-June. The pupal period lasts about 30 days.

6. <u>Seasonal Abundance:</u>

A. BLM - Females oviposit only in new growth, so populations decline as the number of new leaves declines through the season, although adults are active throughout the summer.

BLS - Populations are greatest in July, but are most noticeable in August and September when the mines are large and evident. Adult activity is greatest in early summer.

B. BXM - Adult activity is restricted to a two week period in late April and May. The presence of larvae becomes progressively more obvious throughout the summer as the mines become more noticeable.

C. NHLM - Adults can be observed in the spring, but larvae are most noticeable late in the year as they mature and their mines increase in size.

HLM - See NHLM.

AHLM - Because the AHLM has multiple generations, it is most abundant late in the season.

D. LLM - Populations are largest in the summer and fall.

E. LNM - Adults emerge in July of odd-numbered years. Larval populations are greatest in the late summer or fall of odd-numbered years when first instars hatch from the eggs and migrate to new needles.

7. <u>Responses to Environmental Factors:</u>

A. BLM - Females oviposit only in new growth.

BLS - Dry conditions increase mortality of eggs. Early frosts may result in early leaf-drop, resulting in the death of larvae within the leaves. Prepupae that fall into moist areas suffer greater mortality than those that fall into drier areas. Adults are weak fliers and are easily carried by the wind. They are most active on sunny mornings when the air temperature is 65-85°F. On cool rainy days, or hot afternoons, they cling to the foliage and avoid movement.

B. BXM - Information is not available.

C. NHLM - This insect prefers plants in sunny, exposed sites over those in partially shaded sites (Davidson and Holmes 1980).

HLM - Information is not available.

AHLM - Information is not available.

D. LLM - This species has one generation per year in the northern part of its range and two in the southern parts of its range, indicating that the life history is influenced by temperature or photoperiod or both.

E. LNM - Climatic changes are considered the most important natural factors regulating LNM populations (Struble 1972). Unseasonally low temperatures in late spring and early summer delay pupation and emergence. Mating and oviposition stop when air temperatures drop below 50°F, the wind speed exceeds 5 mph, or during rains. Eggs take longer to hatch and mortality is higher when temperatures are below normal. Overwintering larvae in needles above the snow line can be killed by unusually cold temperatures. In addition, wind, rain, and hail can kill larvae by dislodging infested needles.

8. Impact of Leaf Miners:

8.1 Direct Impact:

A. BLM - See BLS.

BLS - Mining removes photosynthetic material from the tree, resulting in lowered growth rates and a general loss of vigor. Trees seldom are killed; however, Peirson and Brower (1936) found an average loss of 20% annual growth of white birch as a result of heavy infestations of BLS in Maine. In addition, the amount of heart wood was increased.

B. BXM - New growth may be stunted and twigs may die as a result of infestation.

C. NHLM - Mining by the larvae removes photosynthetic material from the leaf, and feeding punctures made by the females result in holes, and twisted, stunted leaf growth. Leaves having three or more mines fall prematurely.

HLM - See NHLM.

AHLM - See NHLM.

D. LLM - Leaf-mining by the larvae and feeding by the adults destroy leaves and impair the health of the trees. Loss of most of the leaves on a tree for two or more seasons in a row may result in the death of the tree.

E. LNM - Unmined portions of infested needles, and uninfested needles in the same fascicle (group of needles) with infested needles, turn yellowish to golden within 11 months of larval attack. Needle loss is great because one infested needle can result in the entire fascicle dropping. Trees become yellowish and appear scorched within a year after infestation, and the crowns become thinner. Infested trees may lose 90% of their needles in the first generation of an outbreak. In successive generations, needles of the terminal shoots are conspicuously shorter than normal needles, and the number of new needles is lower than normal. Terminal growth is severely shortened, and tree mortality during outbreaks is extensive. Outbreaks may last as long as 20 years. Entire forests of lodgepole pines were killed in Yosemite National Park from 1953 to 1963, and others were severely damaged. Mature and overmature stands are most susceptible.

8.2 Indirect Impact:

A. BLM - Leaf discoloration caused by mining may be very unsightly. Loss of growth may be detrimental to commercial users of the trees, and the loss of leaves over consecutive seasons may weaken the trees and make them more susceptible to further damage by other insects and disease.

BLS - See BLM.

B. BXM - The ornamental value of boxwood plantings may be severely impaired.

C. NHLM - Larval mines and deformed, stunted leaves resulting from adult feeding punctures make infested plants unattractive and commercially unprofitable.

HLM, AHLM - See NHLM.

D. LLM - See 8.2.A.

E. LNM - Trees weakened by LNM attack are predisposed to attack and death by the mountain pine beetle. The "ghost forests" created by massive mortality of lodgepole pines killed as a result of attack by the LNM have become a "recreational curiosity and tourist attraction" in Yosemite National Park (Dahlsten and Dreistadt 1984).

9. *Natural Enemies:*

A. BLM - Four species of parasitic wasps are recorded from the BLM: *Chrysocharis pallipes* Gahan, *Closterocerus utahensis* Crawford, *Derostenus fullowayi* Crawford, and *D. diastotae* Howard. Two stink bugs (*Podisus maculiventris* Say and *P. placidus* Uhler) feed on the larvae, and an assassin bug (*Sinea diadema* Fabricus) feeds on the adults. In addition, *Polistes pallipes* Lepeleiter, a paper wasp, feeds on the BLM, and ants eat prepupae on the ground.

BLS - Prepupae in their hibernacula are susceptible to attack by fungi, although it is likely that only larvae that have been injured are normally susceptible. Several species of parasitic wasps attack the immature stages. These include *Trichogramma minuta* Riley and *Cirrospilus flavicinctus* Riley, which parasitize the eggs, *Cirrospilus cinctithorax* Girauld, which parasitizes both eggs and young larvae, and *Agrothereutes slossonae* Cushman, *Epiurus indagator* Creson, *Alophosternum foliicola* Cushman, *Gelis urbanus* Brues, and *G. bucculatricis*, all of which attack mature larvae, prepupae, and/or pupae. In addition, the following species have been introduced into the United States from Europe: *Chrysocharis laricinellae* (Ratzeburg), *Chrysocharis* sp., *Phanomeris phyllotomae* Muesbeck, *Scambus foliae* Cushman, and *Tranosema pedella* (Holmgren). *C. laricinellae* and *P. phyllotomae* became established and have been successful in controlling the BLS. (Most insect natural enemies of leaf miners lack common names and are not commercially available.)

There are several predators of the BLS, including mice, shrews, and birds. Birds are the most important, with some, such as chickadees, warblers, and goldfinches, feeding on larvae in mines on the tree, and others, such as sparrows, robins, and juncos, feeding on prepupae and pupae in hibernacula on the ground. Ants eat BLS larvae they remove from mines on the trees, and prepupae and pupae they remove from hibernacula on the ground. When adult BLS are sluggish in unfavorable weather, ants attack them, also. Other predators include the adults of some parasitic wasps, ground beetles, wireworms, and lacewings.

B. BXM - Information is not available.

C. NHLM - One braconid (*Opius striatriventris*) and two eulophid (*Closterocerus tricinctus* and *Pediobius lithocollectidis*) wasps have been reported parasitizing this species (Kulp 1968).

HLM - Five species of parasitic wasps were imported from England and released in British Columbia in the late thirties and early fourties: *Chrysocharis pubicornis* (Zetterstedt) (=*syma* Walker), *C. gemma* (Walker), *Cyrtogaster vulgaris* Walker, *Opius ilicis* Nixon, and *Sphegigaster flavicornis* (Walker). All but the first became established (Clausen 1978).

AHLM - The following parasitic wasps were recorded from this species by Kulp (1968): *Opius dimidiatus* and *O. striatriventris*, Braconidae; *Closterocerus tricinctus* and *Pediobius lithocollectidis*, Eulophidae.

D. LLM - Weaver and Dorsey (1965) recorded 12 species of parasitic wasps from eggs, larvae, and pupae of the LLM. These species belong to the following families: Ichneumonidae, Eulophidae, Trichogrammatidae, Chalcididae, and Scelionidae. One species of eulophid, *Closterocerus tricinctus* (Ashmead), was found in over 50% of LLM pupae examined. These authors also recorded one vespid wasp predaceous on larvae, and an assassin bug predaceous on larvae and adults.

E. LNM - Larvae are susceptible to a granulosis virus, which has been reported to kill as much as 50% of the population in localized areas. Over 20 species of parasitic wasps were recorded by Struble (1972), as well as many predators, including mites, spiders, brown lacewings, thrips, minute pirate bugs, and flies. Several birds feed on maturing larvae and pupae, and on adults in flight.

III. LEAF MINER MANAGEMENT

1. Population Monitoring Techniques: Record all information from monitoring efforts on a form such as the sample leaf miner monitoring form shown on Page XIX-30. An infestation index is calculated using the approach shown on Page XIX-28, and following the principles discussed on Page XIX-21.

A. BLM - The first emergence of adults is monitored beginning in mid-May by sticky traps or by sweeping the lower branches of trees with an aerial insect net. Sticky traps should be slightly larger than and about the same shape as the leaves, made of white, yellow, or light green plastic, and coated with a commercial sticky substance such as Tanglefoot®. The traps should be placed in the trees among the most actively leafing lower branches. The traps should be examined daily and the number of adults recorded; replace traps that have 10 or more adults adhering. Three weeks after the adults have appeared in large numbers, randomly pick 50 to 100 leaves off the lower branches of a tree (the larger the number of leaves sampled, the more confident you can be of the results, however, you must balance the confidence with the effort involved and the total number of trees monitored). Count the number of leaves containing eggs and larvae and the number of eggs and larvae per leaf. These are easily detected by holding the leaf up to the light. Calculate the infestation index.

BLS - Monitoring techniques are very similar to 1.A, except that there is only one generation per year, the adults emerge in June and July, and the females prefer to oviposit in mature leaves in exposed portions of the trees. If sticky traps are used, they should be dark green, similar to the darker mature leaves preferred by the BLS for oviposition, and placed among the older leaves of the tree.

2. BXM - Begin monitoring for adult emergence in late April, when bush-honeysuckle blooms. Use direct observation, insect aerial nets, or sticky traps similar in color to the new leaves. Following the appearance of the adults, examine plants regularly for signs of oviposition in the leaves. Four weeks after

peak adult emergence, larvae are monitored by examining 50 randomly chosen leaves from each plant or group of plants for the presence of mines. Calculate the infestation index.

C. NHLM - In the spring, at about the time of the emergence of the new leaves, use sticky traps similar in color with the new leaves to monitor adult emergence. In August or September, randomly select 50 of the current year's leaves from each plant or group of plants and examine them for larval mines. Calculate the infestation index.

HLM - See NHLM.

AHLM - Use the same techniques as for the NHLM, but since the AHLM is multivoltine (more than 1 generation per year), sampling for larval mines must begin within a few days of the first emergence of the adults, and continue throughout the season.

D. LLM - When damage becomes noticeable, begin quantitative monitoring by randomly selecting 5 to 10 leaves; each leaf consists of about 7-15 leaflets, and the leaflets will be the sample units. Select leaves from lower, middle, and upper reaches of the tree, using extension loppers for the higher leaves. Count the total number of eggs, larvae, and pupae. To account for the fact that larvae mine several leaflets each, multiply the number of eggs by 5 and the number of larvae by 4. Calculate the infestation index.

E. LNM - Stevens and Stark (1962) present a method for sequential sampling of LNM populations. Sequential sampling means that samples are taken and counts of larvae are added to previous counts until the results fall into one of four infestation classes (Light; Medium; Heavy; Very Heavy). Sample units are 5-year branch tips, unless infestations over a period of years have reduced the needle complement severely; in Yosemite National Park, 2-year tips are used. Samples are taken from midcrown using 12 foot pole pruners. At each site, at least 12-15 trees, distributed over an area of about 1/2 acre, are sampled. All live insects are counted, and the total from each sample is added to all previous totals from the site until the total of live larvae falls into one of the decision classes established by Stevens and Stark (1962) (see Page XIX-29 for sequential sampling decision-making graph). Surveys begin in odd-numbered years: (1) October 1 for first instars; (2) September 15 for fourth instars; (3) June 15 for pupae.

2. Threshold/Action Population Levels:

A-D. Control of leaf miners in natural areas is rarely necessary and normally not recommended. Aesthetic thresholds for leaf-mining insects on ornamental plants are difficult to establish. Each situation is likely to be different and will require that the pest populations be monitored and an infestation index be calculated that can be used to determine whether or not to implement control measures. A sample infestation index is shown on Page XIX-28. The values in this example are only guidelines; it is likely that for each situation encountered, the limits of the infestation classes will be different. For example, a holly bush along a path heavily used by visitors will have a lower aesthetic threshold than one growing in an area where it is viewed from many yards away. In the first case, the lines separating the infestation classes in the sample graph may have to be moved to the left, and in the second case, the lines may be moved to the right. Set threshold and action levels appropriate to each given situation. After adjusting the infestation classes to the situation, consider implementation of nonchemical control measures when the infestation class is Moderate to Heavy, and chemical control measures in critical areas when the infestation class is Heavy.

E. LNM - Struble (1972) established threshold and action levels for LNM in Yosemite National Park, but currently there are no accepted levels for LNM occuring in other environments (T. Hofacker, USFS, pers. comm.).

3. Management Alternatives - Nonchemical:

A. BLM - Weakened and stressed trees are more susceptible to attack than healthy trees. Maintain vigorous growth with sound silvicultural practices. Prune water sprouts and suckers in late summer or fall. In certain circumstances, it may be feasible to kill a significant proportion of the pupae in the ground by tilling to a depth of 3-4 inches under an infested tree. Care must be taken to prevent damaging shallow roots, however. Parasites and predators may be encouraged by planting nectar producing flowers nearby, and predators may also be encouraged by use of food sprays to attract adults (see Rabb et al. [1976] for a review of various natural enemy augmentation methods).

BLS - Excellent control of BLS in the Northeast has been obtained through the introduction of parasitic wasps from Europe (see Page XIX-17). In ornamental plantings, adequate control may be achieved by raking and destroying leaves in the fall, eliminating the pupae.

B. BXM - Prune infested tips in the spring before emergence, and collect and destroy all pruned tips. Where possible, plant resistant English varieties.

C. NHLM - Plant holly in partial shade, as trees in full sun are more heavily infested (Davidson and Holmes 1980). In light infestations, control may be achieved by removing and destroying infested leaves; heavily infested plants should be pruned to remove damaged growth and the prunings destroyed to reduce NHLM populations. Encourage parasites (e.g., by spot treatments of infested trees, planting of nectar sources for adults); their presence can be determined by closely examining the leaves for exit holes made by the adults. These are almost perfectly round, in comparison with "hatch door" exit holes made by the flies (Carol DiSalvo, NCR, NPS, pers. comm.).

HLM - See NHLM. Parasites imported from England became established in British Columbia and may have contributed to control of HLM in some instances (Clausen 1978).

AHLM - See NHLM.

D. LLM - Maintain health and vigor of trees. Rake ground under trees to remove litter and reduce the overwintering habitat of the adult beetles. Encourage parasites and predators.

E. LNM - Use good silvicultural practices to maintain good stand vigor. Be alert for refugial populations of LNM in protected watersheds which can serve as a source of reinfestation during outbreaks. Predators and parasites have little effect on LNM populations during outbreaks, but may help to keep populations in check in nonoutbreak years.

4. Management Alternatives - Chemical:

A. BLM - Carbaryl and malathion are recommended as ground sprays. In addition, carbaryl is used as a foliage spray. Apply chemicals when thresholds have been exceeded.

BLS - None are registered.

B. BXM - Diazinon and malathion may be applied against the young larvae, but are ineffective against older larvae, or may be used against the adults when emergence is at its peak.

C. NHLM, HLM, AHLM - Diazinon may be applied when larval damage is evident. Do not spray adults, as this will kill the parasites which are present at the same time (J. A. Davidson, U. Md. Coop. Ext. Ser., pers. com.).

D. LLM - None are registered.

E. LNM - At present, no chemical or nonchemical controls are recommended for LNM (T. Hofacker, USFS, pers. comm.).

Refer to Hamel (1983) and Schwartz (1982) for details of formulation and application rates. Contact your regional IPM coordinator to determine which, if any, pesticide is best-suited for your leaf miner management problems.

5. Summary of Management Recommendations:

A. BLM - Use sticky traps or nets to monitor adults beginning in mid-May. Examine 50-100 leaves for eggs and larvae. Calculate the infestation index.

Maintain trees in healthy, vigorous condition. Prune water sprouts. If infestation class is Moderate to Heavy, till ground under tree to depth of 3-4 inches to kill pupae in ground; encourage parasites and predators; consider planting resistant species.

If infestation class is Heavy, consider use of carbaryl, or other registered pesticides, to be applied at the peak of adult activity.

BLS - Use sticky traps or nets to monitor adults beginning in mid-May. Examine 100 leaves for eggs, larvae, or hibernacula. Calculate the infestation index.

Maintain trees in healthy, vigorous condition. Rake and destroy fallen leaves. If infestation class is Moderate or Heavy, consider release of parasites. Consult with your local IPM coordinator before releasing any parasites.

B. BXM - Monitor adult emergence beginning in mid-April. Monitor larvae by examining 50 leaves for presence of mines or insects. Calculate infestation index.

Prune infested plants. Consider use of English varieties that are more resistant than American varieties.

Consider use of malathion or other registered pesticides.

C. NHLM - Monitor for adults in spring with sticky traps. Monitor larval activity by examining 50 leaves. Calculate infestation index.

Remove infested leaves if not too numerous. Prune heavily infested plants. Plant new holly in partial shade. Encourage parasites.

Consider use of diazinon or other registered pesticide against larvae.

HLM - See NHLM. Consider release of parasites.

AHLM - See NHLM.

D. LLM - Examine 50 leaves for presence of mines. Calculate infestation index.

Maintain health and vigor of trees. Rake under trees to reduce overwintering habitat of adults. Encourage parasites and predators.

E. LNM - Use sequential sampling strategy to determine infestation level. Scout out refugial populations.

Use good silvicultural practices to maintain stand vigor and reduce susceptibility to attack.

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V. PERSONAL COMMUNICATIONS

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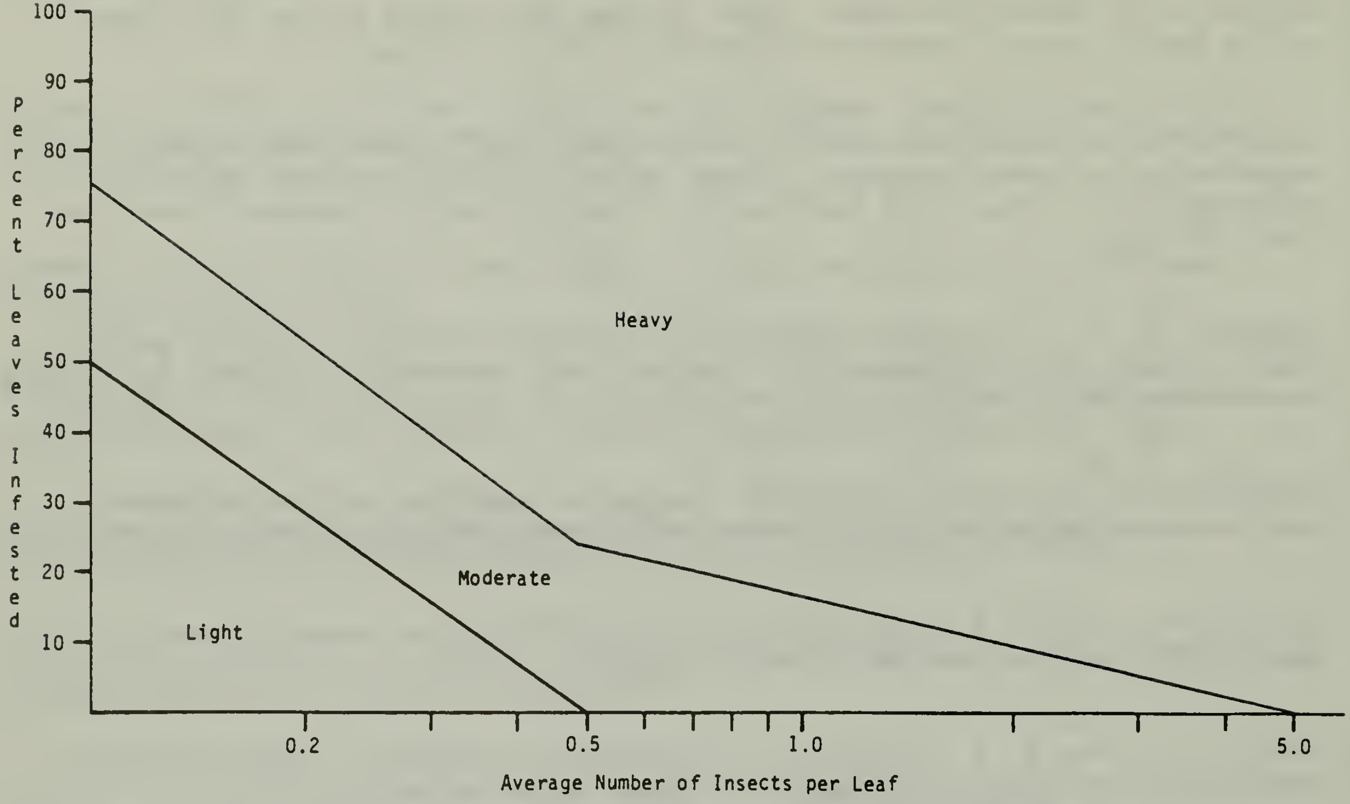
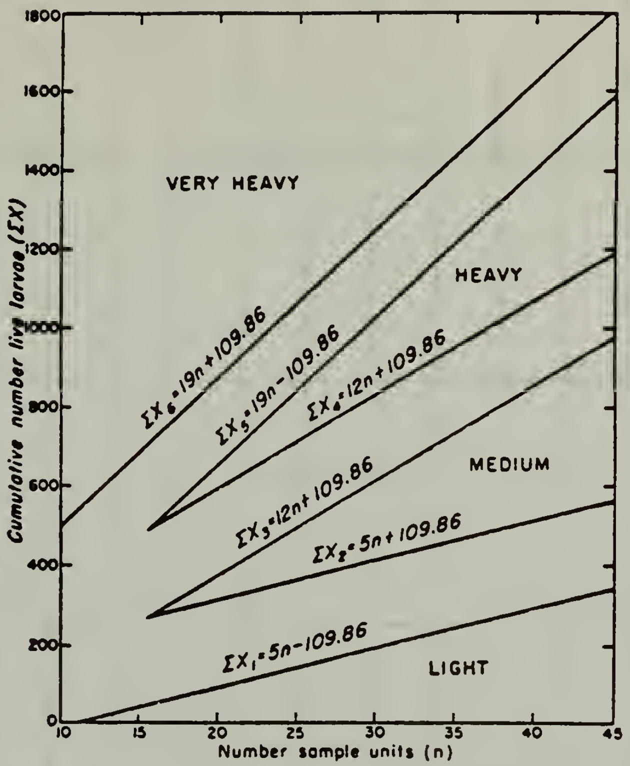
Sample Leaf Miner Infestation Index Graph



This graph was developed by setting tentative aesthetic standards for ornamental plants, based on the subjective opinions of the authors. Three levels of infestation were established; Light, Moderate, and Heavy. The index is designed to take into account the fact that leaves may have more than one larva each. To use, randomly remove 50 to 100 leaves from a plant, and count the number of leaves having live, unparasitized eggs, larvae, and/or pupae. Determine the percent leaves infested by dividing the number of leaves infested by the total number of leaves sampled, and determine the average number of insects per leaf by dividing the total number of insects counted by the total number of leaves sampled. Find the intersection of these two values on the graph and determine the infestation class. If the infestation class is Light, no action is required. If the infestation class is Moderate, nonchemical methods of control should be considered. If the infestation class is Heavy, chemical controls may be necessary.

The values used here are preliminary, and should only be used as guidelines to help establish more precise class limits appropriate to each pest and each park's particular ornamental leaf miner problems. Accurate records of levels of infestation, effectiveness of parasites, visitor awareness of leaf miner activity, efficacy of chemical and nonchemical control methods, ets., are essential to refining these preliminary values.

Sequential sampling lines for LNM (from Stevens and Stark 1962).



The sample unit used is a 2- to 5-internode tip taken from midcrown. All live insects are counted, and the total for each sample unit is added to the total of all previous sample units. Sampling continues until the cumulative number of live larvae (or pupae) falls within one of the four infestation classes. See Pages XIX-20 to XIX-21 and Stevens and Stark (1962) for more details.

SAMPLE LEAF MINER MONITORING FORM.

PARK: ____________________
MONITOR: ____________________
PLANT SPECIES: ____________________

PLANT MAP NUMBER: ____________________(MAP AND COMMENTS ON BACK)

ADULT MONITORING- STICKY BOARD			
DATE	#ADULTS	#PARASITES	INFESTATION CLASS

LARVAL/PUPAL MONITORING - LEAF MINE COUNT					
DATE	#PUPAE	#LARVAE	#LARVAE/PUPAE PARASITIZED	INFESTATION CLASS	AVERAGE # MINES/LEAF

NATIONAL PARK SERVICE
IPM Information Package

LEAFY SPURGE

Final Report

30 November 1984

Submitted To:

Mr. Gary H. Johnston
National Park Service, USDI
Washington, D.C. 20240

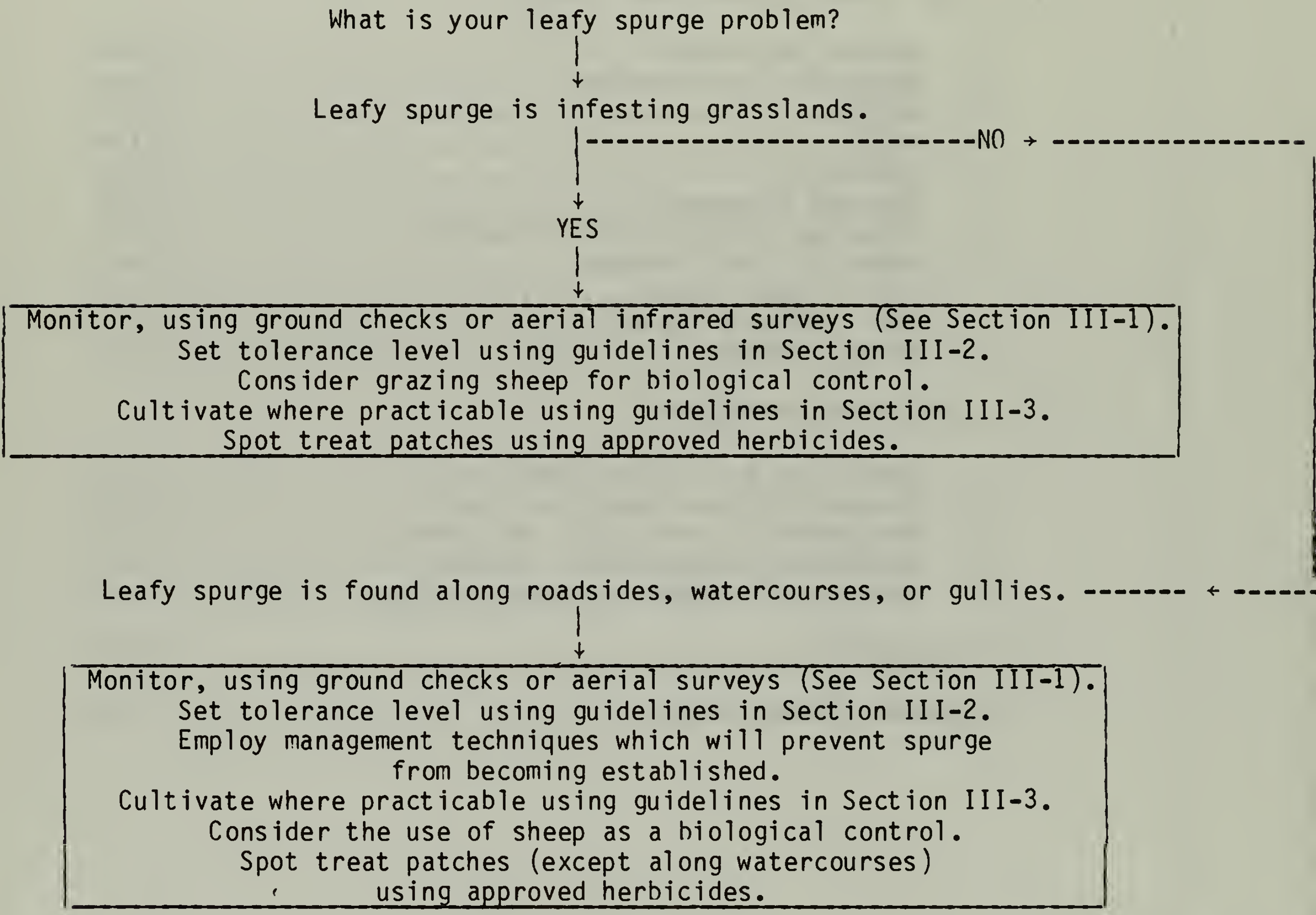
Submitted By:

Dynamac Corporation
11140 Rockville Pike
Rockville, Maryland 20852

CONTENTS

I. LEAFY SPURGE IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



What is your leafy spurge problem?

Leafy spurge is infesting grasslands.

Monitor, using ground checks or aerial infrared surveys (See Section III-1).
Set tolerance level using guidelines in Section III-2.
Consider grazing sheep for biological control.
Cultivate where practicable using guidelines in Section III-3.
Spot treat patches using approved herbicides.

Leafy spurge is found along roadsides, watercourses, or gullies.

Monitor, using ground checks or aerial surveys (See Section III-1).
Set tolerance level using guidelines in Section III-2.
Employ management techniques which will prevent spurge from becoming established.
Cultivate where practicable using guidelines in Section III-3.
Consider the use of sheep as a biological control.
Spot treat patches (except along watercourses) using approved herbicides.

II. LEAFY SPURGE BIOLOGY AND ECOLOGY

1. Species DEscribed:

Leafy Spurge (Euphorbia esula L. of most authors) is a member of the family Euphorbiaceae. It is an herbaceous, deep-rooted perennial broadleaf plant (dicot).

Stems are 16-32 inches tall, unbranched except for flowering heads (umbels). Auxiliary branches may develop when the stem tip is injured. The stem is woody at the base. It is pale green in summer, yellow to red in fall.

Roots commonly extend 12-15 feet into the soil, and may extend into the soil for 30 feet (R. Lorenz, personal communication). Roots are covered with dormant buds, each capable of sprouting and regenerating an entire new stem from almost any depth in the soil when conditions permit. Roots and buds may remain dormant in the soil for 10 or more years (R. Lorenz personal communication).

Leaves are alternate, linear-lanceolate to ovate. They are broader above the middle, tapering to the base. Leaves are sessile (attached directly to the main stem), with entire (smooth-edged) or slightly serrate (saw-toothed) margins. They are blue-green and weakly veined, except at the midrib.

Flowers are borne on an umbel (a flowering head, similar to the flowers of the carrot) at the tip of the stem or on lateral branches near the top of the stem. Flowers are enclosed by prominent yellowish-green bracts (modified leaves which function as petals) forming clusters at the umbel.

Seeds are silvery gray, tinged with purple. The narrow end has a prominent yellow caruncle (a swelling near the scar formed where the seed was attached to the pod) with a longitudinal brown line running through the caruncle to the opposite end of the seed.

See Messersmith (1983) and Eberlein et al. (1982) for complete descriptions and photographs of leafy spurge.

Some controversy exists as to the exact taxonomy of leafy spurge. A wide range of biotypes and phenotypes exists in North America. Over 14 taxonomic names have been given to North American leafy spurge. This wide range of types helps to explain the discrepancies in the literature as to the effect of control measures. The extreme plasticity and genetic diversity of this plant has led several researchers to believe that the leafy spurge of North America is a hybrid between 2 or more Old World spurges including *E. esula*, *E. virgata*, *E. cyparissias*, and *E. uralensis* as well as crosses between hybrids (Schaeffer and Gerhardt, 1984). The difficulty of establishing natural enemies collected from *E. esula* in the Old World tends to support this theory (Harris, 1979). Harris (1979) states that in the *Flora of the USSR*, Canadian leafy spurge specimens key out as *E. virgata*, not *E. esula*.

2. Geographic Distribution:

Leafy spurge was introduced into eastern North America from the Old World in the early 19th century. Other introductions were made in the midwestern U.S. in the late 19th century, probably as contaminants in seed grain (Messersmith and Lym, 1983a). Many reintroductions and crosses have occured in other areas and at other times, resulting in a highly diverse and complex population throughout North America (Lorenz and Messersmith, 1981). Leafy spurge infests over 1.2 million hectares of grassland in the U.S. (Sun, 1981).

The Caucasus region of the USSR is the center of distribution of leafy spurge. It occurs throughout the Eurasian continent from Norway, England, and Portugal through Asia Minor; Turkey, Iran, Afghanistan, and Pakistan. It occurs as far north as Siberia, and as far east as China (Noble et al., 1979). In Eurasia, leafy spurge is an uncommon weed of waste places due to control by over 100 species of natural enemies (R. Lorenz, personal communication).

Leafy spurge is commonly found in Minnesota, North Dakota, South Dakota, Wyoming, Nebraska, Montana, Idaho, and parts of northern Colorado, Nevada, and Utah. It is uncommonly found in scattered locations in other states, and has been reported as far south as Arizona, Delaware, and West Virginia (Noble et al., 1979). It is also common in Saskatchewan, Manitoba, and Alberta, Canada. See Messersmith and Lym (1983a) for detailed range maps.

3. Habitat:

Leafy spurge primarily is a weed of disturbed lands. Pastures and fields left fallow for long periods, tree rows, waste areas, roadsides, and rangelands or open grasslands are all susceptible to infestations by leafy spurge. Leafy spurge is also found commonly along railroad right of ways, water courses, and gullies. It is sometimes found in cultivated lands where infested land has been broken for crop production. It is rare in fields which have been under cultivation for several years, but long-lived roots can regenerate at any time.

Leafy spurge grows on all soils from silty loam bottomlands to bare rock. It can grow on slopes as great as 40° (R. Lorenz, personal communication).

4. Hosts: Not applicable.

5. Life Cycle: Germination from overwintered seed is in early May. True leaves appear 6-10 days after germination (the first pair of true leaves are opposite, later, all leaves are alternate). Stem elongation and vegetative growth occur in mid-May.

Yellow bracts form in late May, with maximum display from early to mid-June.

Flower development is through mid-June, with pollen formed within 48 hours of development of each flower. Leafy spurge is pollinated by insects (Batra, 1983). The first fully developed seeds occur in early July. Seeds are borne in groups of 3 within each pod.

Seed dispersal is in mid-July, during hot, dry weather. Pods burst violently (explosively dehisce) much in the same manner as do the pods of jewel weed, scattering seeds up to 5 m (15 ft.) away from the parent plant.

Leaf loss and late summer dormancy occur during late July to mid-September. Plants releaf in mid-September with the advent of cooler weather. Several leafy branches are formed off the main stem which remains leafless. During this period, photosynthesis resumes, and additional photosynthates are transported to the root system for storage through spring.

Leafy spurge produces vegetative stems from existing roots in late April, making leafy spurge one of the first plants to emerge in spring. Early and rapid growth gives leafy spurge a competitive advantage over most crop and pasture plants.

The root system is extensive, and consists of numerous coarse and fine roots which occupy a large volume of soil. Roots are most abundant in the upper foot of soil, but some roots can extend to a depth of 30 feet. The roots are woody and tough in structure with numerous buds capable of producing new shoots. Roots may be as large as 1/2 inch in diameter in the upper foot of soil, decreasing in size with increasing depth.

The root system contains a large nutrient reserve capable of sustaining the plant for years. Root fragments as small as 1/2 inch long can give rise to new plants. Leafy spurge can withstand repeated mowing and cultivation (Eberlein et al., 1982), due to its well developed food storage system in the roots. Roots have the ability to regenerate plants from almost any depth (R. Lorenz, personal communication).

6. Seasonal Abundance: Leafy spurge usually forms patches which may reach a density of over 200 stems per square yard in sandy soils and higher in heavy clay soils. Patches of leafy spurge usually spread vegetatively with allelopathic chemicals secreted by the root to reduce competition from 1-3 feet per year and form dense patches which crowd out other plants (Eberein et al., 1982). Plants emerge in April (from root stocks) or May (from seed) and persist throughout the growing season. Patches also expand by seed, particularly on the periphery.

7. Responses to Environmental Factors: Leafy spurge, like all weeds capable of colonizing new areas, possesses a great tolerance for soil disturbance and partial defoliation. Seeds may remain viable in the soil for several years until conditions favor germination. Roots are capable of regeneration for many years if the leaves and stem are continuously destroyed. Leafy spurge sprouts earlier than most of the species it displaces, and can grow under a wide range of conditions.

8. Impact of Leafy Spurge

8.1 Direct Impact:

The single greatest direct impact of leafy spurge is the reduction of populations of native grasses and legumes and associated ecosystem changes caused by the superior competitive abilities (rapid growth, and allelopathic properties) of this species (Steenhagen and Zimdahl, 1979).

8.2 Indirect Impact:

Indirect impacts of leafy spurge infestations include the loss of food sources for grazing animals caused by competition with native plants in pastures and on rangeland. Spurge infestations may cut pasture production by 50-75%. Since wildlife and cattle generally avoid grazing in infested areas, carrying capacity may be reduced by up to 75% (Lacey et al., 1984). Leafy spurge is toxic to most grazing mammals, and the milky latex may cause dermatitis if ingested in small quantities by cattle or wildlife. The latex contains esters of cocarcinogenic diterpene irritants and a related antileukemic diterpenoid diester (Batra, 1984).

A second indirect impact of leafy spurge is the cost of attemped control; in some cases, the cost of control may exeed the original cost of the land (Lavigne, 1984). Due to the extremely deep and hardy root system, leafy spurge control in uncultivated areas is costly and control measures must take place continuously over several years. Leafy spurge often regenerates when controls are eased (R. Lorenz, personal communication).

9. Natural Enemies:

No native species of herbivore is known to feed exclusively on leafy spurge. Sheep may graze on leafy spurge without ill effects (Landgraf et al., 1984). Dried spurge may be eaten in hay by stock without ill effects (Messersmith, 1982).

Natural enemies of *E. esula* in Europe and Asia have been introduced in the U.S. and Canada with somewhat inconclusive results. It is thought that hybridization with other introduced spurge species and other factors have changed the genotype of the North American spurge so that most natural enemies from its area of origin have had to date, inconclusive results for leafy spurge control.

III. LEAFY SPURGE MANAGEMENT

1. **Population Monitoring Techniques:**

While leafy spurge is present throughout the growing season, it is most conspicuous when the yellow-green flower-like bracts are open in late May to mid-June.

Leafy spurge usually occurs in patches. To monitor, count or estimate the number of patches per unit area (acre, hectare, etc.). Calculate the average patch size, and count the number of plants per square yard or meter in sample patches. This will give a fairly accurate estimate of the number of plants per area. Careful records should be kept in order to build up a profile of infestation patterns, rates, and treatments.

Leafy spurge can be monitored by aerial infared imagery using the following:

Film: Kodak 1443 color infared (for mapping purposes, use large format 9x9 2443 film)

Filter: Yellow #12

Film Scale: 1:24,000 or larger

Date: 2nd week of June - 2nd week of July

Phenology: Leafy spurge should be in full "bloom" (bract display) and growing vigorously.

The image on false color infared film will be hot pink which is characteristic of leafy spurge at full bloom and not easily confused with any other plant. Patches as small as 10x10 feet (100 sq. ft.) are easily identified using this method. See Armstrong (1979) for futher details.

2. **Threshold/ Action Population Levels:**

Economic thresholds for leafy spurge have not yet been developed. While it is known that heavy infestations can lower range productivity, the cost of mechanical and chemical controls are often considered to be uneconomical in most of the affected areas (Sun, 1981). Most ranchers consider spurge to be below injury level (i.e. "under control") if spurge patches do not expand from year to year (R. Lorenz, personal communication). Thresholds will differ at different sites; heavily visited park lands such as historical or developed sites will have a lower tolerance than will natural areas or grasslands.

In natural areas within the park, leafy spurge management should begin when an infestation is discovered. In areas such as historic or developed sites, or where park lands are adjacent to private or public grazing lands, management techniques should be employed to prevent spurge infestations, and established patches should be controlled to prevent spreading.

3. Management Alternatives-Nonchemical:

A. Biological control - Leafy spurge is attacked in North America by only a few generalist (polyphagus) native herbivorous insects (Harris, 1979).

1. Insects -

Several species of insect herbivores have been screened and/or introduced into North America as possible biological control agents. No single species is likely to achieve complete control throughout the range of leafy spurge, but several species may complement each other to reduce spurge population levels.

The spurge hawk moth, Hyles euphorbiae (L.), (family Sphingidae), was introduced into Canada in 1977 (Forwood and McCarty, 1980). Populations stabilized at densities considered too low to provide effective control, however, and eventually declined to extinction. Subsequent introductions in Montana and New York have become established and introductions are now planned for other states (L. Andres, personal communication). The spurge hawk moth has one generation per year; caterpillars defoliate plants once and go into diapause. Leafy spurge foliage regenerates in most instances (S. Batra, personal communication).

The moth Chamaesphecia tenthrediniformis (Denis & Schiff) was released in Canada in 1970 after promising results in feeding tests. However, all larvae released in the field died without feeding on leafy spurge. In a similar release in Australia, it was observed that the larvae of this moth fed on E. esula, but not on E. virgata (Harris, 1979). A second introduction, using another strain imported from eastern Europe, and which is highly specific to some varieties of leafy spurge thought to be present in North America, is planned for 1985 (L. Andres, personal communication).

Negative results were obtained in Canada following the release of the aphid *Acrythosiphum neerlandicum* which is only known from *E. esula* in Europe. Individuals of this species failed to develop and died on Canadian leafy spurge (Harris, 1979). Two other aphids, *Acyrthosiphon cyparissiae* (Koch) and *Aphis euphorbiae* (Kltb.), are currently under quarantine in Canada. *A. cyparissiae* feeds on leaves of leafy and cypress spurge; *A. euphorbiae* feeds mainly on stems (McClay and Harris, 1984).

The root-boring cerambycid beetle *Oberea erythrocephala* (Schrank), which attacks both *E. esula* and *E. virgata*, is undergoing testing in Canada and holds much promise as a possible biological control agent. This species was released in 1980 and 1982 in Wyoming using stock imported from southern Europe, but failed to establish (Harris, 1979). A second release, using new material from eastern Europe collected from a different form of spurge, was attempted in Montana in 1983. Individuals from this release established and were recovered in 1984 (L. Andres, personal communication).

The cecidomyiid gall midge *Bayeria capitigena*, which forms galls over the branch tips, slowing growth, stunting the plant, and preventing blossoming, has been evaluated and should be available for release in 1985 (L. Andres, personal communication). This species has several generations per year, making it an excellent potential biological control agent.

The flea beetle *Aphthona flava* Guill. will be available for release in 1985. The adults feed on the leaves of leafy spurge, causing minor damage, but larvae feed heavily upon the roots, stunting and eventually killing the plant. There is one generation per year (L. Andres, personal communication). This species has been successfully overwintered in Canada (McClay and Harris, 1984).

Lobesia euphorbiana, a tortricid moth which feeds within and kills the shoot tips of its host plants, is undergoing studies in quarantine in Albany, Ca. It is currently believed that the host range of this species is too broad to recommend its release in the U.S. (Pemberton, 1984).

2. Grazing by sheep -

Although grazing by livestock has not been recommended in the past, Landgraf et al. (1984) have found that sheep may graze on leafy spurge without ill effects. The diet of sheep can contain up to 50% leafy spurge with no significant difference in weight gain compared to sheep feeding in spurge-free pastures. They conclude that sheep are a viable biological control agent for leafy spurge. Pastures grazed by sheep from May to September for 5 successive seasons show up to 98% reduction in spurge populations. Utilization of and effects of leafy spurge on lambs and lactating ewes has not yet been quantified.

Grazing by sheep may not be an appropriate control measure in natural areas. Some varieties of spurge may be rejected by sheep, and in most cases spurge will regenerate the season after grazing pressure ceases (R. Lorenz, personal communication).

If sheep are to be used as a biological control for leafy spurge, the following guidelines from Lacey et al. (1984) should be followed:

1. Grazing should begin in the spring when spurge plants are only a few inches tall.

2. Schedule sheep grazing rotations so that spurge does not go to seed.

3. If sheep graze after seed set, animals should be held for 5 days to allow viable seeds to be passed before sheep are moved to new pastures.

4. Sheep grazing can be combined with herbicide use around the fringes of patches for optimal control.

3. Pathogens -

Several plant pathogens including rust fungi, powdery mildews, soil borne fungi, and foliar pathogens have been tested. To date, none have been found to be desirable control agents due to wide host ranges (which include domestic crops) or lack of permant control. Several rusts and Alternaria species have been collected recently in Europe and are undergoing testing at this time (Littlefield, 1984).

B. Mechanical control - Use of controlled burning has been attempted in North Dakota and in Wyoming. Although burning has little effect on established plants with deep root systems, fire may be highly effective in reducing seed and seedling viability. Controlled burns in the fall against the wind (burning against the wind results in more complete combustion and hotter fires), resulted in germination rates 90% lower than in unburned plots (A. Bjugstad, personal communication). Further tests using fire as a management tool, including spring burning to destroy seedlings, are planned for 1985.

Mowing, especially when used prior to treatments with herbicides, may allow reduced rates of chemicals to provide effective shoot control (Ferrell and Alley, 1984b).

Hand pulling of leafy spurge while in the bloom stage results in reduced regrowth vigor for 2 years. Pulling also damages the root, increasing the chance of infection by pathogenic organisms (Maxwell et al., 1984).

Intensive cultivation at 2-3 week intervals will reduce leafy spurge stands by 90% in the first year, and give complete control in 2 years. Similar results have been achieved by cultivation with a duckfoot cultivator every 2-3 weeks or a springtooth harrow each week (Derscheid, 1979).

C. Cultural control - In areas where planting of competitive crops is possible, crops such as sudangrass or buckwheat may be utilized. Competitive cropping reduced leafy spurge stands by 50% in the first year of trials, and 80% in the second year when given 3 cultivations before seeding, and with stubble plowed after harvest (Derscheid, 1979).

Elimination of leafy spurge was also achieved in 2 years following planting of close-drilled forage sorghum or soybeans. A short season of intensive cultivation, followed by planting of fall seeded crops of bromegrass, reduced leafy spurge populations by 95% (Derscheid, 1979).

Seeding of spring-seeded grains or alfalfa is not recommended due to the superior competitive ability of leafy spurge, which emerges earlier in the season and which has allelopathic (i.e. toxic to other plants) properties.

Reinfestation of leafy spurge from seed can be prevented by using soil-building crop rotations. Legumes (such as sweetclover) will prevent establishment by most leafy spurge seedlings (Derscheid, 1979). Mechanical and cultural controls may not be suitable for use in natural areas, and such controls must be continuous to prevent regeneration from roots (R. Lorenz, personal communication).

Consult with your regional IPM coordinator before employing such methods.

4. Management Alternatives-Chemical:

Herbicides, timed for optimum control based on plant development, give excellent control of leafy spurge. Picloram (Tordon®) and 2,4-D are considered to be among the most effective herbicides for leafy spurge control. Control is most effective when applied during flower and seed development or during fall regrowth before the first killing frost. Picloram is considered to be the most effective herbicide when applied with a roller applicator. The best times for application are considered to be from mid-June until just before seed dispersal in July. The early part of this period is best to control established plants and to prevent seed development. Applications of herbicides later than July do not prevent seed dispersal. Viable seeds remain in the soil for several years following picloram treatments (Bowes and Thomas, 1978). Control effectiveness declines with low soil moisture and unseasonably high or low temperatures. Roots on established plants are killed to a depth of about 18 inches, and reapplications must be made every 3 years (Lacey et al., 1984). Picloram may be used on young plants to achieve complete eradication (due to less extensive root systems). Picloram treatments are not recommended for control of leafy spurge among trees due to the long residual effects in soil of this herbicide, which is toxic to trees (Lym and Messersmith, 1983). Yearly applications of 2,4-D will control leafy topgrowth of leafy spurge growing among trees.

Glyphosate is effective after the seeds have filled their pods in mid-summer or during fall regrowth. Glyphosate gives less long-term control when used on spring growth. Glyphosate treatments should be followed by treatments of picloram or picloram - 2,4-D mixtures the following spring. Glyphosate applied during the fall period of regrowth until the first frost, and followed by spring applications of 2,4-D, is also recommended for spurge control among trees. Care should be taken not to expose tree foliage to glyphosate.

Small infestations should be controlled at once with picloram or Banvel® (to avoid spreading).

These herbicides give 90%-99% control in the first year. Treatments should be followed up for several years because topgrowth only is killed and roots will continue to regenerate. See Alley (1979) and Lym and Messersmith (1983) for further details.

Applications of herbicides must be made on a yearly basis due to the rapid regenerative ability of spurge and the poor translocation of herbicides within the plant (R. Lorenz, personal communication).

Use of plant growth regulators, alone or in combination with herbicides, does not significantly reduce leafy spurge shoot growth or root growth when compared to the effects of herbicides used alone (Ferrell and Alley, 1984a).

Consult your regional IPM coordinator to determine which, if any, herbicide is best suited to your IPM program.

5. Summary of Management Recommendations:

1. Monitor leafy spurge by ground checks or aerial surveys using false color infrared film.
2. Determine injury levels based on land usage (local weed ordinances should be acknowleged).
3. Use cultural or mechanical controls to reduce small to medium infestations. Consider the use of controlled grazing by sheep as a biological control.
4. Use registered herbicides where appropriate; applications should be timed for best control, and follow-up treatments should be applied when necessary.

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NATIONAL PARK SERVICE
IPM Information Package

MUSEUM PESTS:
DERMESTID BEETLES;
CLOTHES MOTH;
CASE MAKING MOTH

Final Report

5 January 1985

Submitted To:

Gary Johnston
U.S. National Park Service
Washington, D.C. 20240

Submitted By:

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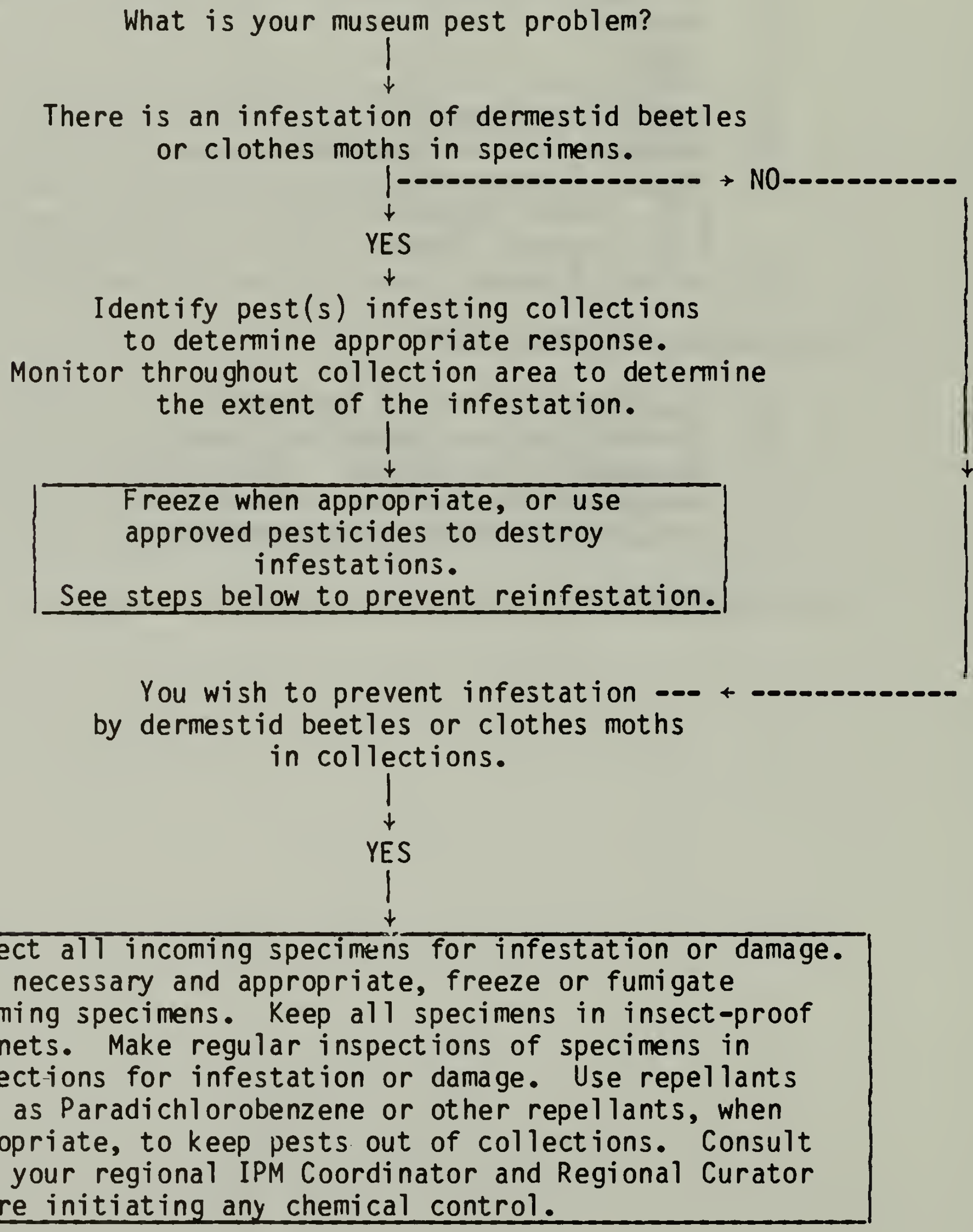
CONTENTS

I. MUSEUM PEST IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All uses of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



What is your museum pest problem?

There is an infestation of dermestid beetles or clothes moths in specimens.

NO

YES

Identify pest(s) infesting collections to determine appropriate response. Monitor throughout collection area to determine the extent of the infestation.

Freeze when appropriate, or use approved pesticides to destroy infestations. See steps below to prevent reinfestation.

You wish to prevent infestation by dermestid beetles or clothes moths in collections.

YES

Inspect all incoming specimens for infestation or damage. When necessary and appropriate, freeze or fumigate incoming specimens. Keep all specimens in insect-proof cabinets. Make regular inspections of specimens in collections for infestation or damage. Use repellants such as Paradichlorobenzene or other repellants, when appropriate, to keep pests out of collections. Consult with your regional IPM Coordinator and Regional Curator before initiating any chemical control.

II. MUSEUM PEST BIOLOGY AND ECOLOGY

1. Species Described: Major pests of proteinaceous museum specimens are members of the beetle family Dermestidae (carpet beetles or dermestids), and moths in the family Tineidae (clothes moths). Three species of beetle and two species of moth are the most common museum pests, cause the most damage to specimens, and are the most difficult to control (Ebeling, 1975).

1. Black Carpet Beetle - *Attagenus unicolor* Brahm. Adults are 2.8 - 5 mm long, dark brown or black in color. Larvae are 7-8 mm long and narrow. They are dark brown to golden in color, with short bristles covering the body, and a "tail" of long bristles.

2. Common Carpet Beetle - *Anthrenus scrophulariae* (L.). Adults are approximately 5 mm in length, oval in shape, blackish with white scales, and a longitudinal stripe of orange and red scales down the middle of the back. Larvae are red to brown with black to brown hairs. They are 2.5-3.5 mm in length.

3. Varied Carpet Beetle - *Anthrenus verbasci* (L.). Adults are 2-3 mm in length, blackish with irregular white, brown, and yellow scales in a variety of patterns. Larvae are 4-5 mm in length, with tufts of bristles on each segment, and a "tail" of long bristles.

4. Webbing Clothes Moth - *Tineola bisselliella* (Hummel). Adults are golden yellow with a tuft of bronze colored hairs on the head, and a wingspan of 8-10 mm in length (females larger than males). Larvae are whitish, 8-10 mm in length.

5. Casemaking Clothes Moth - *Tinea pellionella* L. Adults are brownish with 3 dark spots on each front wing, 7-9 mm in length. Larvae are white, 7-8 mm in length.

See Mallis (1982), Beal (1970), Ebeling (1975), and Edwards, Bell, and King (1981), for illustrations, descriptions, and keys to these and other museum pest species.

2. Geographic Distribution:

1. Black Carpet Beetle - Worldwide distribution. Common throughout the U.S. and Canada, this species is the most important dermestid species in the states east of the Rockies.

2. Common Carpet Beetle - Worldwide distribution. Common throughout the U.S. and Canada, this species is the most common dermestid species in Rocky Mountain, northern tier, and mid-western states (Ebeling, 1975).

3. Varied Carpet Beetle - Worldwide distribution. Found throughout the U.S. and Canada, this species is the most important dermestid species in the Pacific Northwest and California (Ebeling, 1975).

4. Webbing Clothes Moth - Worldwide distribution. This species is the most common moth pest in the U.S.

5. Casemaking Clothes Moth - Worldwide distribution. This species is less common in northern U.S. than webbing clothes moth; more common in the southern U.S. (Ebeling, 1975).

3. Habitat:

1. Black Carpet Beetle - Commonly found in bird nests outdoors. Indoors, found near windows and in or near larval food sources (see Section 4).

2. Common Carpet Beetle - Outdoors; found in nests of small mammals. Indoors; in or near larval food sources (see Section 4).

3. Varied Carpet Beetle - Outdoors; commonly found in wasp nests and bee hives, including honey comb (Ebeling, 1975). Indoors, in or near larval food sources (see Section 4).

4. Webbing Clothes Moth - Indoors only in U.S. Found in or near food sources (see Section 4).

5. Casemaking Clothes Moth - Indoors in northern U.S., may be outdoors in summer in South. An infestation due to moth larvae in owl pellets deposited in a church steeple has been reported (Mallis, 1982).

4. <u>Hosts</u>: The larvae of dermestids and clothes moths are the destructive stage. They are among the few animals which can digest keratin, and keratin containing substances such as wool, fur, and feathers are preferred food materials. Larvae attack other fibers, especially if the fabrics are contaminated with urine, perspiration, beer, milk, or fruit juices. Adults feed on nectar and pollen, or do not feed at all.

1. Black Carpet Beetle - Outdoors, adults feed on pollen and nectar. Indoors, larvae have been observed to feed on the following: woolens, including clothes and rugs; silk fabrics; carpets; felts; fur; skins; yarn; velvet; feathers; hair-filled cushions; meats; leather; museum specimens (including insect collections); spices; seeds; grains; and cereals.

2. Common Carpet Beetle - Outdoors, adults feed on pollen and nectar. Indoors, larvae feed on the following: fabrics; woolens; feathers; leather; fur; silk fabrics; mounted animal and pinned insect specimens in museums; and pressed herbarium specimens.

3. Varied Carpet Beetle - Outdoors, adults feed on pollen and nectar. Indoors, the larvae have been observed to feed on the following: woolens; skins and leather; fur; mounted museum specimens (especially insect specimens); feathers; horn; baleen, bone; hair; silk; plant material; and spices such as Cayenne pepper. Outdoors, this species is commonly found in wasp nests where the larvae feed on dead insects and other wastes (Ebeling, 1975).

4. Webbing Clothes Moth - Larvae feed on hair, feathers, fur, wool, upholstered furniture, piano felts, natural bristles, and lint. Adults are not believed to feed.

5. Casemaking Clothes Moth - Larvae feed on hair, hides, wool, feathers, and some plant material such as stored tobacco, herbarium specimens, drugs, and spices. Adults are not believed to feed.

5. Life Cycles:

1. Black Carpet Beetle - Eggs are laid indoors in lint, trash, or near other food sources. They hatch in 6-11 days at room temperature, longer if temperatures are lower. Larvae go through 5-11 instars under normal conditions, up to 20 if conditions are poor. The larval period takes approximately 260-640 days depending on availability of food, the level of humidity, and temperature. Larvae are repelled by light. Larvae pupate in the skin of the final instar. Pupation takes 6-24 days, longer in cold conditions. The adult may remain in the partially shed pupal skin for up to 3 weeks. Adults live only a short time; females live 30 days or slightly longer, males up to 40 days. Females lay from 40 to 115 eggs. There is 1 generation per year.

2. Common Carpet Beetle - Eggs are small and white, with projections at each end to catch and cling to fibers. Eggs hatch in 10-18 days. Larvae go through 6 instars, taking approximately 11 days per instar. Pupation occurs in the skin of the last instar and lasts about 2 weeks. After emerging, the adult is quiescent in the skin for approximately 3 weeks, then emerges and is active for approximately 30 days. Females lay up to 35 eggs. There is one generation per year outdoors, with overwintering in the pupal form. Indoors, several generations per year may occur.

3. Varied Carpet Beetle - Eggs are oval in shape, up to .55 mm long, changing in color to cream as they mature. Eggs hatch in 17-18 days under normal indoor conditions. Larvae go through 5-16 instars depending on availability of food, levels of humidity, and temperature. The larval period lasts from 1-2 years. Pupation occurs in the skin of the final instar, and lasts 10-13 days. Adult males live 2-4 weeks, females, 2-6 weeks. There is a single generation per year outdoors, but several per year indoors.

4. Webbing Clothes Moth - Eggs are oval, white, 1 mm in size. They are laid singly or in small groups among loose threads in most natural fibers or among hairs in furs. Eggs hatch in 4 days to 3 weeks, depending upon temperature; 4-10 days in summer, longer in winter. Each female averages 40-50 eggs, with some females depositing up to 150.

Larvae are 1 mm long when first hatched, and whitish in color. There are 5-11 instars, depending on temperature and availability and quality of food. The larval stage lasts from 1-29 months. Larvae often spin silken pads (webs) or construct silken feeding tubes on the feeding surface. Larvae are nocturnal. Indoors, pupation lasts 8-10 days in summer (21-28 days in winter in cooler buildings). Adult males live an average of 28 days, females, an average of 16. Life spans are longer at lower temperatures where metabolic processes are slower. Females mate once and begin to oviposit on the same day they emerge from the pupal case. After laying their full compliment of 40-50 eggs, females die. Males mate throughout their adult lives. Males are moderately strong fliers; gravid females walk, but will fly if disturbed. Adults do not fly to light, and will avoid lighted areas.

5. Casemaking Clothes Moth - The life history of the casemaking clothes moth is similar to that of the webbing clothes moth. The larva spins a case of silk and fibers from the food source. The colors of the food source will be represented in the case. This case is carried with the larva throughout its life; the larva will die if removed from the case. The case is 6-9 mm in length, depending on the instar and size of the larva. Larvae graze at random over the food surface; damage is proportional to the time spent in any one area. Larvae are nocturnal. Pupation occurs within the larval case in a protected place.

   There are 2 generations per year in the South, 1 in the North. In the northern U.S., adults are often seen flying between June and August. Adults do not fly to light.

6. Seasonal Abundance:

1. Black Carpet Beetle - Outdoors, most abundant from April to June. Adults are not found after July. Indoors, most abundant from February to July, but can be found at any time under suitable conditions.

2. Common Carpet Beetle - Outdoors, adults are most common in late May to June when they feed on pollen and nectar. Indoors, adults and larvae may be common all year in heated buildings.

3. Varied Carpet Beetle - Outdoors, adults are most common in late spring and early summer when they feed on pollen and nectar. Indoors, adults and immatures may be common all year in heated buildings.

4. Webbing Clothes Moth - Indoors, in heated buildings, webbing clothes moths are active throughout the year.

5. Casemaking Clothes Moth - Indoors, in heated buildings, casemaking clothes moths may be active and breed throughout the year.

6. Response to Environmental Factors: Populations of museum pests are influenced by temperature, humidity, and the availability of food. Humidity, rather than temperature, is thought to be the most critical factor after food. This factor is currently under study.

8. Impact of Museum Pests:

8.1 Direct Impact: Carpet beetles and clothes moths feed on a wide variety of museum specimens, damaging or ruining their scientific, aesthetic, and historical values. The varied carpet beetle may also infest foodstuffs such as cereals and other grain products.

8.2 Indirect Impact: Some species of dermestids may bore through cardboard or paper containers, allowing access by other insect pests.

9. Natural Enemies: Dermestids and clothes moths are preyed upon by other insects, mites, and spiders. Eggs may be destroyed by fungi at high humidities. However, high humidity and fungi create other management problems.

III. MUSEUM PEST MANAGEMENT

1. **Population Monitoring Techniques:**

Specimens on display, as well as all collections, should be monitored on a regular (at least twice a year) basis for dermestids and moths. Use a handlens to examine for eggs if an infestation is suspected. Look for live adults and larvae, and the presence of cast larval skins or sand-like feces which are often the color of the substance being fed upon (dermestids only). Presence of feeding debris around or below specimens is an indication of infestation. Exit holes, feeding holes, hair falling from fur or pelts, mats of fibers under which clothes moth larvae feed, silken feeding tubes, silken larval cases, or moth pupae are all indications of infestation.

Examine window sills on a regular basis as many of these insects fly to the light in search of outdoor flowers and nectar. Larvae may also be found behind baseboards, mouldings, in cracks in floors, behind radiators, or in air ducts. Small sticky boards (3" x 5" cards) randomly placed throughout the facility and/or specimen cases, and routinely examined are useful in detecting early infestations.

Rountine examination and frequent movement of articles, if possible, will also disrupt insect populations and detect infestations.

Damaged materials can be examined under a microscope to determine the species responsible (Pence, 1966).

2. **Threshold/ Action Population Levels:**

Presence of live adults or larvae indicate ongoing infestations which should be treated immediately. Cast larval skins and feeding damage may have resulted from old infestations, but in regularly monitored collections, this should be regarded as an indication of an active infestation. Thus it is vitally important to maintain careful monitoring records.

3. **Management Alternatives - Nonchemical:**

The most effective way to prevent damage from dermestids and clothes moths is to prevent establishment of infestations. All incoming specimens should be examined carefully for damage and live insects,and records kept. Incoming specimens showing signs of infestation may be frozen at -18°C for at least 48 hours before being accessioned (Crisafulli, 1980). Freezing is not recommended for wood, bone, lacquer, painted surfaces, leather, and certain other specimens. Contact your Regional Curator before undertaking any control measures.

All specimens subject to insect damage should be kept in insect-proof cases if possible, and examined on a regular basis.

Lowered humidities and, to lesser extent, lowered temperatures reduce the chance of infestation. Infestations may slow or stop during winter when indoor humidities are their lowest. Under conditions of extreme humidity, dermestid eggs may be attacked by fungi (Mallis, 1982), however humidities high enough to promote fungal growth may be damaging to most specimens. Low humidities may shrink or otherwise damage some specimens.

Windows in areas where specimens are kept should be tightly screened or kept closed at all times to prevent entry by dermestids. Adult dermestids feed on pollen and nectar and cut flowers should be kept out of specimen areas to reduce the chance of accidental infestation.

All air vents and hot air registers should be equiped with filters to trap potential incoming pests. Filters should be changed on a regular basis. All preparation of specimens should take place in areas other than collection rooms.

Vacuum all accesible areas on a regular basis to prevent accumulations of lint, hair, and other carpet beetle and clothes moth food materials.

Research is ongoing concerning the use of B.t. and IGRs for control of museum pests.

4. Management Alternatives - Chemical:

Consult your regional IPM coordinator and Regional Curator to determine which pesticide, if any, is best suited to your IPM program.

Care should be taken when using chemical pesticides, as materials may be hazardous to human health and may damage some specimens.

Paradichlorobenzene and naphthalene are commonly used as repellants in museum cases. These materials do not eliminate infestations, but may be useful in preventing them. Paradichlorobenzene and naphthalene may cause damage to certain plastics (e.g.; bakelite), and may soften and shrink resins, adhesives, and paints. Organic gas filters should be installed on the sides of cabinets to absorb fumes and replaced when the odor is detected in the room.

Dowfume 75 (a mixture of carbon tetrachloride and ethylene dichloride) is used as a fumigant against dermestids and moths. A highly toxic chemical, it should only be used by qualified personnel in fumigation chambers which are inspected often. Manufacture of homemade Dowfume 75 is extremely dangerous and clearly illegal. Dowfume-75 may soften paints and resins.

For emergency use on wooden specimens which are too large to move, vapona strips may be used as a fumigant when placed with the specimen under a plastic tarp for approximately 5 days. It is no longer recommended for use in museums and is listed as unsuitable for this purpose by its manufacturer. It is considered minimally effective for use against insects. Vapona undergoes hydrolysis in the presence of atmospheric moisture (the % relative humidity needed to trigger hydrolysis is unknown), and releases sulfuric acid vapors which soften gums and resins, corrode metals, and weaken cellulose. Vapona strips now being used should be removed to prevent damage to specimens.

Pyrethrum may be used in storage cases as a contact pesticide. The material should not be allowed to contact specimens.

Vikane as a structural fumigant in buildings, under tarps, or in cabinets may be used to destroy infestations. Vikane may be applied only by licensed fumigators. It may contain additives such as chloropicrin to allow detection in the case of leaks. Additives may be damaging to some specimens.

The following insecticides are not yet registered for use in museums, but their use is not inconsistent with the labelling:

Drione powder or aerosol is effective on most insect pests in herbaria for up to 6 months. It is used in cases as a contact insecticide, but should not contact specimens.

Ethylene oxide fumigation has occasionally been used in instances where other methods have failed. Ethylene oxide is not recommended in most cases due to the hazards involved in application of the chemical, with 1 ppm the maximum safe limit (detection at 1 ppm requires equipment not normally available to most museums; the human nose can detect this material at approximately 70 ppm). Ethylene oxide is an active carcinogen. Use of ethylene oxide now requires special fumigaton chambers and other safety precautions. Ethylene oxide adheres to fibers and off-gasses for considerable periods.

5. Summary of Management Recommendations:

1. Inspect collections on a routine basis for signs of infestation and damage. Record all data.
2. Isolate collections from public areas, preparation areas, and from dermestid colonies used in specimen preparation.
3. Inspect all incoming specimens for signs of infestation.
4. All incoming specimens should be quarantined and if appropriate, frozen or fumigated.
5. Where appropriate, use repellents such as paradichlorobenzene to keep dermestids and clothes moths out of collections.
6. If infestation is observed, use of an approved pesticide may be required.
7. Check with Regional IPM Coordinator and Regional Curator prior to initiating any control program.

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NATIONAL PARK SERVICE
IPM Information Package

PRAIRIE DOGS

Final Report

15 April 1985

Submitted To:

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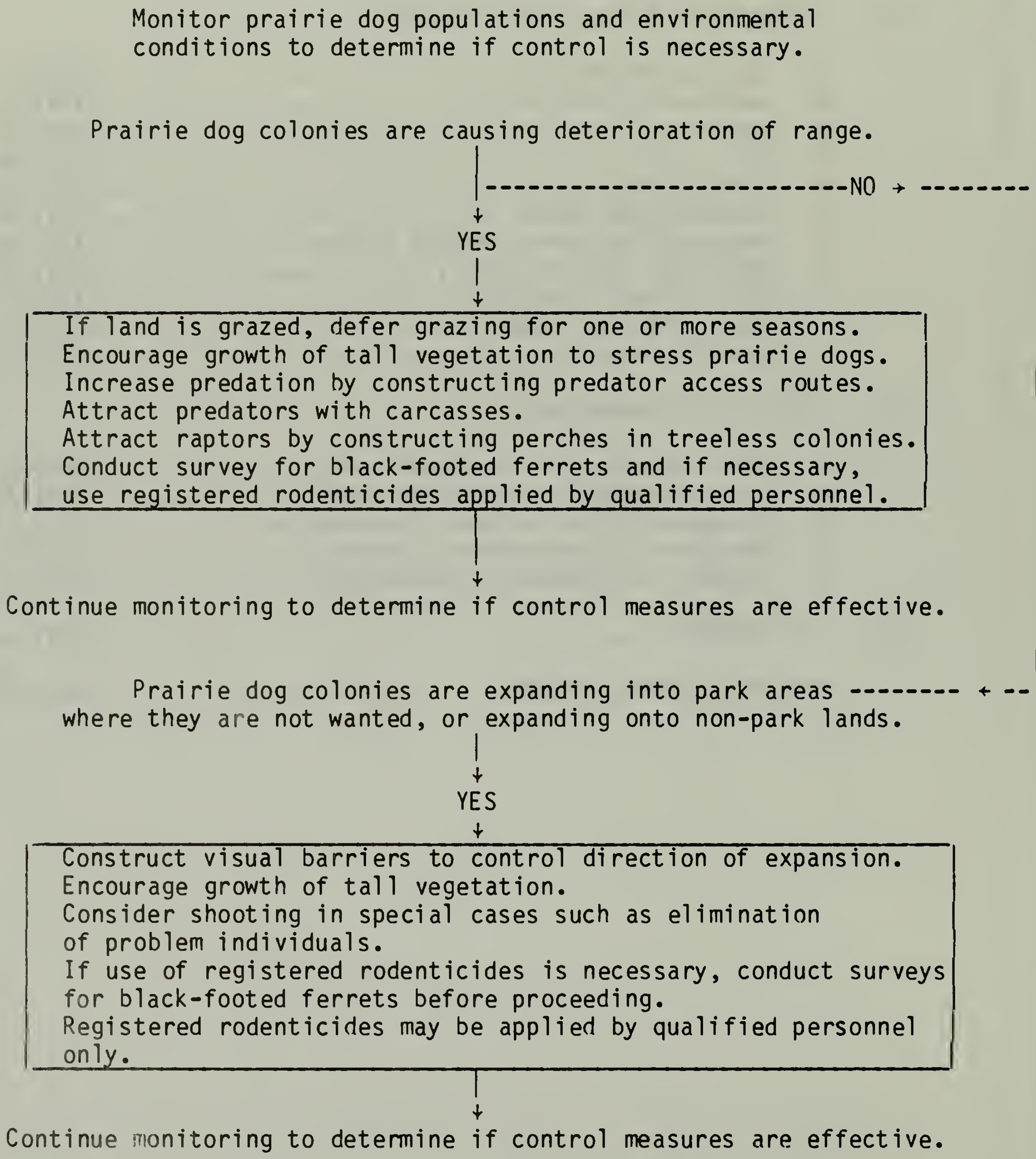
CONTENTS

I. PRAIRIE DOG IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



Monitor prairie dog populations and environmental conditions to determine if control is necessary.

Prairie dog colonies are causing deterioration of range.

If land is grazed, defer grazing for one or more seasons.
Encourage growth of tall vegetation to stress prairie dogs.
Increase predation by constructing predator access routes.
Attract predators with carcasses.
Attract raptors by constructing perches in treeless colonies.
Conduct survey for black-footed ferrets and if necessary, use registered rodenticides applied by qualified personnel.

Continue monitoring to determine if control measures are effective.

Prairie dog colonies are expanding into park areas where they are not wanted, or expanding onto non-park lands.

Construct visual barriers to control direction of expansion.
Encourage growth of tall vegetation.
Consider shooting in special cases such as elimination of problem individuals.
If use of registered rodenticides is necessary, conduct surveys for black-footed ferrets before proceeding.
Registered rodenticides may be applied by qualified personnel only.

Continue monitoring to determine if control measures are effective.

II. PRAIRIE DOG BIOLOGY AND ECOLOGY

1. Species Described:

Prairie dogs are rodents in the family Sciuridae, which includes tree squirrels, ground squirrels, chipmunks, marmots, and woodchucks. All prairie dogs are in the genus *Cynomys*.

Prairie dogs have squat, somewhat stout bodies with short legs and long claws adapted for digging. The fur is coarse with black and buff colored tips. Color ranges from yellowish through sandy to cinnamon. The fur is often stained by the dirt in which they burrow. Belly fur is cream to white in color. The distal third of the tail is black in the blacktailed prairie dog, white in the other species.

The presence of prairie dog colonies is usually evidenced by bare earth mounds 25-75 feet apart. Each mound is approximately 1-2 feet high.

Prairie dogs can be divided into 2 groups, white-tailed and blacktailed. There are 4 generally recognized species of prairie dogs in the United States (the Mexican prairie dog *C. mexicanus*, does not occur in the U.S.), with several subspecies (Costello, 1970).

1. Blacktail prairie dog - *Cynomys ludovicianus*. Total body length (head and body) is approximately 11-13 inches. It is slightly smaller than a house cat in body size, but weighs much less (2-3 lb.). The tail is 3-4 inches in length, with a black tip. There are 8 mammae. This is the species most often considered pestiferous.

   a. Arizona prairie dog - *C. l. arizonensis*. Similar to the blacktail prairie dog, it is not usually considered pestiferous.

2. Whitetail prairie dog - *C. leucurus*. This and the next 3 species are similar. The head and body measure 11-12 inches in length. The tail is 1 1/4 - 2 1/2 inches long; the distal third is white. This species weighs 1 1/2 - 2 1/2 lb. There are 10 mammae. Whitetail prairie dogs are usually not considered pestiferous due to their comparatively low population densities.

3. Utah prairie dog - C. parvidens. Similar to the whitetail prairie dog, but with black spots above the eyes. The fur is uniformly brown or reddish. The distal half of the tail is white. This species is threatened.

4. Gunnison's prairie dog - C. gunnisoni gunnisoni. Similar to the whitetail prairie dog, but the distal half of the tail is white with a gray center.

    a. Zuni prairie dog - C. g. zuniensis. Similar to the Gunnison's prairie dog, but somewhat larger. The fur is pinkish cinnamon, with less buff in the belly fur.

Differences among these species are not apparent to most observers. Burt and Grossenheider (1964) include the whitetail, Gunnison's, Zuni, and Utah prairie dogs under one species, C. gunnisoni, which they refer to as the whitetail prairie dog.

See Burt and Grossenheider (1964) for color illustrations of blacktail and whitetail prairie dogs. See Costello (1970) for black and white photographs of all of the above species.

2. Geographic Distribution:

1. Blacktail prairie dog - Originally found on mixed to short grass prairie from Saskatchewan to west central Texas. The current distribution is from the Rockies to approximately 97°W longitude in central Oklahoma, Kansas, and Nebraska.

    a. Arizona prairie dog - Southeastern Arizona, south and central New Mexico, southwest Texas, and adjacent parts of Sonora and Chihuahua, Mexico. This species is now believed extinct in Arizona.

2. Whitetail prairie dog - Original range in mountainous parts of Montana, Wyoming, Utah, and Colorado, up to 12,500 feet. This species is now thought to be found only in Colorado and Utah.

3. Utah prairie dog - Restricted to Utah, recently found only in 9 counties. A threatened species.

4. Gunnison's prairie dog - Mountainous regions of central and south-central Colorado and New Mexico. Formerly abundant throughout its range, this species has been nearly extirpated by poisoning programs and sylvatic plague.

a. Zuni prairie dog - Formerly widely distributed in southeastern Utah, southwest Colorado, northwest and central New Mexico, and north central Arizona. This species is greatly reduced in numbers and range.

See Burt and Grossenheider (1964) and Boddicker (1983) for detailed range maps.

3. Habitat:

Prairie dogs are found in grassland or short shrubland habitats. They prefer areas of low vegetation with open vistas. In semiarid shortgrass, mixed, and midgrass rangelands, they seem to prefer to establish colonies near intermittent streams, buffalo wallows, temporary rain catch basins, water impoundments, old fields, homestead sites, windmills, old cemetaries, and similar situations. They do not tolerate tall vegetation well and avoid heavy brush and heavily timbered areas (Boddicker, 1983). Prairie dogs are sometimes found in tall grass prairie, but only in areas where heavy grazing by cattle, other livestock or wild ruminants keeps grasses short.

1. Blacktail prairie dogs - Dry upland prairies, shortgrass, or mixed grass prairie. Species in this group form colonies ("dogtowns") of up to several thousand (historically several million) individuals. Colonies may be up to several hundred acres in size.

2. Whitetail prairie dogs - Mountain valleys up to 12,500 feet in altitude, or desert, depending on species or subspecies. In open or slightly brushy country, with scattered woody plants. Usually found in isolated pairs, small families, or temporary family groups (clans); the species in this group are not as social as the blacktail prairie dogs.

4. Diet:

Prairie dogs feed primarily on grasses and forbs. Summers and Linder (1978) found buffalo grass, scarlet globemallow, threadleaf sedge, blue grama, and western wheatgrass to be the preferred food items. Other dietary components include six-weeks fescue, sand dropseed, foxtail, and various brome grasses. Prairie dogs eat seeds, succulent leaves, and stems as well as roots of plants. When grasses are scarce, they eat cactus, four-wing saltbush, rabbit brush, and bark from oak sprouts. They also feed heavily on grasshoppers and other insects during summer months (Whitehead, 1927). Large prairie dog populations can graze significant amounts of forage from the range, causing vegetational changes which may persist for many years or decades, depending on such factors as soil type and climate.

5. Life Cycles:

1. Blacktail prairie dogs - Blacktails are diurnal, gregarious, and live in colonies (towns) of up to several thousand individuals. Population densities vary from 5 to over 50 prairie dogs per acre, with as many as 50 burrows per acre in varying degrees of use. Colonies are subdivided by topographical features into smaller assemblages known as wards which in turn are further divided into groups of related individuals (coteries) which defend home territories. Territories vary in size from .5 acres (Tileston and Lechleitner, 1966) to .7 acre (King, 1959). The coterie is the basic social unit, usually consisting of one or more adult males, several adult females, and associated offspring. Adult males are dominant within the coterie; when two or more males occur within the same coterie, one tends to dominate (Tileston and Lechleitner, 1966). All members of the coterie share food and burrows. Coterie members spend much time grooming and playing with each other. At least one animal in each coterie is on alert for predators while the others feed, play, or rest on top of the mounds. There is a highly developed communication system, with separate danger signals for terrestrial and aerial predators. Blacktail prairie dogs utilize at least 10 different sounds for various communications (Costello, 1970; Waring, 1970). Blacktail prairie dogs are not true hibernators but may become dormant in winter for periods of up to several days.

Burrow systems range from very simple to extensive, with several chambers and escape tunnels. Separate chambers may be used as nurseries, latrines, resting areas, and air pockets in the event of flooding. Burrow systems vary in size, according to local soil conditions, depth of water tables, and the needs of individual prairie dogs. Burrows range from 15 to over 85 feet in length; depth varies from 3 feet to 10 feet (Sheets et al., 1971). Most tunnels have an entrance through a crater-shaped earthen mound, or a dome mound (a pile of earth beside the entrance hole) usually with a chamber a short distance inside, where the animal can turn around. It is believed that burrow systems with both dome and crater entrances provide ventilation to chambers far below ground (Ferrara, 1985). Some burrow systems interconnect, and by plugging and unplugging tunnels and chambers, prairie dogs can modify systems to suit their current needs (Costello, 1970).

Mounds at entrances are built by pushing up soil from below and packing it firm. Blacktail prairie dogs use their forepaws and noses to pack earth. Mounds serve as lookout posts for predators and as dikes in the event of flooding.

Mating season for blacktail prairie dogs begins during early January in Kansas, and continues for the next 2-3 weeks. Further north, breeding may begin as late as early March. Most prairie dogs first breed at the age of 2 years, although some females may breed in their first year (Hoogland, 1982). After a gestation period of 28-32 days, 2-5 (rarely up to 8) young are born. Only one litter of young per year is produced. Young are born blind and hairless. Eyes open at 5 weeks, and by 6 weeks, the young venture above ground. Females do not allow adult males in or near nest burrows from the time they mate until the pups first appear aboveground. Females usually remain in a single coterie for life but young males tend to disperse from parental coteries from June to September, and adult males tend to move to other coteries before their female offspring mature (Hoogland, 1982). Blacktail prairie dogs live 4-5 years in the wild, and up to 8 years in captivity.

2. Whitetail prairie dogs - Life cycles are similar to those of the blacktail prairie dogs with some exceptions. Whitetails hibernate from October or November to March in high mountain valleys. The young of the year hibernate with adults in parental burrow systems. The mating season occurs from late March to mid-April, and young are born in early May. In deserts, whitetail prairie dogs may aestivate in July and August.

   Burrows are extensive, but mounds are seldom constructed. Whitetail and Gunnison's prairie dogs form piles of dirt around the burrow entrance. Zuni prairie dogs often form earthen ramps at the entrances to their burrows (Costello, 1970).

Due to the shorter season at higher elevations, whitetail prairie dogs spend less time above ground, and are not as colonial as blacktail prairie dogs; social interactions are not as well developed as among blacktail prairie dogs, due possibly to the more extensive vegetation which affords cover from predators, reducing the need for high levels of vigilance (Hoogland, 1981). Whitetail prairie dogs form clans (temporary family groups), usually dominated by adult females and consisting of mothers and their current litters occupying specific burrow systems. Well defined and defended boundaries do not exist between clans as is the case with coteries. Members of the same clan feed together and members of different clans normally interact with little conflict. Whitetail prairie dogs occupy larger areas and have much lower population densities than do blacktails.

Whitetail prairie dogs disperse in much the same manner as do blacktails, but do so over a wider area, resulting in much lower population densities.

Gunnison's prairie dogs usually occur singly, in pairs, or in widely separated family groups.

6. Seasonal Abundance:

Prairie dogs, like most small mammals, are most abundant after the young have been born in February and March, and before they disperse from the burrows, beginning in June.

Populations are lowest at the beginning of the breeding season, at about 2.5 per acre for blacktail and 1.4 per acre for whitetail prairie dogs. Maximum population densities are quite variable and are strongly influenced by local environmental factors (Campbell and Clark, 1981). Tileston and Lechleitner (1966) report maximum densities of approximately 13 blacktail or 3.5 whitetail prairie dogs per acre, but colonies with nearly 30 blacktail prairie dogs per acre have been reported (Alexander, in litt.). Whitetail prairie dogs have been reported at densities of 20 per acre (Alexander, in litt.).

7. Responses to Environmental Factors:

Exceptionally dry years, in conjunction with heavy grazing by wildlife or cattle, enchance prairie dog colony expansion. Wet years with abundant vegetation growth produce the opposite effect (Boddicker, 1983). Prairie dogs are most abundant in areas intensively grazed by livestock or wild ruminants which keep plants clipped to low heights (Uresk et al., 1981). Low vegetation allows prairie dogs to see predators and communicate visually with each other. In areas where vegetation is allowed to grow tall, blacktail colonies tend to decline. Vegetation height has little effect on whitetail prairie dog colonies (Hoogland, 1981). Dispersal of young in June to September serves to expand existing colonies, establish new colonies, or reestablish old, abandoned, or poisoned colonies in suitable habitat (Garrett and Franklin, 1981). Movement of breeding blacktail prairie dog males to different coteries helps to prevent inbreeding (Hoogland, 1982).

Colonies tend to expand outward after available vegetation in the central areas has been replaced by unpalatable plants (mostly forbs) which are not grazed by prairie dogs (Fagerstone, 1981). Immigration and emmigration have little impact on the overall dynamics of blacktail prairie dogs, but may be important for whitetails (Tileston and Lechleitner, 1966).

8. Impact of Prairie Dogs:

8.1 Direct Impact:

Blacktail prairie dogs cut vegetation from around their burrows for food, for nest lining material, and for removal of possible cover for predators, as well as to keep open lines of sight for communication and to scout for predators. Prairie dogs feed on the same forage as cattle and native ruminants, competing directly with them. The amount of aboveground forage eaten or made unavailable to livestock and other wildlife due to prairie dogs and other dogtown inhabitants is about 24% of the total potential annual production (Hansen and Gold, 1977). Whitetail prairie dogs generally do not cut vegetation for other than food uses (Tileston and Lechleitner, 1966).

Burrowing and the resultant bare earth around burrow entrances in colonies (resulting from subsoil being brought to the surface), may cause rough pasture surfaces and slow grass regeneration (Boddicker, 1983).

High-density colonies often overgraze forage to the point of reducing food sources for other wildlife. When the flora of an area has been changed by the action of prairie dog colonies, the fauna also changes. Declines in numbers of sharp-tailed grouse, pheasants, quail, and other game birds have been noted in the vicinity of colonies. Use of areas by mule deer and white-tailed deer may also be decreased.

Cottontail rabbit and jackrabbit populations may increase due in part to increased forb populations. Black-footed ferrets depend on prairie dogs as their sole source of food and shelter (ferrets nest in abandoned burrows). Burrowing owls and prairie rattlesnakes are also often found in prairie dog colonies, living in abandoned burrow systems. Pronghorn antelope thrive on range where prairie dog colonies occur, feeding on forbs which grow in place of grasses (Boddicker, 1983). Short-term prairie dog impact enhances grazing for bison by increasing forage nitrogen concentration and forage accessibility (Layne, 1980). Prairie chickens and sharp-tailed grouse may utilize colonies for leks (mating display areas) during breeding season. Mice, ground squirrels, toads, tiger salamanders, and ornate box turtles, as well as many insect species, may utilize burrows for temporary or permanent shelter.

8.2 Indirect Impact:

Prairie dogs are susceptible to and may harbor the ectoparasites which transmit sylvatic plague. Prairie dogs are the most frequently cited "reservoirs" for sylvatic plague in the western U.S. (Hansen and Gold, 1977).

9. <u>Natural Enemies:</u>

Prairie dogs are preyed upon by a wide variety of predators including the following: coyotes; bobcats; swift, kit, red, and gray foxes; badgers; longtailed weasels; prairie rattlesnakes; bull snakes; golden eagles; ferruginous hawks; rough-legged hawks, and other large raptors; and the endangered black-footed ferret. Badgers are the principal predator of both whitetail and blacktail prairie dogs (Tileston and Lechleitner, 1966).

Predators are believed to have minimal impacts on prairie dog populations (Campbell and Clark, 1981).

Female prairie dogs have been observed to kill the litters of other (related) females in the same coterie. Presumably this behavior is related to crowding and associated stresses, but has not yet been investigated fully (Ferrara, 1985).

Diseases such as sylvatic plague and tularemia may sweep through overcrowded colonies, killing many of the residents, and causing colony decline due to associated social stresses, as well as leading to increased predation caused by fewer animals acting as sentinels.

III. PRAIRIE DOG MANAGEMENT

1. Population Monitoring Techniques:

The endangered black-footed ferret occurs with and depends upon prairie dogs for food and shelter. Any control program for prairie dogs should recognize the possibility of the existence of black-footed ferrets in the area. Pre-control surveys for black-footed ferrets should be conducted. Contact the U.S. Fish and Wildlife Service, Pierre, S.D., for assistance and information (see Page 15 for addresses and phone numbers).

Color infrared aerial photography at 4000 feet (1370 m) above ground has been used to delineate active prairie dog towns and possible expansion directions. Changes in vegetation common in and around prairie dog towns are used as key indicators which appear as different colors on aerial photographs (Dalsted et al., 1981). Color infrared (CIR) is superior to black and white films because CIR can detect towns less than 4 ha (9.5 acres) in size, a detail not possible with most black and white films.

Observation of colonies using binoculars from a blind is an excellent method of monitoring for activity and to obtain population estimates. See Hoogland (1981; 1982) for interpretations of various behaviors.

Mapping dogtowns, using standard surveying equipment, is an accurate (although expensive in terms of manpower and time) method of surveying the extent of prairie dog towns. It should be kept in mind that not all burrows are occupied at the same time. A rough estimate of numbers can be made by carefully examining burrow entrances for fresh dirt, feces, or signs of dirt packing, and assuming 1 animal per active entrance (Costello, 1970).

Records and detailed maps should be kept to chart the growth or recession of colonies over time.

2. Threshold/Action Population Levels:

Prairie dogs are native animals and in most cases should be left unmolested. In some instances, control may be necessary to prevent colony expansion into areas where the presence of prairie dogs is not desired, where other park resources are threatened, or where diseases such as sylvatic plague may be transmitted.

Contact NPS health and safety officer if sylvatic plague is suspected in your area.

3. <u>Management Alternatives-Nonchemical:</u>

Prairie dogs are usually a problem in areas where the range has been chronically overgrazed by livestock or wildlife.

A nonchemical management program would ideally consist of habitat changes which increase vegetative growth allowing greater predator access, changes in food sources, and resultant social stresses to reduce prairie dog densities to the point where they are no longer pestiferous (Garrett and Franklin, 1981).

Consider resting overgrazed pasture or range by excluding livestock or wildlife for at least 1 season. Snell and Hlavachick (1980) deferred grazing in selected plots in June, July, and August to allow vegetation to recover, followed by spring grazing at double the normal grazing pressure to compete with prairie dogs for early cool-season vegetation. In an area where pastures were managed for 4 successive growing seasons, the prairie dog colony under study was reduced from 110 acres to 12, and grasses reestablished. The authors emphasized that this was not a scientific experiment, but a series of trials and observations.

Increased predation in the area was attempted by placing hay bales in general lines 15-20 feet apart from the edge of draws and other existing cover to the center of prairie dog colonies. No significant difference in predation, however, was noted between the study area and nearby bale-free areas (Snell and Hlavachick, 1980). Further attempts were made to attract predators into the area by placing carcasses of cattle or other livestock, which had died during the winter, in the middle of a colony, although it is not known if predation on prairie dogs was increased. Snell and Hlavachick further suggest that predation by birds may be enhanced by placing raptor perches in the form of dead trees around otherwise treeless colonies. Perches may serve to increase predation, and to stress prairie dogs by the increased presence of raptors, although supporting data are lacking.

Colony expansion may be curtailed or changed in direction by the use of visual barriers. Garrett and Franklin (1981) report that barriers constructed of rows of burlap affixed to steel stakes positioned 30 feet (10 m) apart served to significantly reduce colony expansion compared to areas with no barriers. Barriers may also serve to increase predation by providing increased cover to predators.

Shooting, although historically used to control prairie dogs, is not recommended for general use due to possible safety hazards and overall lack of effectiveness. Shooting tends to make prairie dogs wary of human presence, but does not significantly reduce numbers. Shooting to eliminate individual prairie dogs in special circumstances, and when used in conjunction with other control techniques, may be a viable control measure.

Traps have been used to capture individual prairie dogs causing damage in small areas. Box traps, snares, and #110 Conibear® or equivalent traps have been used with success (Boddicker, 1983). Steel leghold traps are not recommended for humane reasons.

4. Management Alternatives-Chemical:

Diethylstilbestrol (DES), a synthetic estrogen compound, has been found to inhibit reproduction in prairie dogs under experimental conditions (Garrett and Franklin, 1981). Because prairie dogs produce only one litter per year, and females within a colony come into estrus at approximately the same time, management efforts were minimal and did not interfere with other wildlife species. Reproductive inhibitors are expensive, and may be prohibitively so on low value areas such as rangelands, but in some circumstances (such as preventing colony growth onto private lands), may be useful control measures. They must be reapplied each year to inhibit reproduction. Potential effects of DES or other reproductive inhibitors on the food chain have not been quantified. DES is experimental only and is not registered by the EPA. It is not currently recommended for NPS use.

The use of most lethal chemicals on prairie dogs are restricted (with the exception of gas cartridges); permits are required. Zinc phosphide and gas cartridges have been recommended for prairie dog control in the past. Lethal baits for prairie dog control should be used by qualified personnel only. Surveys to determine the presence of black-footed ferrets should be conducted prior to treatment. Contact the U.S. Fish and Wildlife Service for assistance and guidelines in conducting ferret surveys. Henderson (1983) details the proceedure and gives the names and addresses of contact personnel.

Rodenticides are mixed with oats and usually follow a prebait application of non-treated oats. Mortality following rodenticide treatments is generally in excess of 90%. Retreatments are usually required within 3 years due to immigration from untreated areas.

Other methods such as automobile exhaust, dry ice, and gasoline fumes have been used to kill prairie dogs in their burrows. These techniques are not recommended due to lack of effectiveness or danger involved.

Consult your regional IPM coordinator to determine which pesticide, if any, is best suited to your IPM program.

5. Summary of Management Recommendations:

1. Determine the nature of the prairie dog problem. If prairie dogs are in a natural area, no action is recommended.

2. Conduct surveys using aerial photography, mapping, observation, or hole counting methods to estimate populations and densities.

3. Consider habitat modification by deferred grazing or use of visual barriers to control or direct colony growth and expansion. Records should be kept concerning the outcome of such attempts.

4. If necessary, use zinc phosphide or gas cartridges, to reduce prairie dog populations to tolerable levels. Rodenticides may be used in combination with trapping and/or shooting. If rodenticide use is planned, contact:

U.S. Fish and Wildlife Service
P.O. Box 250
Pierre, South Dakota 57501
(605) 224-8692
FTS: 782-5226

for information and assistance in conducting surveys for prairie dog with respect to black-footed ferret populations.

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NATIONAL PARK SERVICE
IPM Information Package

SLUGS AND SNAILS

Final Report

4 January 1985

Submitted To:

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U.S. National Park Service
Washington, D.C. 20240

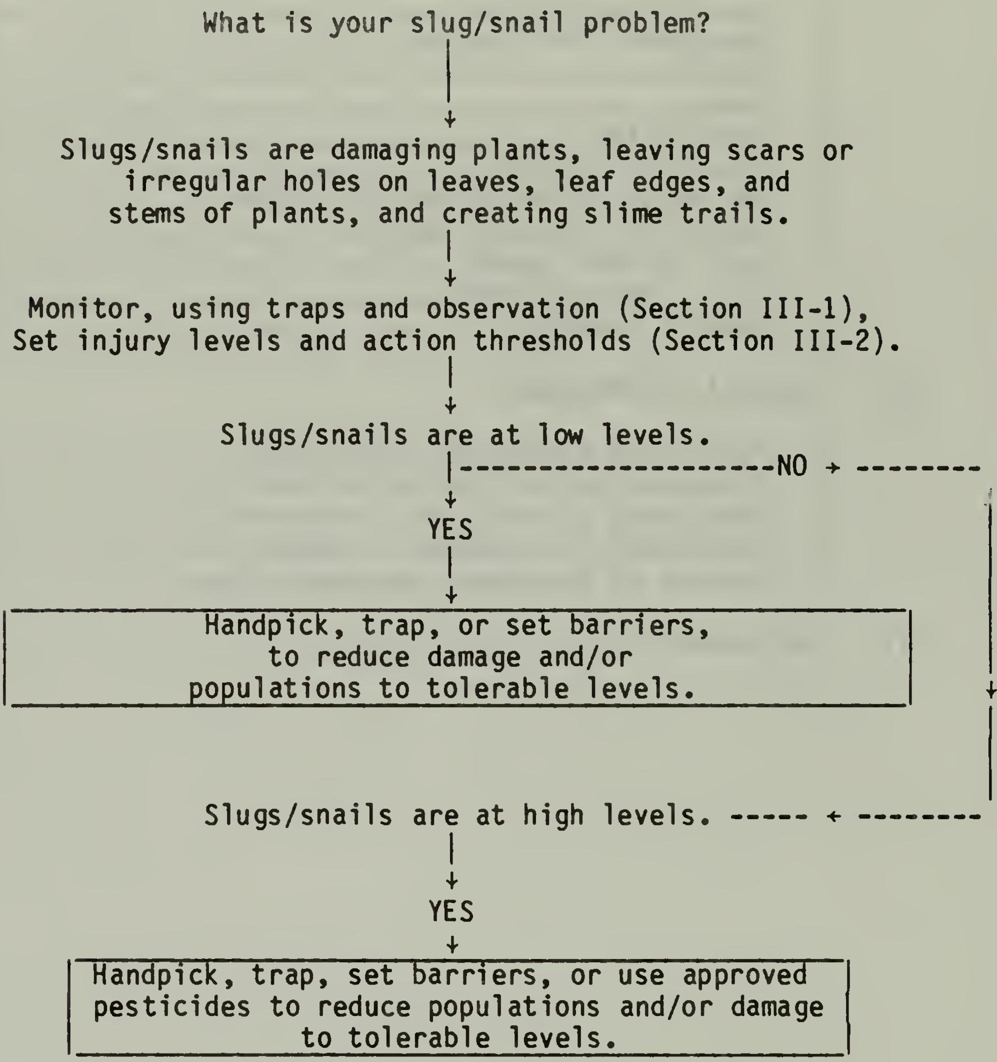
Submitted By:

Dynamac Corporation
11140 Rockville Pike
Rockville, Maryland 20852

CONTENTS

I. SLUG/SNAIL IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



What is your slug/snail problem?

Slugs/snails are damaging plants, leaving scars or irregular holes on leaves, leaf edges, and stems of plants, and creating slime trails.

Monitor, using traps and observation (Section III-1), Set injury levels and action thresholds (Section III-2).

Slugs/snails are at low levels.

Handpick, trap, or set barriers, to reduce damage and/or populations to tolerable levels.

Slugs/snails are at high levels.

Handpick, trap, set barriers, or use approved pesticides to reduce populations and/or damage to tolerable levels.

II. SLUG/SNAIL BIOLOGY AND ECOLOGY

1. Species Described:

Snails and slugs are mollusks, members of the class Gastropoda (the single-shelled mollusks), in the order Stylommatophora. Slugs can be thought of as snails without shells or with shells (mantles) which have been reduced and internalized.

Snails have 2 pairs of tentacles: a large pair with eyes at the tips, and a smaller pair with nostrils at the tips. The mouth is in the center of the head, below the lower pair of tentacles. Below the mouth is the mucus or slime gland. The shell is formed over the visceral hump which contains the internal organs. The shell is formed by the mantle, which forms a fold where the shell joins the body of the snail (foot). The foot contains mucus glands and muscles by which the snail crawls. Several hundred species of snails exist in North America, and it is beyond the scope of this IPM Package to discuss them all. The following species were selected as being among the most important pest species.

1. Brown garden snail - Helix aspersa Muller. The shell is grayish yellow with 5 brown bands and with 4 1/2 - 5 whorls (1 whorl in young specimens). In the adult, the shell is 1 1/4 -1 1/2 inches in diameter.

2. Banded wood snail - Cepaea nemoralis (L.). The shell is light yellow with longitudinal brown stripes. The shell diameter is approximately 1 inch.

3. White garden snail - Theba pisana (Muller). The shell is white with irregular brown markings. The shell is approximately 1 inch in diameter.

4. Subulina snail - Subulina octona (Bruguiere). A small species, less than 1 inch in diameter with a gray, elongate pointed shell.

5. Cellar snails - Oxychilus cellarius (Muller),
   O. draparnaldi (Beck),
   O. helviticus (Blum), and
   O. allairius Muller.

   These are small snails, with shells 1/2 inch in diameter, gray to brown in color, with flat coils. The 4 species are similar in appearance.

Slugs are similar to snails but lack the visceral hump and shell. The mantle (saddle) is a smooth area in the front third of the back. There are over 30 species of slugs in North America. Their life histories and distributions are not completely known. The following species were selected as among the most important pest species.

1. Spotted garden slug - *Limax maximus* L. Body length of this species ranges from 1 1/2 - 7 inches, with the average at 3-5 inches. Smaller or young specimens are dark gray or black. Large adults are yellow gray or brown with 3 rows of black spots from mantle ("saddle") to the rear of the body.

2. Tawny garden slug - *Limax flavus* L. This species is up to 4 inches in length. The color is uniformly tawny to yellowish green with lighter yellow spots. The mantle is yellow and tentacles are bluish. This species exudes a yellowish slime when disturbed.

3. Greenhouse slug - *Milax gagates* (Draparnaud). Body length of this species is 1 1/2 - 3 inches. The color is black to dark gray with longitudinal ridges down the body and a diamond-shaped mark in the center of the back. This species has a prominent, sharp dorsal keel which extends the length of the entire mantle.

4. Gray garden slug - *Deroceras reticulatum* (Muller). The body length is from 3/4 - 1 1/2 inches. The color varies from white to pale yellow, lavender, purple, to almost black. This slug usually has black or brown specks or mottling except on very dark specimens. This species exudes a milky slime when disturbed (Ebeling, 1975).

2. *Geographic Distribution:*

1. Brown garden snail - Found worldwide, this species was introduced as a food animal in California in 1850. This species is common in the southern U.S., where winters are mild.

2. Banded wood snail - This species is found throughout the southern U.S. This species also occurs in Utah.

3. White garden snail - Originating in the Old World, this species was introduced into California in 1914 as a food animal.

4. Subulina snail - Commonly found in greenhouses, throughout the temperate regions. This species is readily transported on potted plants.

5. Cellar snails - These species are found in greenhouses and damp cellars throughout North America.

Slugs are found throughout North America in damp places and where the temperatures are mild in summer. Their distributions and life histories are incompletely known.

1. Spotted garden slug - This species was introduced from Europe. It now occurs throughout the U.S.

2. Tawny slug - Introduced from Europe, this species is widely distributed throughout the U.S., especially in the Southeast (Ebeling, 1975).

3. Greenhouse slug - Introduced from Europe in the 1880's, this species is now widely distributed in the U.S.

4. Gray garden slug - Introduced from Europe, now widely distributed throughout the U.S., especially in humid coastal areas.

3. Habitat:

Snails and slugs are active at night or on dark, cloudy days. They become less active at lower temperatures (below 50°F, 10°C).

Snails and slugs shelter in damp or moist places under or near accumulations of rotting vegetation, piles of boards, bricks, stones, under dense, low vegetation such as ivy, or under the strap-like leaves of such plants as iris. They also may be found in drain pipes, damp cellars or basements, and on well walls.

Snails tend to remain in one area all their lives. Slugs tend to wander; the larger species may travel up to 40 feet per night.

4. Hosts: Slugs and snails feed on a wide variety of dead and living plants. They feed heavily on succulent plants and seedlings. They are common and severe pests in gardens, lawns, and orchards, particularly citrus in California and Florida.

5. Life Cycles: Slugs and snails are hermaphroditic; each individual is capable of fertilizing the eggs of another, and of being fertilized in turn. In some species, individuals change sex as they mature; young adults are males and become females when older.

   Snails lay eggs in nests in the soil or in protected areas under objects. Eggs are laid in masses of 10-200, depending on the season and age of the parent. Incubation is dependent on ambient temperatures, but usually lasts 15-20 days. Young snails remain close to the nest, wandering farther as they grow. Snails reach maturity in 4 months to 3 years, depending on the species and conditions. Common garden snails may live up to 9 years. Outdoors, in colder regions, snails overwinter in sheltered locations.

   Slugs lay eggs in masses of 25 or more under boards, trash, or other damp places. Eggs are oval, light yellow, and covered with a tough elastic membrane. Eggs hatch in 25-30 days, depending on the temperature. Eggs are deposited from early spring to late fall, and in winter in greenhouses. Young slugs normally mature in approximately 1 year, or in 2 years for the larger species. Most slugs overwinter in the egg stage, but some adults may survive mild winters in drain pipes, cellars, storage pits, well walls, or beneath trash or compost piles. Slugs and snails may be active year-round in warm regions and in greenhouses.

6. Seasonal Abundance: Snails and slugs are most common outdoors from early spring (slugs are among the earliest garden pests) to late fall. Most snails become inactive after the first heavy frost, while most slugs are killed by heavy frost. Most snails hibernate under debris, as do some slugs in mild climates.

7. Responses to Environmental Factors:

Aside from seasonal cold, the major factor affecting slugs and snails is moisture. In dry weather, they seek out damp areas or may bury themselves in the ground. Snails may close off their shell by means of the operculum (a horny or limey plate at the entrance), and aestivate for long periods.

8. Impact of Slugs and Snails:

8.1. Direct Impact:

The major direct impact of slugs and snails is the damage caused by their feeding on ornamental and crop plants. Plants not entirely consumed are often ruined for aesthetic purposes by holes in leaves or on the surface of the fruit.

Feeding damage from slugs and snails usually consists of irregular holes in leaves, fruit, or other plant parts, and is frequently associated with slime trails.

8.2. Indirect Impact:

Indirect impacts of slugs and snails are the revulsion they cause to most people, as well as the slime trails they deposit on leaves and other surfaces. In some cases, slugs and snails have been so abundant on roadsides that they have constituted a skid hazard to vehicles.

9. Natural Enemies:

Slugs and snails are preyed upon by toads, box turtles and other tortoises, some predacious beetles and their larvae (e.g. lightning bug larvae), shrews, and birds. Ducks and geese, in particular, are considered to be effective predators of slugs (Vasvary, 1979). Larvae of flies in the family Sciomyzidae are predaceous on snails, and have been considered as biological controls for several snail-borne tropical diseases (Berg and Knutson, 1978). Snails are harvested for human consumption in many areas of the world.

III. SLUG/SNAIL MANAGEMENT

1. Population Monitoring Techniques:

Slugs and snails can be monitored by means of baited traps. Shallow saucers or jars of beer or fermented grape juice set with the tops flush with the soil surface attract snails and slugs, which fall into the liquid and drown. Honey and yeast can be added to the bait to increase effectiveness. Traps should be placed around the area at intervals of about 10 feet, and should be monitored daily to remove accidentally trapped animals.

Clay pots or hollowed-out grapefruit halves can be turned upside down to provide harborage for slugs. Boards or cabbage leaves placed around beds and between rows as resting traps are also effective. Slugs and snails hide beneath these objects during the day and can be identified, counted, and destroyed in the morning.

Keep records on trap placement and on the numbers and types of animals captured. If a trap fails to capture slugs or snails after 2-3 nights, move it to a new location. Change baits twice weekly.

Feeding damage from slugs and snails usually consists of irregular holes in leaves, fruit, or other plant parts, and is frequently associated with slime trails. Slime trails themselves provide evidence of the presence of slugs and snails, and the number of trails per unit area (e.g. per square foot) can provide a rough estimate to the relative abundance of slugs and snails.

2. Threshold/ Action Population Levels:

Levels will vary with area, crops grown (ornamentals, vegetables, orchard), and the species of slug or snail. One large spotted garden slug can cause more damage than several individuals of the smaller species. Tolerable levels of damage will be very low in situations where appearance is important. Most damage to older plants is cosmetic.

3. Management Alternatives - Nonchemical:

Sanitation (the elimination of hiding places such as trash, boards, etc.), will reduce slug and snail populations. Dense, low-growing plants such as ivy or periwinkle (vinca) should not be planted near gardens.

Because snails are rather sedentary, handpicking will usually reduce snail populations below injury levels. Snails should be handpicked daily with records kept of the numbers captured. After the collection frequency falls off sharply, picking can be reduced to once a week. Watering the area in the afternoon is recommended to activate snails and make them easier to locate. Slugs are not easily controlled by handpicking, due to their more migratory habits.

Traps, such as those used for monitoring, are often effective in eliminating small to medium populations of slugs and snails. Large populations may be reduced below injury levels by the use of traps. Records should be kept to determine how well traps are controlling pest slugs and snails.

Barriers of wood ashes, hydrated lime, diatomaceous earth, or Snailproof ® (a commercial product consisting of ground incense-cedar sawmill by-products) applied in bands around gardens have been shown to keep slugs and snails out by acting as repellants or dessicants. Bands should be 1-4 inches wide, and 1/2 inch thick. Bands lose most of their effectiveness when wet. See Barclay (1983) for details and comparisions of various materials.

Snail fences have been used with good results in many areas. Snail fences typically consist of wire window screening with the top inch unravelled and bent out at right angles to provide a sharp barrier over which snails and slugs cannot crawl. Fences should be 8-12 inches high and placed around areas to be protected (McLeod, no date).

Bands of 30 mesh copper screen, placed aound the base of trees in 4-8 inch widths, have been used to prevent snails from climbing into avocado and citrus trees. It should be noted that barriers are not lethal, and that slugs and snails will be diverted to other, unprotected plants.

Biological controls against slugs and snails include domestic ducks, geese, and several species of rove beetles (Staphylinidae), especially the black rove beetle (Ocypus olens), which was introduced into California in 1926. A ciliate protozoan (Tetrahymena rostrata) is under study for use against both snails and slugs.

The Decollate snail (Rumina decollata), a predatory snail, has been introduced into California and Hawaii for control of the brown snail and giant African snail (California Department of Food and Agriculture, 1981). The Decollate snail is believed to be responsible for the serious decline of native tree snail populations on Hawaii and other Pacific islands.

Contact your regional IPM Coordinator before any attempt at introduction is made in your area.

4. Management Alternatives - Chemical:

Baits for slugs and snails are available commercially. Nearly all baits are metaldehyde based. Baits are available in pellet form and should be placed under cover to reduce their attractiveness to wildlife, pets, and children.

Alum, mixed with salt of sulfate of potash and sulfate of alumina, has been used to control slugs in Australia (McLeod, no date).

Fertosan Slug/Snail Killer®, an herbal product; is said to be effective against snails and slugs while harmless to pets and livestock, but no data were found to support this claim. For information, contact: Ecology Action, 5798 Ridgewood Rd., Willits, CA. 95490.

Consult your regional IPM coordinator to determine which pesticide, if any, is best suited to your IPM program.

5. Summary of Management Recommendations:

1. Monitor for snails and slugs using beer traps and resting traps.
2. Handpick or trap snails, and trap slugs to reduce populations below injury levels.
3. Use barriers when feasible to prevent damage in selected areas.
4. Use approved pesticides, such as metaldehyde baits, if necessary.

IV. BIBLIOGRAPHY

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NATIONAL PARK SERVICE
IPM Information Package

STRUCTURAL PESTS I: TERMITES

Final Report

30 September 1984

Submitted To:

William E. Currie
U.S. Environmental Protection Agency
Arlington, Virginia 22202

Submitted By:

Dynamac Corporation
11140 Rockville Pike
Rockville, Maryland 20852

CONTENTS

I. TERMITE IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.

START

↓

What is your termite management problem?

↓

You wish to establish a preventative program to minimize termite damage in existing structures.

--> YES---> Go to Page XXXIVA-3.

↓

NO

↓

You wish to control termites currently infesting one or more structures.

---> YES---> Go to Page XXXIVA-4.

↓

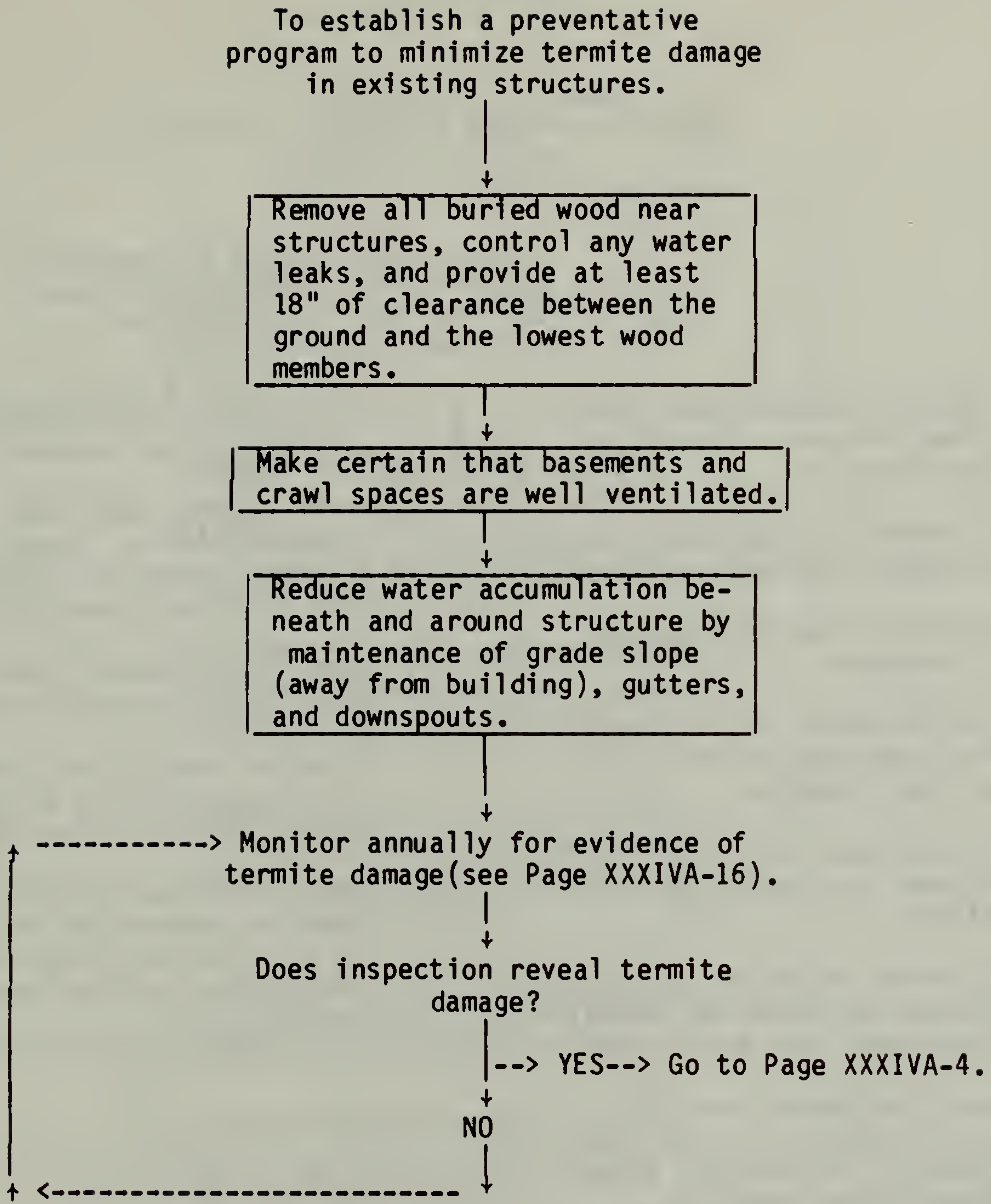
NO

↓

You wish to establish a preventative program to minimize termite damage in buildings to be constructed.

↓

Go to Page XXXIVA-5.



To establish a preventative program to minimize termite damage in existing structures.

Remove all buried wood near structures, control any water leaks, and provide at least 18" of clearance between the ground and the lowest wood members.

Make certain that basements and crawl spaces are well ventilated.

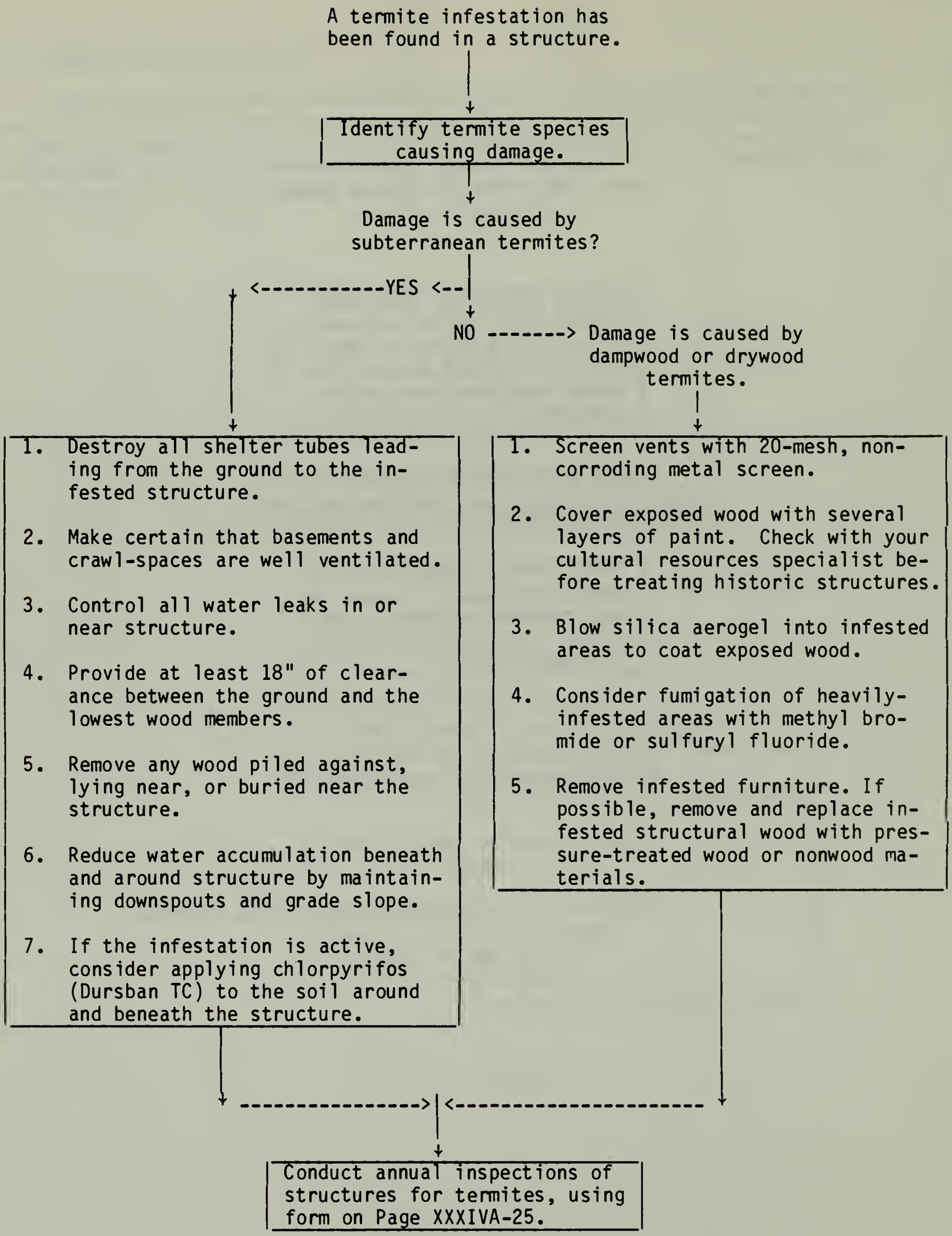
Reduce water accumulation beneath and around structure by maintenance of grade slope (away from building), gutters, and downspouts.

Monitor annually for evidence of termite damage(see Page XXXIVA-16).

Does inspection reveal termite damage?

YES--> Go to Page XXXIVA-4.

NO



A termite infestation has been found in a structure.

Identify termite species causing damage.

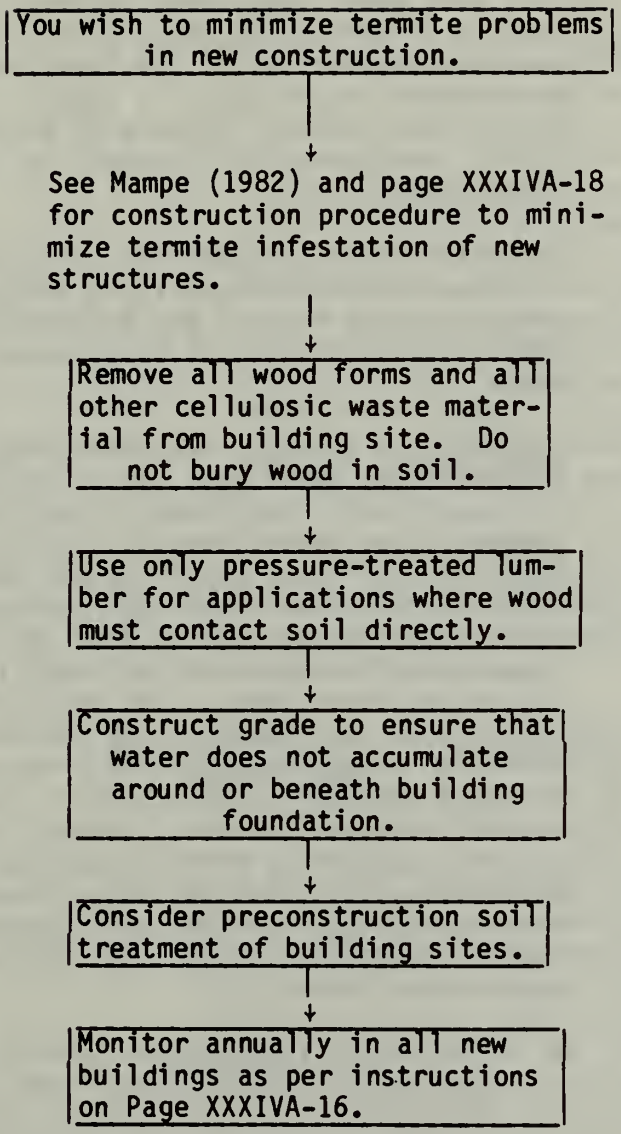
Damage is caused by subterranean termites?

1. Destroy all shelter tubes leading from the ground to the infested structure.
2. Make certain that basements and crawl-spaces are well ventilated.
3. Control all water leaks in or near structure.
4. Provide at least 18" of clearance between the ground and the lowest wood members.
5. Remove any wood piled against, lying near, or buried near the structure.
6. Reduce water accumulation beneath and around structure by maintaining downspouts and grade slope.
7. If the infestation is active, consider applying chlorpyrifos (Dursban TC) to the soil around and beneath the structure.

Damage is caused by dampwood or drywood termites.

1. Screen vents with 20-mesh, noncorroding metal screen.
2. Cover exposed wood with several layers of paint. Check with your cultural resources specialist before treating historic structures.
3. Blow silica aerogel into infested areas to coat exposed wood.
4. Consider fumigation of heavily-infested areas with methyl bromide or sulfuryl fluoride.
5. Remove infested furniture. If possible, remove and replace infested structural wood with pressure-treated wood or nonwood materials.

Conduct annual inspections of structures for termites, using form on Page XXXIVA-25.



You wish to minimize termite problems in new construction.

See Mampe (1982) and page XXXIVA-18 for construction procedure to minimize termite infestation of new structures.

Remove all wood forms and all other cellulosic waste material from building site. Do not bury wood in soil.

Use only pressure-treated lumber for applications where wood must contact soil directly.

Construct grade to ensure that water does not accumulate around or beneath building foundation.

Consider preconstruction soil treatment of building sites.

Monitor annually in all new buildings as per instructions on Page XXXIVA-16.

II. BIOLOGY AND ECOLOGY OF TERMITES

1. Species Described:

Between 45 and 50 species of termites are native to the United States (Mampe, 1982). While these insects are among the most beneficial in their natural habitat due to their consumption and removal of cellulose (in the form of fallen or moribund trees and other plants), this diet makes them among the most destructive insects to structures, furnishings, and commerce. A complete description of the life history and social structure of these pests is beyond the scope of this information package: reviews by Mampe (1982), Ebeling (1975), and Moore (1979) should be consulted for more information.

Termites can be grouped into soil-inhabiting (subterranean) and wood-inhabiting species. The second group includes drywood and dampwood termites. The major characteristics of these groups are described below.

Subterranean termites are relatively small insects which nest in moist soil, or in cellulosic material in contact with the ground. They characteristically produce tubes (made of sand or soil particles cemented together with fecal materials and adhesive secretions) which connect their in-ground colonies with colonized wood or other above-ground food materials. These tubes allow the termites to travel from the relatively dry above-ground wood to the moist soil colony periodically to replace lost body moisture. The tubes serve to isolate the termites from drying wind and sunlight, as well as from natural enemies. When winged reproductive forms (alates) leave the colony to mate, they do so through swarming tubes, which may extend from 4 to 8 inches above ground level. Subterranean termites do the most damage to structural wood of any termite type. The following species are described in detail:

A. The Eastern subterranean termite (Reticulotermes flavipes Kollar) includes three distinct morphologic forms (castes). Alates (winged reproductive forms) are about 1/5" long, black in color with four white opaque wings of equal length. Soldiers are about 1/4" long, pale in color, with enlarged, dark heads bearing very large mandibles. Workers are less than 1/5" long, and are pale to white in color. Alates (referred to as kings and queens) are the only reproductive forms. Soldiers are responsible for defending the colony against enemies. Workers tend and feed the other forms, care for the eggs laid by the queen, and do the work of extending the colony and building shelter tubes.

B. The Western subterranean termite (*R. hesperus* Banks) resembles *R. flavipes* in form and caste structure.

C. The Formosan termite (*Coptotermes formosanus* Shiraki) also includes three castes. Alates have yellow-brown bodies about 1/2 inch in length and are larger and lighter in color than alates of native species. Wings are 2/5" long, and have many hairs. Soldiers usually make up 10-25% of a colony (compared to 2-3% in other subterranean species). They can be identified by their enlarged, rounded heads. Each soldier bears a forward-facing opening (fontanelle) in the front of the head, which can release a sticky, acidic secretion used in colony defense. Workers are pale, and resemble the workers of other subterranean species. All forms are larger than those of native species.

Wood-inhabiting termites do not colonize soil, but live entirely within infested wood. They are much less common than the subterranean species. The two major groups of wood-inhabiting termites found in the United States are;

Drywood termites (*Kalotermes*, *Incisitermes*, and *Cryptotermes* spp.), which live within relatively dry, nondecayed wood, and do not require contact with soil. Their colonies are generally smaller than those of subterranean termites, and may occur in furniture, wood boxes, and dead tree limbs. Drywood termites characteristically produce solid fecal pellets which may be heavily sculptured in appearance, and which can often be found in sawdust-like piles near kick-out holes in infested wood. *Cryptotermes* spp. are the most common wood-inhabiting termites. They attack furniture, woodwork, and floors. These termites require very little moisture, do not require ground contact, and appear to be spread by movement of infested wood.

D. The Western drywood termite (*Incisitermes minor*) produces no worker caste. The work of the colony is performed by juveniles (nymphs), which grow into alates or soldiers. *I. minor* alates are nearly 1/2" long, and are dark brown with red-brown heads and thoraxes, and smoky-black wings. Soldiers are similar in size, wingless, pale in color, with enlarged, pigmented heads with enlarged mandibles.

E. Alates of the common drywood termite (Cryptotermes brevis Walker) are brown, 3/8" long with colorless wings bearing brown veins. Soldiers are about the same length as alates, with broad, high, concave, black heads. The work of the colony is performed by pale nymphs.

Dampwood termites (Zootermopsis spp.) are larger than other types, and require more moisture; therefore, their colonies are usually found in damp or decaying wood or logs. No connection with the soil is required for these species. As in the drywood termites, no worker caste is produced. The feces of these species are large, oval pellets.

F. Alates of the Pacific dampwood termite (Z. angusticollis Hagen) are up to 1" long, yellow-brown or brown in color, with dark brown wings. Soldiers are up to 3/4" long; the elongated head is black, while the thorax and abdomen are a light reddish-brown. The mandibles of these forms are long and toothed. Nymphs are cream-colored to white, and about 1/8-1/2" long. They perform the work of the colony, and mature into reproductives or soldiers. Eggs are about 1/10" long, white, and beanshaped. Liquid feces are produced, along with roughly hexagonal pellets about 1/2" long.

In addition to the species noted above, numerous other subterranean, dampwood, and drywood termites occur in various regions of the U.S. The examples noted are included as being representative of their groups. Consult your U.S. Department of Agriculture Cooperative Extension Service representative or state university entomologist for details on species occuring in your area.

2. Geographic Distribution:

A. Eastern Subterranean Termite - Occurs throughout the United States east of the 100th meridian (e.g., mid-Kansas) where average minimum winter temperatures do not fall below -20°F.

B. Western Subterranean Termite - Occurs throughout the Pacific Coast from British Columbia to Baja California, and east into Idaho and Nevada.

C. Formosan Termite - This species is native to Formosa, Japan, and China, and was probably introduced into the U.S. with military supplies after World War II. Infestations have been recorded in Hawaii, Texas, Louisiana, South Carolina, California, and Florida.

D. Western Drywood Termite - Occurs in California and Arizona; also in the Carribean zone. Isolated infestations may occur elsewhere, due to spread in infested lumber.

E. Common Drywood Termite - Natural range includes most tropical and subtropical regions of the earth. It is believed that _C. brevis_ was introduced into the U.S. on shipments of infested lumber. Its U.S. range includes Florida, Louisiana, and Hawaii, but it is transported to all states in infested wood.

F. Pacific Dampwood Termite - Occurs throughout the Pacific Coast, from British Columbia to Baja California.

3. Habitat:

A. Eastern Subterranean Termite - Lives in soil, or in wood or other cellulosic material contacting soil. Inhabits above-ground wood if connections to soil are maintained through closed earth and cellulose shelter tubes. May on occation colonize permanently moist wood (Ebeling, 1975).

B. Western Subterranean Termite - Similar to A. in habitat preference, but prefers cool, moist, shady locations.

C. Formosan Termite - Lives in wood, several species of living trees, stumps, poles, and buried wood debris. Nests may be independent of soil if another source of water is available.

D. Western Drywood Termite - Lives in nondecayed wood with low moisture content. Substrates include lumber and trees.

E. Common Drywood Termite - This species has not been reported from any natural habitat within the U.S. It apparently only occurs in buildings in the U.S.

F. Pacific Dampwood Termite - Found in wood of fallen conifers in cold and humid areas, in beach areas with high water tables, coastal forests, and in areas where wood is kept moist by irrigation water.

4. Hosts:

A. Eastern Subterranean Termite - Most wood, except juniper, teak, and redwood (heartwood).

B. Western Subterranean Termite - See 4.A.

C. Formosan Termite - Most wood.

D. Western Drywood Termite - Hosts include English walnut, eucalyptus, *Citrus* spp., apricot, avocado, alder, almond, cherry, California laurel, Monterey cyprus, oak, peach, pear, sycamore, plum, willow, and other ornamentals. Termites enter trees through injuries.

E. Common Drywood Termite - No natural hosts known in U.S.

F. Pacific Dampwood Termite - Pacific coniferous trees and shrubs.

5. Life Cycles:

A. Eastern Subterranean Termite - Alates swarm in January to August, depending on location. Termites are not good fliers, and usually do not spread more than 200 yards from their swarm tubes before landing, shedding their wings, and pairing off. Each new pair seeks a dark cavity in nearby soil or ground-level wood, excavates the cavity, and mates within a day after swarming. Mated pairs remain together until death.

Growth of a colony from a primary pair may be slow. As few as 12-15 eggs are laid during the first year. The eggs hatch in about a month. Young nymphs resemble miniature adults during their first two instars, each of which may last up to a month. By the third instar, future reproductives can be differentiated from future workers and soldiers. Workers and soldiers mature in about 1 year, while reproductives may require 2 years. A functional colony may contain as many as 100,000 individuals. In addition to a primary king and queen, secondary reproductives (with wing buds) may be present in large numbers (up to several hundred). These also contribute eggs to the colony and can take over the functions of the primary pair should they die. The life span of an individual termite may be as long as 5 years. Such longevity, combined with their cryptobiotic ("hidden") lifestyle, constant reproduction, and permanent food supply, make termite colonies long-lived.

Newly-hatched nymphs do not contain the intestinal flora needed for wood feeding. They obtain it during the first instar by fecal feeding and by feeding on the abdominal secretions of workers.

B. Western Subterranean Termite - The life cycle of this species is similar to that of the Eastern subterranean termite (*R. flavipes*). Alates swarm in the fall and winter, especially after rain. Sporadic flights may occur during the winter or spring if the weather is dry. Colony formation varies from that of *R. flavipes* in that the Western termite queen lives in a large, special chamber with the king and a number of soldiers. Periodically, the queen and her entourage will move to another chamber of similar size.

C. Formosan Termite - Alates swarm from March through July in the U.S., and in spring and fall in Hawaii. In Louisiana, alates fly between dusk and midnight (Jones and La Fage, 1980). After flying briefly, the reproductives pair off and search for a suitable nesting site in wood or the soil. When a site is located, the insects construct a small chamber in which mating occurs. The queen begins laying eggs about 5-15 days after the mating chamber is built. One to four eggs are produced each day, until about 30 are laid. The eggs hatch in 24-32 days. About 10% develop into soldiers, while the rest become workers; this ratio is maintained during the first few years of colony growth. No more eggs are produced until after the first brood hatches. Mature queens may lay up to 1000 eggs per day. Colony development is slow; a two year old nest may contain only 250 insects. Third year colonies may contain 1,250, and fourth-year colonies may contain 50,000. The age of a colony when the first alates are produced is not known, but is believed to be greater than 5 years (Jones and La Fage, 1980). Old colonies (over 15 years) may contain millions of individuals.

D. Western Drywood Termite - Alates swarm from June to December depending on location. They may travel up to a mile in wind currents before dropping to the ground, shedding their wings, pairing off, mating, and searching for a site to begin a colony. Once a pair excavates a cavity, they seal themselves in, and the queen lays 2-5 eggs during the first year. Nymphs obtain their intestinal flora from secretions of the adults and begin expanding the colony. Each year, from late spring to late fall, the queen lays 1-12 eggs per day for 7-10 days, rests for about 30 days, and repeats the egg-laying cycle. By the end of the second year, the colony may contain up to 40 termites; by the end of the fifteenth year, there may be as many as 2,600. The first alates are released after about 4 years.

Alates and soldiers require about 1 year to mature. Alates develop after 7 nymphal instars; soldiers require 4-7. Queens reach their maximum egg laying rate at about 10-12 years of age, after which the rate decreases and another female takes over as the primary queen.

E. Common Drywood Termite - Similar to 5.D., but swarming occurs in May or June.

F. Pacific Dampwood Termite - Alates swarm mainly from August to October. Females excavate openings in wet or decaying wood, which are later entered by males. The opening is then sealed with wood pellets and feces, and the pair mate within two weeks of flying. Within 14-18 days after mating, the female lays between 6 and 22 eggs. A second clutch is laid the following spring. Supplementary reproductives contribute additional eggs, so that a colony may eventually have 4,000 individuals.

6. <u>Seasonal Abundance</u>:

A. Eastern Subterranean Termite - Swarming usually occurs in mid- to late spring, although flights may occur as early as January or (rarely) as late as July or August. Swarms produced during the early part of the year are usually larger than later swarms. Outdoor colonies must move below the frostline during cold weather.

B. Western Subterranean Termite - Swarming may occur from early autumn through winter, and (rarely) as late as May or June. The largest swarms occur in the fall, especially on sunny days following rain. In dry years, emergence may be delayed until the soil has been softened by winter or spring rains so that nests can be established.

C. Formosan Termite - Swarming occurs from March through July in the southern U.S., peaking in May and June in Louisiana. Spring and fall swarming occur in Hawaii.

D. Western Drywood Termite - Alates swarm during sunny days in early summer to late fall.

E. Common Drywood Termite - Swarming takes place in May or June in the United States.

F. Pacific Dampwood Termite - Swarming may take place throughout the year, but is most evident between August and October, especially after rain.

7. Responses to Environmental Factors:

A. Eastern Subterranean Termite - Alates are attracted to light; all other forms avoid light, possibly as a means of avoiding dry air. Colonies will move to lower chambers in response to cold surface temperatures. Individuals cannot survive in above ground wood without frequently traveling to the in-soil colony through shelter tubes to replenish lost body moisture. Blockage of access to the in-ground colony generally results in the death of individuals trapped above-ground.

B. Western Subterranean Termite - Alates are attracted to light, and generally swarm on sunny days. They will fly during cloudy weather, if the temperature is above 64°F (Light, 1934). Also see 7.A.

C. Formosan Termite - Alates are attracted to light. Bess (1970) has suggested that high humidity is required for colony initiation. Nests are constructed of "carton", a composite of feces, saliva, and digested wood which retains available water. If soil water is not available, Formosan termites will consume water that condenses on pipes or that collects in structural defects and rain gutters. This species often colonizes soil that is poor in cellulosic materials, and will extend foraging galleries up to 200 feet from the main nest.

D. Western Drywood Termite - Alates are attracted to light. Under very dry conditions, individuals will avoid dessication by sealing themselves within cavities in wood and huddling together to conserve moisture. One individual was reported to have survived for 245 days in a block of kiln-dried wood under dessication; when placed near water, the termite drank until turgid, then behaved normally (Ebeling, 1975).

E. Common Drywood Termite - No information was available.

F. Pacific Dampwood Termite - Alates are attracted to light. Swarming is most evident after rains.

8. Impact of Termites:

8.1 Direct Impact:

A. Eastern Subterranean Termite - This pest will destroy any cellulosic material, including lumber, paper, cotton, books, and dead tree roots. Noncellulosic materials (e.g., underground cables, cement) may be damaged by termites which chew through them in search of food.

B. Western Subterranean Termite - See 8.1.A.

C. Formosan Termite - See 8.1.A. These termites will consume wood that is resistant to attack by native termites, and can destroy wood up to six times faster than native species can. Soldiers can penetrate lead, asphalt, plaster, mortar, creosote, rubber,and plastics (by means of the acidic secretions of their fontanelles) to obtain underlying wood.

D. Western Drywood Termite - This species will damage dead trees, lumber, utility poles, wooden structural members in buildings, bridge and marine pilings, and redwood (which native subterranean termites usually do not attack).

E. Common Drywood Termite - Damages furniture, woodwork, and flooring, but can attack structural wood if infestations are allowed to proceed for many years.

F. Pacific Dampwood Termite - Causes most damage where ground-wood contact points exist (e.g., bridge timbers, foundation wood). Will work up from foundations to roof rafters.

8.2 Indirect Impact:

All termites can spread wood-rotting fungi while tunneling through infested wood.. In addition, termite damage to wood flooring and foundations may reduce the structural soundness of the materials, which could lead to injury to personnel and visitors using the infested structure. Swarming alates may create nuisance situations, especially if flights occur inside infested structures.

9. Natural Enemies: Natural enemies of termites include ants, birds, spiders, centipedes, amphibians, and small mammals. Except during swarming, termites are protected from their natural enemies within infested wood or in shelter tubes. Several species of fungi are known to infect termites; however, none has undergone field testing as a biocontrol agent. A nematode (Neoplectana carpocapsae) that carries a parasitic bacterium (Xenorhabdus nematophilus) is under development as a biologic termiticide (Weidner, 1983).

III. TERMITE MANAGEMENT

1. Population Monitoring Techniques:

The most effective technique available for monitoring termite populations in or near buildings is an annual inspection for evidence of termite damage in wood. Signs of termite presence include:

A. Eastern Subterranean Termite -

1. Reports or observation of large numbers of alates in or near a structure. The alates may be the first evidence of infestation, and may be confused with flying ants. Termite alates have four wings of the same size, all nearly 2 times the body length. Ants have one pair of wings longer than the other, with the longest as long as the body. Termite alates are not "wasp-waisted", as are ants. Also, the antennae of ants are elbowed, while those of termites are not.

2. Dark areas or blisters in flooring or other wood framing. Such areas can easily be crushed with a sharp tool, revealing termite cavities and perhaps the insects themselves.

3. Termite-infested wood comes apart easily when probed with a screwdriver, revealing termite tunnels, frass or fecal deposits, and live termites, if the infestation is active.

4. Subterranean termite infestations are connected to the underground colony by earth and cellulose shelter tubes. The presence of such tubes on the surface of wood or other structural surfaces indicates that an infestation exists. The absence of such tubes, however, does not mean that termites are not present.

B. Western Subterranean Termite - See 1.A.

C. Formosan Termite - See 1.A. Also monitor for the following signs of infestation:

1. Reports of unusually large numbers of alates, and/or of unusually large proportions of soldiers.

2. Soldiers will swarm onto the hand of anyone probing an infested site with a finger (and will bite).

3. Formosan termite alates swarm after sunset, unlike those of native species, which are day fliers.

4. Evidence of termite activity may be found near sources of water, such as plumbing leaks or wood under roofs.

D. Western Drywood Termite:

1. The first evidence of drywood termite attack is usually the presence of piles of brownish fecal pellets below small holes (about 1/8" in diameter) or cracks in infested wood.

2. The flight of alates in or near a structure during warm, sunny autumn days indicates a nearby colony.

3. See 1.A.3. Infested wood may be filled with loose fecal pellets. Since drywood termites consume wood up to the paint, apparent paint blisters are formed. These break on slight pressure, releasing fecal pellets.

E. Common Drywood Termite - See 1.A.

F. Pacific Dampwood Termite - See 1.A.

A sample termite inspection report form is shown on Page XXXIVA-25. Similar forms should be made out for each building inspected, and retained for later reference. A key for identifying the signs and symptoms of termite damage can be found in Kaae and Young (1976).

2. <u>Threshold/ Action Population Levels:</u>

Since the damage caused by termites (especially subterranean and drywood species) can be so extensive, and the pests themselves may survive undetected until damaging levels are reached, the presence of an active infestation is the threshold level for termites. In natural areas, termites are beneficial, and should not be managed.

3. Management Alternatives-Nonchemical:

A. Eastern Subterranean Termite -

1. Prevention of wood-ground contact - Structural wood should not be less than 18" above the ground. Wood steps should be supported on a concrete base extending at least 6" above ground level, and should be separated from the main structure by a metal shield. Termites will build shelter tubes over the shield to reach wood; such tubes will be easily seen. Any shelter tubes found should be destroyed.

2. Removal of wood debris - All wood (stumps, scrap wood, wood chips, sawdust, or form boards) should be removed from beneath all structures. No wood should be buried in the fill near buildings.

3. Avoidance of excess moisture - Subterranean termites prefer moist soil; therefore, building sites must be graded to prevent accumulations of moisture around or under a structure. Downspouts should carry water away from the building. All plumbing leaks should be corrected. Vents should allow cross-ventilation and removal of moisture. Placing plastic sheets on the ground can keep moisture out of the structure.

4. Foundation protection - Building foundations should be of solid reinforced concrete, to prevent cracking. Where stone, brick, or masonry is used, it should have a metal shield or 4" concrete capping. Exterior foundation walls should extend at least 18" above the outside grade line. Where foundation walls are even with or lower than the outside grade, they should be raised to at least 4" above grade, or a concrete (1:3 Portland cement:sand) flash wall should be installed against the building, extending from 6" below the grade line to at least 6" above the grade line. This procedure cannot be performed on historic buildings.

5. Ground leveling - The ground beneath a building should be leveled, and should provide at least 18" of clear space between horizontal timbers and the ground (24-30" in humid parts of the U.S.).

6. Basement protection - Pressure-treated wood (see Page XXXIVA-20, 4.A.2) should be used in the basement of a structure, if the wood will touch the ground. No wood should extend into the foundation concrete, and all form pieces should be removed. Foundation or wall cracks should be sealed with noncorrosive metal expansion joints. Wooden partitions or stored materials

should be placed on concrete or shielded supports. Hatchways and windowframes should be made of concrete, metal, or pressure-treated wood.

7. Basement venting - In unfinished basements, vents should provide cross-ventilation. At least 2 square feet of air space per each 25 linear feet of foundation wall should be provided. Dead air pockets should be prevented. Vent frames must not be in contact with ground.

Detailed information on these and other related techniques can be found in Mampe (1982), Anon. (1971), Anon. (1977), and Moore (1979).

B. Western Subterranean Termite - See 3.A.

C. Formosan Termite - See 3.A.

D. Western Drywood Termite

1. Building protection - Infested wood should be removed and replaced with pressure treated lumber. Coating exposed wood with several layers of paint will inhibit termite activity (painting of historic structures may not be possible). Also, covering vents with 20-gauge mesh will prevent entry of termites. Attics should be screened to keep swarming alates out.

2. Stored lumber protection - Wood to be protected should be supported on piers made of concrete or pressure-treated wood. Debris should be removed from the storage area.

E. Common Drywood Termite - No information is available.

F. Pacific Dampwood termite

1. Moisture reduction - See 3.A.3.

4. Management Alternatives - Chemical:

A. Eastern Subterranean Termite

1. Soil treatment - The goal of soil treatment is the creation of a zone of poisonous soil between the structure to be protected and the termites. Soil treatment should be performed, prior to construction, by trained applicators (due to the complexity of the operations required and the toxicity of the chemicals used). Chlorpyrifos (Dursban TC) is the only pesticide currently recommended for soil application by the National Park Service.

2. Wood preservatives - Lumber directly exposed to termite attack should be treated with preservatives to inhibit infestation. Most lumber manufacturers use a pressure-treatment process to impregnate lumber with a registered preservative, such as chromated copper arsenate (CCA). Treated lumber should be used only where termite attack or fungal decay is likely. An additional benefit of the use of pressure-treated lumber is that, since termites will not consume it, they must tunnel over it, exposing their shelter tubes to view.

B. Western Subterranean Termite - See 4.A.

C. Formosan Termite - See 4.A. In addition, studies are being conducted to support labeling of methyl bromide, sulfuryl fluoride, and Vikane® for fumigation of secondary colonies.

D. Western Drywood Termite

1. Silica aerogel - Silica aerogel can be blown into attics and similar areas where active infestations are found. The dust coats exposed wood members, termite fecal pellets, and swarming alates, and is effective indefinitely under dry conditions.

2. Fumigation - Individual infested structures can be fumigated with methyl bromide to eliminate infestations. This procedure is hazardous, and should only be performed by certified applicators. Sulfuryl floride has also been used.

E. Common Drywood Termite -

1. Fumigation - Fumigation of infested wood with sulfuryl floride, chloropicrin, or methyl bromide

has been shown to be effective against these insects (Bess, 1971). Active colonies may be controlled by injection of chlorpyrifos solutions into galleries through kickout holes.

F. Pacific Dampwood Termite - No information is available.

5. <u>Summary of Management Recommendations:</u>

A. Eastern Subterranean Termite -

1. Inspect all structures for termite damage every year.
2. Remove all ground-wood contact points.
3. Use only wood which has been pressure-treated with a preservative (e.g., CCA) where wood will contact ground.
4. Maximize drainage and cross-ventilation under structures, and use other techniques to limit moisture in and around structural wood.
5. Use construction practices noted on Pages XXXIVA-18-19 to inhibit termite damage.
6. Remove all cellulosic debris (such as wood and paper) from soil in building area.
7. Where active infestations are found, treat soil around and under foundations with chlorpyrifos. Spot-treat sources of infestation, if possible.

B. Western Subterranean Termite - See 5.A.

C. Formosan Termite - See 5.A. Also repair or remove sources of water. Find and treat nearby colonies in wood, structures, or soil.

D. Western Drywood Termite -

1. Cover all entry points (windows, vents, louvers, eaves, etc.) with 20 mesh noncorrosive metal screening, to prevent termite entry into structure.
2. Maintain smooth exterior building surfaces; fill all cracks and joints before painting. A heavy layer of paint on exterior wood will inhibit infestation.

3. Replace infested wood with pressure-treated lumber, or inject approved termiticides into active termite nests.

4. Treat attics and similar spaces with silica aerogels if active infestations are found.

5. Fumigate heavily-infested structures with a registered fumigant.

E. Common Drywood Termites - See 5.D.

F. Pacific Dampwood Termite - See 5.D.

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V. SAMPLE TERMITE INSPECTION REPORT FORM (SIDE 1.)

Building Name or Number ______________________________

Building Address or Location ______________________________

Person to Contact ______________________________ Phone ______________

Survey Date ______________ Inspector ______________________________

Building Data (Check One):

1. TYPE OF STRUCTURE
   basement___ slab___
   crawl___ combination___

2. FOUNDATION
   concrete___ hollow block or tile___
   stone___ multiple brick___
   brick veneer___ single brick___
   piers only___ combination___

3. EXTERIOR
   wood___ hollow block___
   stone___ stucco on frame___
   brick___ stucco on masonry___

4. PORCHES
   wood___ dirt filled___
   masonry___ hollow___
   type flooring on masonry___

5. BASEMENT
   ceiling finished___with______
   walls finished___with______
   floor finished___with______
   venting sufficient?___
   vents screened?___

6. SLAB
   supported___ floating___
   monolithic___ wood over slab___
   floor covered___with______
   plumbing accessible?___
   heating accessible?___
   type of heat______________
   blue prints available?___

7. GENERAL INFORMATION
   paving against foundation___feet
   planters___ shrubs, plants___
   soil type___________
   clearance in crawl space___inches
   are all areas accessible?___
   must openings be made?___
   are wood supports in contact with
   ground or embedded in slab?___
   are form boards present?___
   buried wood waste present?___
   accumulations of water present?___
   plumbing leaks present?___
   is grade correct?___
   wells or other special precautions?___

Inspection Data:

1. INFESTATION FOUND
   termites?___ species__________________ is infestation active?___
   other insects?___ species__________
   wood decay fungi?___

2. LOCATION, INTENSITY OF INFESTATION
   Describe fully, showing locations on building diagram on reverse side of this sheet.

SAMPLE TERMITE INSPECTION REPORT FORM (SIDE 2.)

3. TREATMENT/REPAIR RECOMMENDATIONS

4. DATE OF TREATMENT__________________________
DATE OF REPAIR____________________________

5. REMARKS:

6. BUILDING DIAGRAM: show infestations, needed repairs, and nearby sources of termites.

INSTRUCTIONS FOR TERMITE INSPECTION

When inspecting a structure for termite damage, the following procedures should be carried out:

- Inside and outside walls should be carefully examined for termite shelter tubes, especially near soil, in basements, and in crawl spaces.

- Exposed wood (e.g., floor joists, sills, roof trusses) should be tapped with a tool, to indicate (by difference in sound) whether the wood has been damaged internally. Probe suspicious areas with a screwdriver. Infested wood comes apart easily, revealing termite tunnels, frass deposits, fecal pellets, and/or the termites themselves. Wood showing paint blistering, and flooring showing blistering or isolated stained areas, should also be probed.

- Wood paneling and other wall finishings on basement walls, wood partitions, and other basement wood which extends from masonry to sills or joists should be examined.

- Plumbing and utility fixture passages through the basement floor or foundation should be checked.

- Stone, concrete, cinder block, hollow tile or brick walls should be examined for cracks or holes through which termites could enter.

- Unscreened openings should be noted.

- Signs of termites, such as shed wings, dead individuals, or piles of fecal pellets should be noted.

- Signs of buried wood near or under the building should be noted, and exposed debris should be examined for termite damage.

- The results of each inspection should be recorded on a form, and retained for future reference. A drawing of each building inspected should be made on the reverse side of the form used for that building, detailing a) sites of observed infestations, and b) structural or other repairs necessary to maintain structure so as to prevent new infestations.

NATIONAL PARK SERVICE
IPM Information Package

WATER HYACINTH

Final Report

7 December 1984

Submitted To:

Mr. Gary H. Johnston
National Park Service, USDI
Washington, D.C. 20240

Submitted By:

Dynamac Corporation
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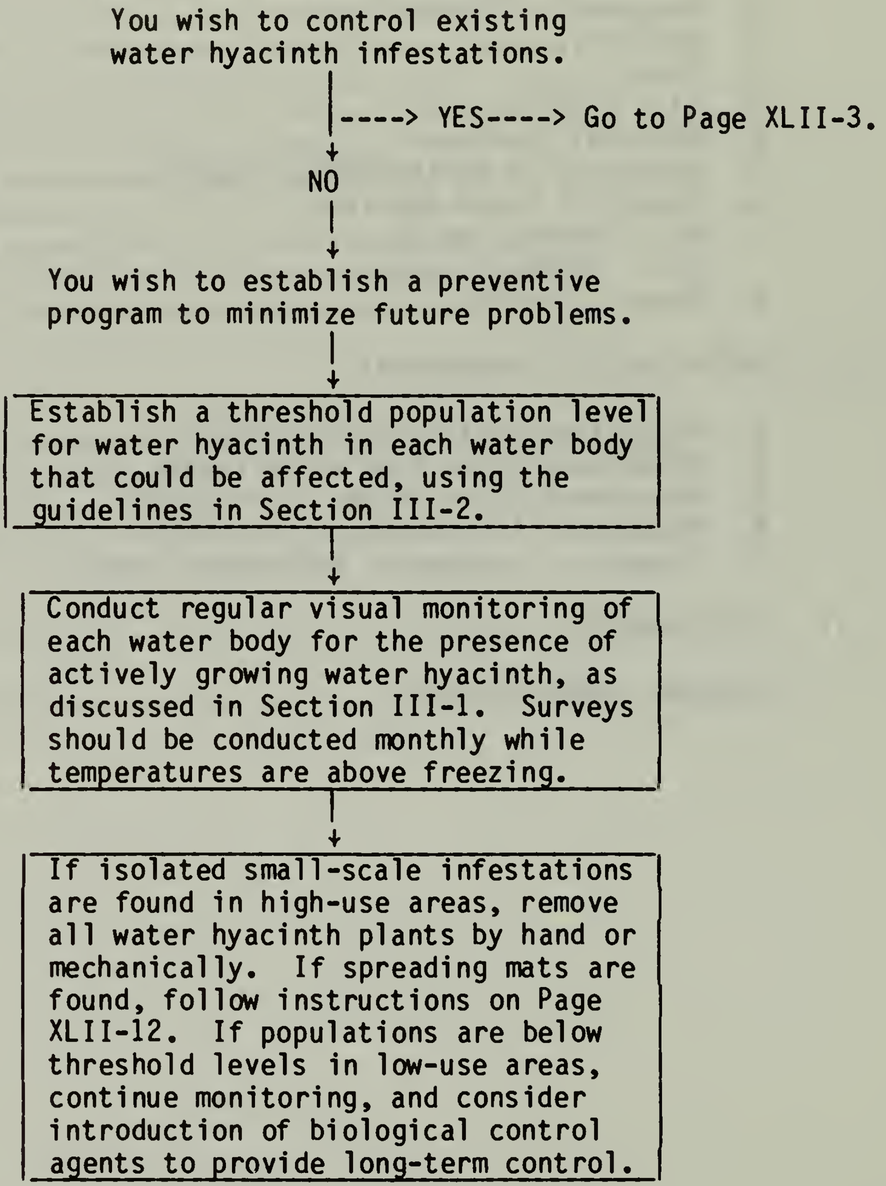
CONTENTS

I. WATER HYACINTH IPM DECISION TREE

These recommendations represent an approach of minimal pesticide use to maintain pest populations below injurious levels. If additional actions are necessary, consult further with NPS Pest Management Staff. All use of pesticides must conform to Environmental Protection Agency label instructions and be approved on an annual basis by the Director, NPS.



You wish to control existing water hyacinth infestations.

----> YES----> Go to Page XLII-3.

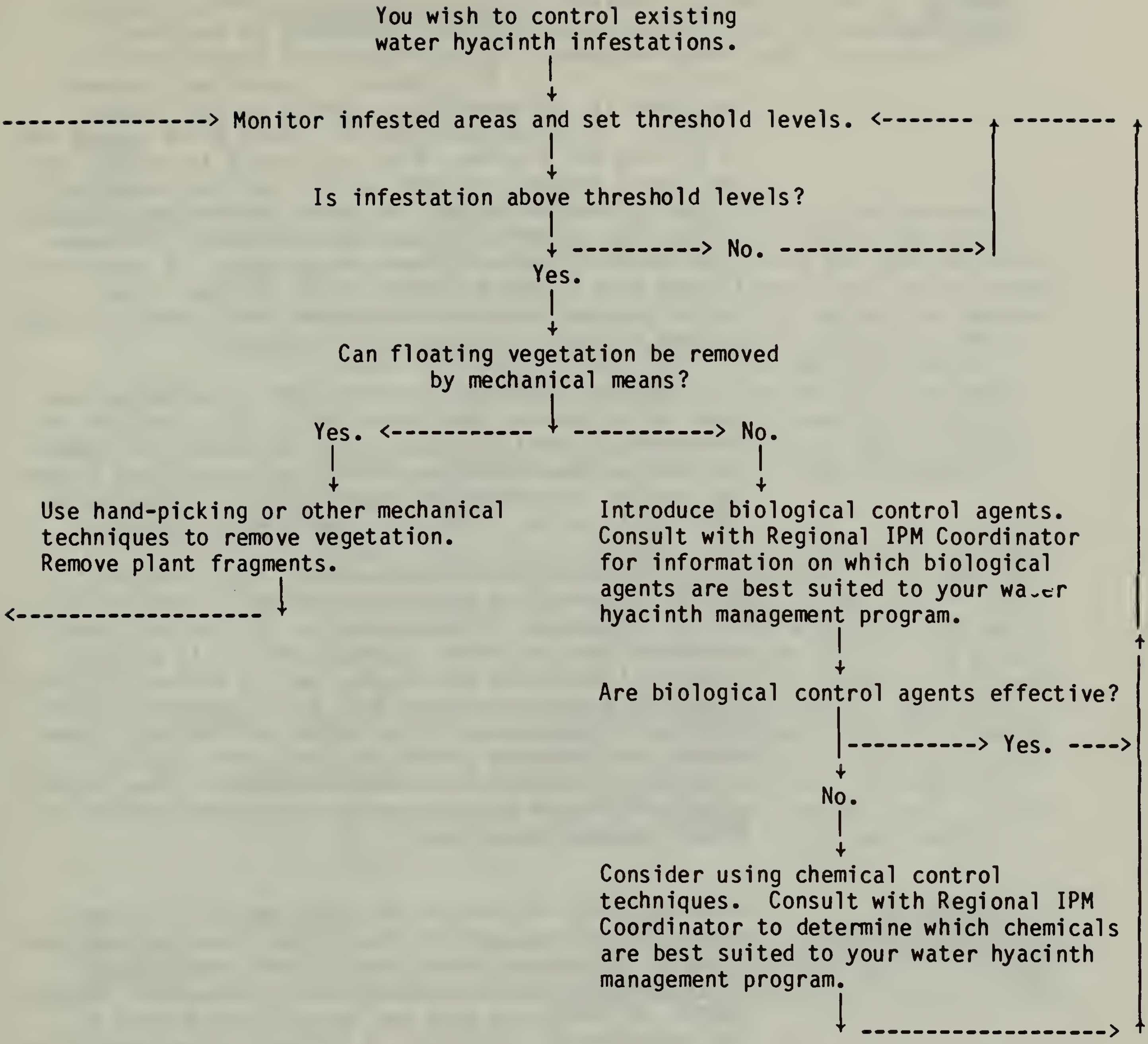
NO

You wish to establish a preventive program to minimize future problems.

Establish a threshold population level for water hyacinth in each water body that could be affected, using the guidelines in Section III-2.

Conduct regular visual monitoring of each water body for the presence of actively growing water hyacinth, as discussed in Section III-1. Surveys should be conducted monthly while temperatures are above freezing.

If isolated small-scale infestations are found in high-use areas, remove all water hyacinth plants by hand or mechanically. If spreading mats are found, follow instructions on Page XLII-12. If populations are below threshold levels in low-use areas, continue monitoring, and consider introduction of biological control agents to provide long-term control.



You wish to control existing water hyacinth infestations.

Monitor infested areas and set threshold levels.

Is infestation above threshold levels?

No.

Yes.

Can floating vegetation be removed by mechanical means?

Yes.

No.

Use hand-picking or other mechanical techniques to remove vegetation. Remove plant fragments.

Introduce biological control agents. Consult with Regional IPM Coordinator for information on which biological agents are best suited to your water hyacinth management program.

Are biological control agents effective?

Yes.

No.

Consider using chemical control techniques. Consult with Regional IPM Coordinator to determine which chemicals are best suited to your water hyacinth management program.

II. WATER HYACINTH BIOLOGY AND ECOLOGY

1. *Species Described*:

Water hyacinth (*Eichhornia crassipes* [Mart.] Solms) is a floating aquatic monocot belonging to the family Ponterderiaceae. The water hyacinth plant varies in size from 1" to 4' in diameter. A submerged rhizome and crown is surrounded by a rosette of dark green, leathery, circular to elliptical leaves which may be up to 8" long and 6" wide. Each leaf stalk is spongy, and may have a bulbus, expanded central portion, especially in small plants or plants in sparse populations (Aurand, 1982). The growth of water hyacinth shoots is monopodial (shoots branch off from a main stem). A branching, fibrous root system arises from the rhizome. The feathery-appearing roots may extend 6-24" below the water surface, but do not contact the hydrosoil.

Water hyacinth produces large numbers of submerged stolons, which produce daughter plants. In a suitable environment, actively-growing plants may double in number every two weeks (Gangstad, 1978), producing large floating mats of interconnected plants which can completely cover small ponds, lakes, or slow-flowing streams.

An upright stalk containing several light blue to violet flowers 2" in diameter is produced from the center of each plant. Flowers consist of 6 petals, the uppermost of which bears a yellow patch. In tropical areas pollination is by insects, but in subtropical and temperate areas such as the U.S. *E. crassipies* is self-pollinated (Gangstad, 1978). After pollination, flower spikes bend at three points, so that seed capsules develop and release their seeds underwater. See Tarver, *et al*. (1979) for photographs and additional details of water hyacinth morphology.

2. *Geographic Distribution:*

Water hyacinth is a native of South America. It was reportedly introduced into the United States at the New Orleans, LA, Cotton Exhibition of 1884, where plants imported from Venezuela were given as souveniers (Aurand, 1982). By 1900, the weed had spread through most of the waterways of Louisiana. A visitor to the exhibition took several plants to Florida, for use as ornamentals, and by 1949, 63,000 acres of Florida water were covered by water hyacinth. Other states in which the weed occurs are Alabama, California, Georgia, Hawaii, Mississippi, North Carolina, South Carolina, Texas, and Virginia.

3. **Habitat:** Water hyacinth is a free-floating plant which lives in standing or slowly moving fresh water. It requires relatively high light levels and above-freezing temperatures. Wind and currents may move the weed throughout a water body, or from one water body to another.

4. **Hosts:** Not applicable.

5. **Life Cycle:** Water hyacinth reproduces both sexually and asexually in the U.S. Self-fertilization of flowers can result in the production of 45,000,000 seeds per acre of water hyacinth. However, only 5% of those seeds may germinate (Gangstad, 1978), due to infertility and/or the absence of proper conditions for germination (Barrett, 1980). Seeds are released underwater, and may fall to the hydrosoil or remain trapped in the roots of the parent plant. Fertile seeds may remain viable for up to 7 years, but only will germinate after an exact process of drying and rewetting has taken place (Wolverton, no date).

   Once a plant is established, it is capable of extremely rapid growth under favorable conditions. The most effective and common means of spread in the U.S. is the production of daughter plants from underwater stolons growing from the crown of the parent plant. One plant can produce up to 300 interconnected offspring in less than a month. In a single growing season, 10 plants could produce enough offspring (about 600-700 thousand) to completely cover an acre of water surface (United States Army Corps of Engineers, 1973). Such growth results in the formation of water hyacinth mats, which can spread at an average rate of 2 feet each month (Aurand, 1982).

6. **Seasonal Abundance:** Water hyacinth attains maximum standing crop in May and June, but will continue to grow as long as air and water temperatures remain favorable (Center and Spencer, 1981).

7. **Responses to Environmental Factors:** Water hyacinth is the fastest growing plant known to man (Wolverton and McDonald, 1977). It can grow in water ranging in temperature from 53°F to 100°F, but optimum growth occurs within the range 71°F to 95°F (Knipling et al., 1970). Seed germination is optimal at water temperatures of 82°F to 97°F, and is retarded at water temperatures below 50°F. Water hyacinth plants can be killed by repeated exposure to subfreezing temperatures. These plants have a high light

requirement, and cannot grow well under forest canopies. Growth is poor in acid, soft, or saline (over 0.6 ppt) water. Water hyacinth plants may be spread by wind and/or water currents. Broken stems or crowns are capable of regrowth, so that mechanical damage may actually increase the plant population.

8. Impact of Water Hyacinth:

8.1 Direct Impact: The major impact of water hyacinth is the clogging of water bodies by floating mats of weeds, reducing the usefulness of the water for swimming or boating. Since water hyacinth is such a rapid grower, it may displace or eliminate desirable plant species. The root systems of water hyacinth may remove large quantities of nutrients from the water in which they grow.

8.2 Indirect Impact: The major indirect impact of water hyacinth is the displacement of animals and plants from infested waters, due to overgrowth of the weed. In addition, mosquitoes or other insects of public health or nuisance importance may breed in the water hyacinth mats. Water hyacinth may also cause the suppression of other undesirable aquatic weeds (e.g., alligatorweed or waterlettuce) (Aurand, 1982).

Water hyacinth removes nutrients, pesticides, and heavy metals from the water. NASA uses water hyacinth in its sewage treatment lagoons in Bay St. Louis, Mississippi, and has helped to establish wastewater treatment programs using water hyacinth in numerous localities throughout Mississippi, Louisiana, Florida, and in San Diego, California (W. Wolverton, personal communication). In these treatment systems, water hyacinth greatly improves the quality of the effluent (Wolverton and McDonald, 1979a; McDonald and Wolverton, 1980). The overgrowth is harvested and can be used for compost, human and animal food, and for the generation of biogas to produce electricity (Wolverton and McDonald, 1979b; Wolverton and McDonald, 1981).

9. Natural Enemies:

A. Insects -

1. Water hyacinth weevils - *Neochetina eichhorniae* Warner (the mottled water hyacinth weevil) and *N. bruchi* Hustache (the chevroned water hyacinth weevil) are native to Argentina, Bolivia, and Trinidad. Adult beetles are nocturnal feeders that produce 1/8" feeding

scars (on leaf blades and petioles) in which eggs are laid. Eggs may also be laid in injured leaf tissues. Larvae mine within the petioles during their five developmental instars, then migrate to submerged roots to pupate. Adults begin feeding immediately after emerging. The tunneling and feeding activities of the insects may completely kill stems and leaves. In addition, pathogenic microbes infect plant tissues exposed by larval tunneling, often resulting in leaf death or abcission. These insects have been introduced into the U.S. as biocontrol agents (Theriot, 1982)(see Section III.3).

2. Water hyacinth moth - Newly hatched larvae of the Argentine pyralid moth _Sameodes albiguttalis_ (Warren) feed on the surfaces of leaves and petioles, creating irregularly shaped lesions. Older larvae burrow into petioles (especially inflated petioles) and feed internally. Water accumulates in the damaged area, leading to waterlogging and submerging of infested leaves. Newly hatched larvae begin feeding just below the epidermis (outer layer) of the petioles, and feeding damage becomes evident after 1-2 days. Some may burrow into the youngest petiole and excavate the end of the rhizome, destroying the apical bud, halting development of the shoot. Fifth instar larvae may damage several petioles, exiting and entering at contiguous petiole bases (Center, 1981). This species has been released in the U.S. as a biocontrol agent (See Section III.3.B.1).

3. _Arzama densa_ - The native American noctuid moth _A. densa_ (Walker) attacks water hyacinth and pickerelweed. Larvae tunnel into petioles and crowns, producing extensive feeding damage (Cofrancesco, 1982). Effective biocontrol of water hyacinth has not been achieved using _A. densa_, due to the presence of parasites which attack larvae during the later (4-7) instars (Cofrancesco, 1982), but the moth is being considered for use in Hawaii where it could be introduced free of parasites (E. Theriot, personal communication).

5. Water hyacinth mite - The mite _Orthogalumna terebrantis_ Wallwork attacks water hyacinth in Florida and Louisiana, and may have been introduced into the U.S. with the weed (Del

Fosse, 1978). The mite bores feeding galleries beneath the epidermis of leaves, causing moderate to severe damage. The biocontrol potential of this species in the U.S. is not known, but Sanders *et al.* (1982) reported only slight impact on infested plants in Panama.

B. Fungi - *Cercospora rodmanii* Conway - This fungus was isolated from diseased water hyacinth leaves in a reservoir in Florida in 1973. The pathogen causes symptoms ranging from small, dark spots on the leaf blade or petiole to destruction of the entire leaf and petiole. *C. rodmanii* is specific for *E. crassipes*. Abbott Laboratories, Chicago, IL, is developing a commercial formulation of the fungus (Pennington and Theriot, 1983). Theriot (1981) has reported successful control of *E. crassipes* using this agent.

C. Mammals - The manatee (*Trichechus manatus* L.) is a large (up to 15 feet long and 1300 lbs in weight), roughly torpedo-shaped, slow-moving, social aquatic mammal commonly known as the "sea cow." Manatees live in warm, shallow coastal waters of Florida and range from Texas to southern North Carolina. Manatees are listed as an endangered species in the U.S., and are relatively rare even in Florida, where there are only about 1000 individuals. They feed on aquatic plants, consuming as much as 100 lbs of vegetation each day. Water hyacinth is a favored food of manatees. In the U.S., research was conducted in the 1960's on the use of manatees to control aquatic weeds, but the status of the manatee as an endangered species has made it doubtful that they will be practicable biocontrol agents (Blackburn and Andres, 1968; Gluckman, 1983; McGehee and Zeiger, 1977). However, aquatic weed managers should keep in mind the potential effects on water hyacinth of manatees in any areas where they occur, and the influence of water hyacinth control measures on manatee populations.

III. WATER HYACINTH MANAGEMENT

1. **Population Monitoring Techniques:** The only effective technique for monitoring water hyacinth population levels is periodic visual inspection of water bodies for the presence of water hyacinth plants. Surveillance programs are based on remote sensing and/or ground reconnaissnace. False-color infrared aerial photography is highly effective in water hyacinth survey programs (Leonard, 1982). The photographs are taken at a scale of 1:12000 in the spring and fall of each year. The distribution of water hyacinth is traced onto transparent base maps and their area of coverage calculated. The interpretation of false-color infrared photographs requires trained personnel.

2. **Threshold/ Action Population Levels:** Standardized threshold levels for water hyacinth populations have not been formulated. The Army Corps of Engineers, Environmental Protection Agency, and state and local agencies concerned with water hyacinth management base their treatment decisions on a cost/ benifit analysis. A unique threshold level must be established for each water body, based on considerations such as the type and size of the water body (site), the activities (e.g., fishing, swimming, boating) which occur at the site, the numbers and types of desirable flora and fauna inhabiting the site, and so on. Such information can be correlated with inspection data (e.g., population levels and conditions, extent of coverage) to produce the threshold and action population levels for each water body. In general, high-use aquatic sites (e.g., swimming beaches, boat docks) will be more sensitive to water hyacinth infestations than will low-use sites (e.g., wildlife preserve or shore-only fishing areas). In addition, at sites where the weed population is under attack by predators or parasites, allowing natural controls to operate may produce more effective long-term control than would the application of additional control measures. In such areas, a higher threshold level may be beneficial.

   The U.S. Environmental Protection Agency has developed a computerized "expert system" for the determination of threshold levels and the design of programs for water hyacinth management in water bodies in several Southeastern states (Rodgers et al., 1983). Use of the system requires the input of information concerning the location and type of the affected water body, available water quality data, water uses, growing season data, infestation area, and the known or reported effects of the infestation on water body uses (Anonymous, 1983). The system ("Decision Matrix for Integrated Control of Aquatic Weeds") is currently useable on Apple III® computers. For information concerning the availability of the system, contact:

Charles D. Reese
Office of Pesticide Programs
U.S. Environmental Protection Agency
Washington, D.C.

3. <u>Management Alternatives - Nonchemical:</u>

A. Mechanical harvesting - Mechanical harvesting is recommended for the following situations:

1. Where shallow zones of ponds or lakes are covered by dense stands of weeds;
2. When the use of other methods is undesirable because of potential adverse impacts on water uses, animals, or plants;
3. Where nuisance weeds are resistant to herbicides;
4. Where nutrient loading from decomposing weeds left in the water could promote eutrophic conditions; and
5. In small lakes, ponds, or embayments (less than 100 acres).

Mechanical harvesting may not be beneficial where internal obstructions in the water body would impair harvesting, where shallow areas would be disturbed by the procedures, or where weed fragments could be dispersed by currents or wind, compounding the weed problem (Rodgers <u>et al</u>., 1983). A disadvantage of mechanical harvesting is its high cost relative to the use of chemical or biological control measures (Canellos, 1981).

Mechanical harvesting methods include:

1. Hand removal - Small-scale infestations can be eliminated by handpicking the weeds, which can then be transported to an on-shore disposal site. While this technique is labor intensive, it could be incorporated into the survey process, allowing small infestations to be eliminated as soon as they are discovered.

2. Harvesting machines - Mechanical harvesting machines generally consist of a boat-mounted reciprocating cutting and collecting system, which may feed cut material into a conveyor for shore dumping, or may throw cut material onto the shore, or may not collect material (in this case, an additional boat with rakes or other collecting devices is required). Harvesters cost $6,000 to $170,000 (1982 basis), and are available from the following manufacturers:

   a. Aquamarine Corp.
      Box 616
      Waukesha, WI 53186

   b. Altosar Aquatic Weed Harvesters
      3147 Losey Blvd.
      LaCrosse, WI 54601

   c. Hickney Co.
      913 Cogswell Drive
      Silver Lake, WI 53170

   d. Limnos, Ltd.
      22 Roe Ave.
      Toronto, Ontario, CANADA

   e. Mudcat Division
      National Car Rental Co.
      P.O. Box 16247
      St. Louis Park, MN 55416

   f. Mariner Water Weed Harvesters
      104 Locust St.
      Polmyra, WI 53156

B. Biological Control -

1. Insects - The exotic water hyacinth weevils _Neochetina bruchi_ and _N. eichhorniae_ have been released at numerous sites in the U.S. and appear to be spreading throughout most of the range of the plant (Gangstad, 1978 and personal communication). The insects can be obtained from the Aquatic Plant Operations Support Center (APOSC), U.S. Army Corps of Engineers, P.O. Box 4970, Jacksonville, FL 32201. APOSC does not charge for control agents (except for shipping and handling fees). The Argentine water hyacinth moth (_Sameodes albiguttalis_) has been released and is established in southern Florida (Center, 1982) and Louisiana (Aurand, 1982). Field studies

of the effectiveness of this moth as a biocontrol agent are ongoing. For information on the efficacy and availability of S. albiguttalis, contact:

Ted D. Center
USDA Aquatic Plant Management Lab
3205 SW College Ave.
Ft. Lauderdale, FL 33314.

2. Pathogens - While numerous fungal pathogens of water hyacinth have been found in worldwide searches, to date only the native species Cercospora rodmanii has been found to be an effective biocontrol agent in large-scale field tests (Pennington and Theriot, 1983). For further information on this agent, contact:

Edwin Theriot
U.S. Army Corps of Engineers
Waterways Experiment Station
Vicksburg, MS 39180.

3. Integrated Methods - Perkins (1977) found that water hyacinth plants treated with 2,4-D became more attractive to mottled weevils, increasing control. The effectiveness of herbicides against water hyacinth was found to be greater in plants attacked by the fungus Acremonium zonatum (Perkins, 1974).

4. Management Alternatives-Chemical: Several herbicides are registered for control of water hyacinth. Consult your regional IPM Coordinator to determine which, if any, of these herbicides is best suited for inclusion in your water hyacinth management program.

5. Summary of Management Recommendations:

A. Monitor all water bodies likely to be infested with water hyacinth, and set thresholds for each water body.

B. Where feasible institute mechanical harvesting techniques. Remove plant fragments to prevent regrowth.

C. Consider the introduction of biocontrol agents to provide long-term management of chronic infestations. Consult with your Regional IPM Coordinator or local

United States Army Corps of Engineers District personnel regarding the feasibility of biocontrol techniques in your water hyacinth management program.

C. Consider the use of a registered herbicide to provide rapid reduction of severe infestations. Consult with your Regional IPM Coordinator to determine which chemical, if any, is recommended for your water hyacinth management program.

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