ELEMENT STEWARDSHIP ABSTRACT
for

Sorghum halepense

Johnson Grass

To the User:

Element Stewardship Abstracts (ESAs) are prepared to provide The Nature Conservancy's Stewardship staff and other land managers with current management-related information on those species and communities that are most important to protect, or most important to control. The abstracts organize and summarize data from numerous sources including literature and researchers and managers actively working with the species or community.

We hope, by providing this abstract free of charge, to encourage users to contribute their information to the abstract. This sharing of information will benefit all land managers by ensuring the availability of an abstract that contains up-to-date information on management
techniques and knowledgeable contacts. Contributors of information will be acknowledged within the abstract and receive updated editions. To contribute information, contact the editor whose address is listed at the end of the document.

For ease of update and retrievability, the abstracts are stored on computer at the national office of The Nature Conservancy. This abstract is a compilation of available information and is not an endorsement of particular practices or products.

Please do not remove this cover statement from the attached abstract.

Authors of this Abstract:
Dara Newman

The Nature Conservancy
Element Stewardship Abstract
For *Sorghum halepense*

I. IDENTIFIERS

Common Name: JOHNSON GRASS Global Rank: G?

General Description:
Perennial grass with culms reaching up to 3 m high and a purplish panicle up to 50 cm in length.

Pest/Weed Considerations:
Sorghum halepense is considered to be one of the ten worst weeds in the world (Holm et al. 1977). Fifty-three countries, ranging in latitude from 55 N to 45 S report Johnson grass as a major problem; the problem is most serious in the region from the Mediterranean to the Middle East and India, Australia, central South America and the Gulf Coast of the United States (Holm et al. 1977).

Diagnostic Characteristics:
The distinguishing characteristics of Sorghum halepense are the ribbed leaf sheath, the conspicuous midrib, the large, purplish panicle and the extensive rhizome system. Panicum bulbosum, which has been confused with Sorghum halepense, can be recognized by its short, knotty rhizomes and bulbous swellings at the base of the culms.

II. STEWARDSHIP SUMMARY

Sorghum halepense is an invasive and tenacious weed which thrives in disturbed soils. The prolific seed production, extensive rhizome system, sprouting ability of fragmented rhizomes and ability to grow in a wide range of environments make Johnson grass difficult to control. The best time to implement control techniques is during the first two weeks of growth when new rhizome development has not yet begun and when the carbohydrate supply is at its lowest concentration. During the fall the rhizome carbohydrate levels are again low, due to the formation of over- wintering rhizomes, making this an appropriate time for herbicide application. A combination of mowing, tilling, and herbicide applications may provide adequate control of Sorghum halepense and may produce better effects than just one technique alone. Once successful control has been reached, a rapid re-vegetation project should be implemented for the establishment of native plants. If transplants are to be used, plants should be grown during the eradication period (Newman 1989). Subsequent spot control of remaining Johnson grass, that avoids jeopardizing the native plants, may be necessary during the subsequent years to fully eradicate this weed.

III. NATURAL HISTORY

Range:
Sorghum halepense is a cosmopolitan weed thought to be native to the Mediterranean region (Holm et al. 1977), but with controversy over its origin. It was introduced to the United States in the early 1800s as a potential forage crop. By the end of the 19th century Johnson grass was growing throughout most of the United States (McWhorter 1981). New ecotypes have evolved allowing the species to expand its range. The common name originates from farmer Johnson who introduced it into Alabama from South Carolina in 1840 (Warwick and Black 1983).

Habitat:
In the United States Sorghum halepense grows in disturbed lands. As a weed, this plant is found in ditches, cultivated fields and wastelands throughout Arizona, commonly growing below 5,000 feet elevation in irrigated croplands (Gould 1951).

Sorghum halepense is adapted to a large range of conditions. An ideal environment for Johnson grass is the subtropics: warm and humid with summer rainfall. Most of the ecotypes are frost sensitive; the aerial portions are killed during winter freezes. Temperatures below 13 C tend to inhibit flowering (Holm et al. 1977). Cold tolerant ecotypes have been found growing in northern United States and southern Canada (Warwick and Black 1983). Rhizomes are also intolerant of high temperatures; rhizomes exposed to 50 C to 60 C (122 F to 140 F) temperatures for several days died (Warwick and Black 1983).

Johnson grass is adapted to a wide variety of soil types, however fertile porous soils support larger plants than poorly drained clay soils (Warwick and Black 1983). Soils with a pH of between 5 and 7.5 are ideal for Johnson grass. Experiments using flooding as a control measure revealed that aeration is not critical for survival of rhizomes or seeds (Horowitz 1972a).

Most land-types, especially disturbed and flooded bottom lands, are susceptible to this tenacious and invasive weed (McWhorter 1981). The massive creeping rhizome system and the abundance of dormant seeds protect this plant from severe conditions. Rhizomes have been found 120 cm deep in cultivated soil and five year old seeds displayed 50% viability (Warwick and Black 1983), making complete eradication of this species very difficult. Once the environmental conditions improve these dormant structures are available to resume growth and invade adjacent land. A single Johnson grass plant produces 200 to 300 feet of rhizomes in one month and 10 bushels of seed can be produced on one acre in a single growing season (McWhorter 1981).

Ecology:
GENERAL LIFE-CYCLE: The life-cycle of Sorghum halepense is conducive to its survival in a wide range of environmental conditions (Holm et al. 1977, Monaghan 1979, Warwick and Black 1983). Growth from the apical or axillary nodes on the primary rhizomes which have survived the winter begins as temperatures increase in the spring. The secondary structure, the annual above- and below-ground growth, uses the stored

carbohydrates from the primary rhizome to get a rapid, early start in growth (Holm et al. 1977). The development of rhizome spurs and secondary tillers begins a month after growth has resumed when approximately six leaves are present. Depending on the climate, flowering begins roughly two months after growth commences and continues throughout the growing season. Most of the year's rhizome growth takes place after flower production (Warwick and Black 1983), however, no causal relationship exists between the two (Horowitz 1972b, Monaghan 1979). The tertiary rhizomes which grow deep into the soil (as deep as 120 cm) survive the winter and become the following season's primary structure (Holm et al. 1977, Warwick and Black 1983). The remains of the primary rhizomes from the previous year's growth decay as the temperature begins to drop (Holm et al. 1977).

Hundreds of seeds are produced on each panicle throughout the summer flowering period (Monaghan 1979, Warwick and Black 1983). Seedlings emerge later and grow slower than rhizome sprouts, but the pattern of development is similar between the two structures (Warwick and Black 1983).

SEXUAL DEVELOPMENT: Self-compatibility, immense seed production, effective dispersal techniques, seed dormancy and seed longevity are features which make Sorghum halepense a prolific weed. Most members of the genus Sorghum are self-compatible (Warwick and Black 1983). Johnson grass plants growing less than 130 m apart will cross-fertilize. However, less than 5% of fertilized plants are the result of crossing, even in fields where plants are closely spaced (Warwick and Black 1983). The self-compatibility insures seed production throughout the growing season.

Plants begin to flower approximately two months after growth commences. The exact flowering time depends on temperature, plant vigor and photoperiod (Horowitz 1972b, Warwick and Black 1983). Johnson grass is a short-day plant (Ghersa et al. 1985). Experiments in Mississippi resulted in flowering of all treatments, ranging from 8 to 16 hours of light. However, seedhead formation was inhibited in the 16 hour treatment and reduced in the 14 hour treatment. The 10.5 and 12 hour photoperiod treatments resulted in the greatest amount of seed production (Warwick and Black 1983). Temperatures below 13 C to 15 C (55 F to 59 F) inhibit floral production (Warwick and Black 1983). In Arizona flowering begins in April and continues through November (Kearney and Peebles 1951).

Seed production is prolific. Depending on the ecotype, anywhere from 37 to 352 seeds can form on a panicle (Warwick and Black 1983). Seeds form only in the sessile spikelets, with a maximum of one seed per spikelet (Warwick and Black 1983). In Israel, a single plant produced 28,000 seeds (84 g) in one growing season, and in Mississippi the average plant produced 1.1 kg of seeds per season (Horowitz 1972b, Warwick and Black 1983).

Shattering of the seeds from the spikelets enhances seed dispersal (Holm et al. 1977). Seeds that land in a water source may be carried, possibly quite far, to new sites. Other modes of dispersal include wind, livestock and contaminated machinery, grain or hay.

Seeds pass unharmed through birds and cattle (Holm et al. 1977, Warwick and Black 1983).

Seed dormancy and longevity provide a source of seed in the soil which is ready to germinate when unfavorable conditions subside. The seed dormancy properties are dependent on the ecotype (Monaghan 1979). Due to the mechanical reduction of water penetration by the tannins in the seed coat, seeds will not germinate naturally when first wetted (Warwick and Black 1983). Less than 10% of seeds from 43 ecotypes germinated at 20 C (68 F) (Monaghan 1979). However, when the seed coat is removed or if the seed is treated with sulfuric acid and then exposed to temperatures greater than 20 C, the seed will germinate (Monaghan 1979, Warwick and Black 1983). Five months in dry storage at room temperature overcomes most dormancy (Monaghan 1979). The longevity of seeds varies depending on the environmental conditions in which the seed was stored. Seeds stored in the laboratory under dry conditions remained viable for over seven years. Studies in California showed a 50% viability in seeds stored for five years, however another study resulted in only 2% viability in seeds which remained in the soil for six years (Warwick and Black 1983). Seeds buried in the soil for two and a half years displayed a 60% to 75% viability (Warwick and Black 1983).

Most seeds do not germinate the year they are produced, but germinate readily the following year (Holm et al. 1977). The soil type and seed depth influence germination; the deeper a seed is buried and the greater the clay content of the soil the less likely a seed will be to germinate (Warwick and Black 1983). The majority of seedlings arise from seeds in the top 7 cm of the soil, but seeds as deep as 15 cm can germinate (Warwick and Black 1983). Seeds require temperatures 10 C higher to germinate than do rhizomes to sprout (Horowitz 1972b). The light conditions affect the temperature requirements for germination. The highest germination rate is in seeds pre-chilled for 20 days at 10 C (50 F) and then exposed to alternating temperatures of 24 C and 40 C (75 F and 104 F) with continuous light (Warwick and Black 1983).

VEGETATIVE DEVELOPMENT: The prolific seed production ability of Johnson grass accounts for the dispersion of this invasive weed, but the immense, rapidly growing rhizome system gives Sorghum halepense its competitive edge against other species. The extensively creeping rhizomes, which regenerate easily when cut into small pieces, are capable of growing or remaining dormant in a wide range of environmental conditions; these qualities make it exceptionally difficult to eradicate Sorghum halepense.

The existence of physiological dormancy in Johnson grass' rhizomes is controversial (Monaghan 1979). However, apical dominance in the rhizome system prevents axillary buds from sprouting. Nodes furthest from the apex have a reduction in apical suppression (Hull 1970). The importance of this in the survival of the plant should not be overlooked. At all times, including during favorable conditions, the majority of rhizome buds will not be growing, thus the application of herbicides will have little to no effect on the viability of these inactive regions.

Primary rhizomes provide stored energy for an initial rapid growth period in the spring. No causal relationship between tillering, flowering and rhizome formation is found (Horowitz 1972b, Monaghan 1979). New rhizomes may grow either during or after tiller formation and either before or after flower production (Horowitz 1972b), although ninety percent of the year's rhizome production occurs after flowering (Warwick and Black 1983). A decrease in the percent of the plant's total fresh weight in shoots and roots and an increase in the percentage of the plant's fresh weight as rhizomes occurs from April through August (Warwick and Black 1983).

Rhizome production can be immense depending on soil type, light and temperature. Sixty to 90 m of rhizomes were produced per plant in one growing season in Mississippi (Warwick and Black 1983). A 600-fold increase in the rhizome length occurred in 120 days starting with a 7.5 cm section (McWhorter 1981).

The depth of the rhizomes depends on the heaviness of the soil (Monaghan 1979). Cultivated soils and soils with a low percentage of clay are more likely to have deep rhizomes than high clay- content soils; rhizomes have been found 120 cm deep in cultivated soils, but the majority of rhizomes are found in the top 20 cm of soil (Holm et al. 1977). Longer rhizome segments (20 cm long) started growing 20 days earlier and had more rapid growth than segments half their size. Both depth and size of the rhizome are important in determining growth patterns. Usually, the bigger the segment the deeper it can be located in the soil while still successfully producing shoots (Warwick and Black 1983).

Since carbohydrate production depends on photosynthesis it follows that a larger amount of rhizomes may be produced at greater light intensities (Monaghan 1979). Temperature has a major effect on rhizome production, due to both photosynthetic rates and to physiological factors at temperature extremes. Variations in the effects of temperature on rhizome growth exist between ecotypes (Holm et al. 1977). In Israel, the minimum temperature for rhizome production was 15 C and the maximum was 30 C (Horowitz 1972b). Similar temperature requirements were found by Hull (1970) in Rhode Island, the minimum temperature was 15 C, however the optimum temperature was 30 C (the maximum temperature was not given).

Most ecotypes have rhizomes that cannot tolerate freezing temperatures or hot drying conditions (Monaghan 1979, Warwick and Black 1983). Unlike most perennial grasses which store carbohydrates in the form of fructosans, Johnson grass stores starch and sucrose; this may partially explain the lack of cold tolerance in Johnson grass as compared to other perennial plants (Monaghan 1979). In the laboratory, rhizome buds were killed by exposures to -3 C (27 F) for 24 hours and in the field 20 cm deep rhizomes were killed at temperatures below -9 C (16 F) (Warwick and Black 1983). Adaptation and the formation of new ecotypes account for the geographic spread of Johnson grass in northern U.S. and southern Canada. From 1959 until 1977 Johnson grass died during the cold winters of Canada; in 1977 the first vegetative structure survived the winter from a newly evolved cold tolerant ecotype (Alex et al. 1979).

Most Johnson grass is intolerant of high temperatures. Three days of 55 C (131 F) kills most rhizome buds. Heat and drying has a synergistic effect; seven days of desiccation at 30 C (86 F) results in rhizome death (Warwick and Black 1983). Six days of drying at 14 C to 54 C (57 F to 130 F) results in a 78% weight loss and a cessation of sprouting of the nodal buds (Horowitz 1972a). Johnson grass exhibits greater damage from desiccation than do CYNODON DACTYLON or CYPERUS ROTUNDUS.

CARBOHYDRATE CYCLE: The temporal pattern of the levels of fructose, glucose, sucrose and starch in the rhizomes are similar, although the individual concentrations are unrelated (Horowitz 1972d, McWhorter 1974). Sucrose and starch are the primary forms of carbohydrate storage in the rhizomes, and glucose is the principle reducing sugar in the developing leaves (McWhorter 1974). The concentration of total soluble carbohydrates in the rhizomes decreases during the first three weeks after germination or sprouting, reaching the lowest concentration of the year between 10 and 30 days after emergence; this occurs with a concurrent increase in glucose level in the developing leaves (McWhorter 1974). An increase in the rhizome carbohydrate level begins following the development of the first few leaves and peaks during flower maturation; this is followed by a sharp decrease in the carbohydrate level and then a gradual accumulation of carbohydrates in the late fall (McWhorter 1974). Rhizomes reach a maximum weight following high carbohydrate levels during the period of seed maturation (Oyer et al. 1959). The annual double cycle of the rhizome carbohydrate level, high in early winter and early summer and low in early spring and fall, results in a stored energy supply during the winter that allows for early spring growth followed by a replenishment of carbohydrates during the summer for fall rhizome growth (Horowitz 1972d).

Reproduction:
SEXUAL DEVELOPMENT: Self-compatibility, immense seed production, effective dispersal techniques, seed dormancy and seed longevity are features which make Sorghum halepense a prolific weed. Most members of the genus Sorghum are self-compatible (Warwick and Black 1983). Johnson grass plants growing less than 130 m apart will cross-fertilize. However, less than 5% of fertilized plants are the result of crossing, even in fields where plants are closely spaced (Warwick and Black 1983). The self- compatibility insures seed production throughout the growing season.

VEGETATIVE DEVELOPMENT: The prolific seed production ability of Johnson grass accounts for the dispersion of this invasive weed, but the immense, rapidly growing rhizome system gives Sorghum halepense its competitive edge against other species. The extensively creeping rhizomes, which regenerate easily when cut into small pieces, are capable of growing or remaining dormant in a wide range of environmental conditions; these qualities make it exceptionally difficult to eradicate Sorghum halepense. (Also see section on Ecology.)

Impacts:

COMPETITIVE RELATIONSHIPS: The extensive rhizome system and prolific seed production make Sorghum halepense highly competitive and invasive. The massive size (up to 3 m tall (Gould 1951) of this plant creates difficulties for the establishment of other plants. The rapid growth of Sorghum halepense's rhizomes also provides the plant with a competitive edge over other species; the plant directly shades other plants, decreases nutrient and moisture availability to other plants, and possibly inhibits the growth of other plants via the production of allelopathic chemicals (Holm et al. 1977, Warwick and Black 1983).

Sorghum halepense plants grown free from competition of other plants spread by growth of the rhizomes followed by the production of aerial shoots arising at increasing distances from the original plant (Horowitz 1973). Tiller formation occurs after the development of the first shoot, resulting in clumps of Johnson grass. The area of Johnson grass increases 1.3 m2 per month resulting in patches 17 m2 in two and a half years from single Johnson grass sprigs (Horowitz 1973).

Crowding of Johnson grass results in intraspecific competition. A delay in rhizome and panicle formation, and a reduction in total final dry weight results as the density increases from one to twenty plants per 20 cm pot (Williams and Ingber 1977). Shoot growth is affected more than rhizome growth when the plants are stressed. The initial rhizome growth rate, but not the final size of the rhizomes, is affected by crowding. This quality, which accounts for the exceptional competitive ability, could result from an accelerated transfer of photosynthates from the shoot to the rhizomes. Although delayed, flower and seed development occurs even in the most crowded conditions, indicating a significant partitioning of photosynthates to insure reproductive success (Williams and Ingber 1977).

In addition to Sorghum halepense's competitive ability is the production of allelopathic substances. Friedman and Horowitz (1970) showed a retardation effect in barley, mustard and wheat as well as in other Johnson grass plants by both an exudate from Johnson grass and decaying plant matter. An allelopath from Johnson grass, dhurrin, inhibited seed germination and seedling development in several plant species (Warwick and Black 1983). The significance of the allelopath in the field is unknown.

The tremendous invasive potential of Sorghum halepense stems from several features. The extensive over-wintering rhizome system provides an available energy source for growth early in the season. This early-season growth is advantageous in two ways. The plant is larger and thus has more photosynthesizing surface area than other plants so it can physically crowd out seedlings or young plants. In addition, since the roots and rhizomes are deeper and more extensive than new plants, Johnson grass can deplete nutrients from a large volume of soil.

Sorghum halepense is a problem in agricultural and natural settings. However, Johnson grass is considered a desirable forage crop and erosion stabilizer by some range scientists (Roundy pers. comm.). Many of the introduced species, including Johnson grass, invade disturbed areas much more readily than they do natural areas (Cox pers. comm.). Once the area is disturbed continuation of the disturbance will intensify the problem. The allelopathic substance produced by Johnson grass may inhibit germination of other species. Once Johnson grass is present it is extremely difficult to eradicate. Reduction in land disturbance and nurturing of native plants in neighboring uninvaded land may reduce the spread of Sorghum halepense.

Other threats from this species include contamination of agricultural seed stock and the increase in disease of crops due to the role of Johnson grass as an alternate host to several crop pathogens (Holm et al. 1977). The cyanogenic compounds present a threat to cattle and possibly to wildlife. Johnson grass' high pollen production leads to its notoriety as a major cause of allergies (Kearney and Peebles 1951). Large plants which dry out during summer heat may become an extreme fire hazard.

IV. CONDITION

V. MANAGEMENT/MONITORING

Preserve Selection & Design Considerations:
Complete eradication of Sorghum halepense is extremely difficult. The immense number of seeds which are produced on each panicle and which remain dormant until favorable conditions prevail provide a reservoir of potential plants. Rhizomes which over- winter deep in the soil make the subterranean portion of the plant difficult to control. The majority of the buds on the rhizomes remain dormant and thus herbicide or cultivation techniques will not harm these inactive regions. However, fragmenting the rhizomes will relieve axillary buds from apical dominance, allowing for their growth and thus increasing their susceptibility to control techniques. In addition, seed viability is drastically reduced from 50% to 2% after five years and six years, respectively, in the soil. The seed-supply in the soil can be expunged if seed development is prevented. The most likely mode of preventing the spread of Sorghum halepense is by maintaining undisturbed land contiguous to the invaded area.

Management Requirements:
Sorghum halepense is a highly competitive and invasive weed. It invades disturbed areas readily. The optimal management plan depends on the invasiveness of Johnson grass in the particular site. Two main approaches to management exist: (1) maximum reduction in the quantity of the weed is required before re- vegetation projects can commence in areas with spreading, dense coverage of Johnson grass, (2) spot control of individual Johnson grass plants with a concomitant encouragement of the surrounding established native plants is advisable in sites with low density coverage of Johnson grass and in pretreated, re-vegetated areas. In either case, restrictions in land disturbances in the surrounding area and in the pre-controlled site will reduce the likelihood of invasion by the surviving rhizome fragments.

Management practices for preventing invasion by Sorghum halepense should include avoiding soil and vegetation disturbances. Areas where the soil and native plants are kept intact should have few problems with Johnson grass. Although no formal studies have been conducted, personal observations have indicated that Johnson grass does not usually invade nondisturbed sites (Gould 1951, Diamond pers. comm., McWhorter pers. comm., Weigel pers. comm.).

Several techniques may be helpful in controlling Sorghum halepense: torching and burning, mowing and grazing, tilling and plowing and herbicide applications. These methods primarily focus on starving the plants by reducing growth, thus limiting photosynthesis which results in a reduction of stored carbohydrates (Oyer et al. 1959, McWhorter 1961a&b, Horowitz 1972d, McWhorter 1974).

For twenty years, torching fields of Johnson grass in the Southwest was the preferred control practice. Every two weeks throughout the summer, fields were burned using butane-propane burners. The cost of burning is now more than the cost of herbicides so this practice has been ceased (McWhorter 1981).

The timing of the burn is crucial. In Georgia, a single burn in late-winter resulted in a drastic increase in Johnson grass from 0.2 g/m2 to 27.4 g/m2 in that year's growing season (Odum et al. 1973). Whereas a natural fire occurring in late April reduced the amount of Johnson grass in a Texas field; this resulted in the establishment of little bluestem (Andropogon scoparius) (Wood pers. comm.). This native grass permanently outcompeted Sorghum halepense in the burned field, whereas in the adjacent unburned field Johnson grass remains a problem. In general, burning only slows the growth of perennial grasses (Lorenzi and Jefferey 1987). Weigel (pers. comm.) feels that burning may have little long term effect on controlling Johnson grass.

Clipping plants grown in containers close to the soil bi-weekly or monthly from spring through winter resulted in a severe decrease in growth (Horowitz 1972c). Three-quarters of the plants died, but one quarter of the clipped plants resumed growth the following spring. Mowing reduced the number of flowering stalks and clipping killed seedlings less than three weeks old. Frequent mowing depletes carbohydrate storage; thus, plants should eventually be exhausted of an energy supply, weakened and less likely to compete successfully (Horowitz 1972c).

Geese have been used successfully in cotton fields to control Johnson grass. They selectively eat grass seedlings (McWhorter 1981). Fencing off small pastures of Johnson grass and then overgrazing the area for several years with cows or horses may eventually weaken the plants (Cox pers. comm.). Intensive grazing at the proper time, when rhizome carbohydrate levels are low, may help in controlling Johnson grass; however, conventional cattle grazing often intensifies the problem of introduced grasses (Newman 1989).

Repeated tilling and plowing has resulted in the control of Johnson grass in some locations (Warwick and Black 1983). More information on desiccating rhizomes by tilling the soil is discussed below.

A single application of the herbicide glyphosate results in an 85% reduction in Johnson grass (Heathman pers. comm.). The encouraging effects of chemicals on the control of Johnson grass is addressed below.

MOWING: Mowing Johnson grass for several seasons weakens the plants and reduces rhizome growth (McWhorter 1981, Hamilton B. pers. comm.). Removing aerial grass shoots close to the ground is a technique used to exhaust the stored carbohydrates of perennial weeds (Horowitz 1972c). Horowitz (1972c) reports that clipping three week old seedlings will kill them, whereas McWhorter (1961b) claims that seedlings must be clipped within 14 days after emergence for death of the plants to occur. As compared to the single clipping of seedlings, plants arising from rhizomes require two clippings within the first two weeks of growth to insure death of the plant (McWhorter 1961b). Because the lowest rhizome carbohydrate concentration occurs in the spring, during initial above-ground growth, and in the fall, during over-wintering rhizome formation, clipping at this time will have the maximum controlling effect by preventing the formation of photosynthates and thus precluding a stored energy supply (Horowitz 1972d).

Repeated clipping is required to control plants which emerged more than 20 days prior to the initial treatment. Slight amounts of rhizome growth occurs even under continuous clipping (McWhorter 1961b). Bi-weekly clipping of potted plants severely reduces growth during that growing season, however, one quarter of clipped plants display renewed growth the following year (Horowitz 1972c). A single clipping of the aerial growth of plants 28 days after germination or sprouting reduces the amount of total carbohydrates in the rhizomes by 25%, however a rapid replenishment of carbohydrates is seen within 30 days after clipping (McWhorter 1974). McWhorter (1981) reports maximum growth reduction when plants are allowed to reach 12 to 15 inches in height before cutting them at ground level, whereas Lorenzi and Jefferey (1987) feel that eight inches is the maximum size that Johnson grass should be allowed to reach in order to starve the plants.

TILLING AND PLOWING: Hand hoeing is practical only where the concentration of Johnson grass is low. Shallow cultivation using sharp hoes, shovels, knives or hand pulling will remove the plants and the rhizomes from the upper portion of the soil without dividing or pulling up deep rhizomes (Heathman et al. 1986, Lorenzi and Jefferey 1987). Hoeing early in the season when plants are under three weeks old will be much more effective than hoeing older plants which have larger rhizome systems and greater concentrations of stored carbohydrates (McWhorter 1961b). Six to eight fallow plowings throughout the summer is the most effective tilling routine for large scale problems. Plows break up the rhizomes and bring them to the surface of the soil where they desiccate (McWhorter 1981). A 99% reduction in rhizome production resulted from six thorough tillings at two week intervals (Warwick and Black 1983). However, plowing could spread the rhizomes and increase the problem if contaminated machinery is used in uninfested areas (Cox pers. comm.).

CHEMICAL CONTROL: Extensive literature is available on herbicides available for Johnson grass control. The use of soil-active herbicides is not recommended due to the residual activity seen eight years after application (Hunter et al 1978). Herbicides alone will not successfully eradicate Johnson grass (Cox pers. comm.). Yearly applications will be required for an effective control plan. Extremely high herbicide rates are necessary to control Johnson grass in wheat fields if no other mechanical control is employed (Brown et al. 1987).

Many herbicides are recommended for use on Johnson grass. Only two of them are foliar sprays that are mildly toxic and rapidly decay in the soil: glyphosate (commercial name -- roundup) and dalapon (commercial name -- dowpon). Both of these chemicals are translocated to the underground tissue and act on all of the growing sites (Ross 1986). The use of paraffinic oil instead of water as the diluent reduces the required amount of five other herbicides tested on Johnson grass (Barrentine and McWhorter 1988). This may be the case with dalapon and glyphosate. A lack of consistency in dalapon's control of Johnson grass may be due to varying environmental conditions; moist conditions are required for optimal results (McWhorter 1981).

Dalapon and glyphosate are not specific for grasses and will kill any plant that is accidentally sprayed (McWhorter 1981). A new herbicide, Poast, is selective for monocots and can be used to kill grasses in mixed fields with broadleaf plants. Poast is expensive and since it kills all monocots, native grasses, etc. would also be destroyed (Heathman pers. comm.).

Glyphosate is recommended for controlling Johnson grass in natural, non-agricultural sites (Brookbank pers. comm., K. Hamilton pers. comm., Heathman pers. comm., Lorenzi and Jefferey 1987). K. Hamilton (pers. comm.) recommends using spot applications of glyphosate with a knapsack sprayer to control small areas of Johnson grass. Multiple applications for several years will be required. An 85% reduction in Johnson grass is commonly seen during the first year of application using glyphosate. Seeds and nonactive rhizomes account for the 15% regrowth of Johnson grass during herbicide activity (Heathman pers. comm.).

Silberman (pers. comm.) recommends the use of glyphosate in southern California restoration projects. The following sequence of events are used in controlling weeds and restoring native vegetation: (1) spraying a 1% solution of glyphosate (for Johnson grass) either in a backpack sprayer or from a helicopter, (2) plant natives, (3) spot applications of glyphosate using a backpack sprayer (Silberman pers. comm.).

Best results in controlling Johnson grass with glyphosate have been seen when the plants are actively growing, greater than 18 inches tall and have reached the bloom-to-head stage of growth (Silberman pers. comm., McWhorter 1981). The inflorescences should be removed to prevent the dispersal of mature seeds. In southern Arizona maximum control of Johnson grass occurs with fall applications of glyphosate (Brookbank pers. comm.). The low amount of rhizome carbohydrates in the fall may account for the effectiveness of the herbicide during this season of maximum rhizome growth. The land should not be tilled for at least a week after applying the herbicide in order to insure optimal efficiency from the single application (McWhorter 1981).

Significant control of Johnson grass occurs when dalapon is applied at either the late boot stage or the early growth stage prior to blooming; low levels of rhizome carbohydrates are found during both of these stages (McWhorter 1961a). No regrowth occurred on plants sprayed with dalapon during early growth, from initial leaf formation through seedstalk initiation, or during late boot stage (McWhorter 1961a). Oyer et al. (1959) reports maximum control from dalapon during early growth, before the seven-leaf stage is reached; this stage corresponds to the lowest rhizome carbohydrate level. RESPONSE TO MANIPULATIONS AND ABIOTIC FACTORS: Rhizome sections submerged in running water for eight days, or in still water for four weeks had little reduction in sprouting capacity (Horowitz 1972a). Thus drowning does not seem to be a control option.

Sorghum halepense grown under high salt and low water conditions result in reduced plant growth (Sinha et al. 1986). High salt concentrations have little effect on the overall biomass accumulation when water availability is not reduced. There is however, a decrease in the ratio of growth between shoots and roots with increased salinity (Sinha et al. 1986). The lack of a significant effect of water shortage or salt excess on roots is an advantage to Johnson grass plants growing in southern Arizona.

Management Programs:
Currently there are no active management projects of Sorghum halepense. However plans for managing Johnson grass at Patagonia-Sonoita Creek Preserve are being discussed. Contact: Jeffrey Cooper, Preserve Manager, Patagonia-Sonoita Creek Preserve, (602) 684-2772.

Possible source of sacaton seeds to be used for re-vegetating Johnson grass invaded sites include: Jerry Cox, Range Scientist, USDA, Aridland Watershed Management Research Unit, Tucson, Az.

Gene Knoder, Manager, Elgin Research Station, Elgin, AZ, (602) 455-5522. Referred by Jane Bock, Professor, University of Colorado, Boulder, CO.

Monitoring Requirements:
Monitoring of Sorghum halepense is easy, particularly in the summer, due to the large size of the plants. The number of inflorescences could predict the potential seed supply and the spread of plants into adjacent land would reveal information on the rhizome parameters. Recording the quantity of Johnson grass plants in marked quadrants and the boundary size of the invaded area will aid in determining the optimal control technique for the specific site. The ratio of natives to Johnson grass will be indicative of the success of control on the weed as well as the effects of the manipulation on the survival and competitiveness of the native vegetation.

Yearly summer monitoring will provide information needed to determine whether the plants' aerial extent is diminishing and whether the number of inflorescences are decreasing. Tagging the specific location of each cluster of Johnson grass will help in two ways: (1) detailed monitoring of the invasiveness of each plant clump and (2) correct identification of seedlings allowing for early control of the weed.

Both the density of the plants in the existing stands and the degree of invasion into previously uninfested land should be recorded. "Calibrated eyeballing" is the easiest and most rapid method of estimating the size of land invaded by the weed, as well as the competitiveness of native plants in an adjacent area (Weigel pers. comm.). Plant density measurements would be a more accurate, but more complicated, technique of determining the actual amount of plants (or stems) in an existing cluster. The change in infestation would probably be predicted earlier by precise numbers of plants per area rather than "eyeballed" measurements. Nested mini-plots ranging from 0.75 inches to 6.0 inches in diameter could be used to trace the gradual thinning of a stand (Sather 1987).

Monitoring Programs:
Presently no formal monitoring programs of Johnson grass are known. The following people "eyeball" the distribution of the weed: (1) Jeff Weigel, Director of Stewardship, The Nature Conservancy, Texas; (2) Jeffrey Cooper, Preserve Manager, Patagonia-Sonoita Creek Sanctuary, The Nature Conservancy, Arizona.

VI. RESEARCH

Management Research Programs:
No specific research on Johnson grass control in natural plant communities is being conducted. However ongoing research on the eradication of Johnson grass with the use of newly synthesized herbicides in the agricultural milieu takes place in most of the southern land-grant universities (K. Hamilton pers. comm.).

Management Research Needs:
An extensive amount of information on Sorghum halepense is available. The phenology, life-cycle, history, genetics, environmental requirements, beneficial and deleterious characteristics and control techniques of Johnson grass are all well documented. However, most of the information on controlling this weed deals with problems in agricultural fields. The techniques for agricultural control are most often not financially or practically feasible in a natural setting. Range management information on Johnson grass does not address control of this forage crop.

Information on controlling Sorghum halepense in a natural setting is needed. Two aspects of control appear to be essential in reducing the amount of Johnson grass: (1) destructive manipulation of Johnson grass which would allow for (2) natural competitors to become established.

Information on both optimal manipulation and native competitor establishment must be specific for the problem site. The temperature, precipitation, humidity and elevation will determine the optimal control technique. Introduced species in low elevation sites with little precipitation and high temperatures are often difficult to control (Cox pers. comm.).

Experimental plots should be employed for long term studies of various manipulation techniques including burning, mowing and tilling. Spring burns conducted during the first three weeks of shoot growth, when the carbohydrate supply is at a minimum level, followed by weekly mowings for one to several years may provide maximum control. If possible, the plots should be separated enough (greater than 10 m apart) to reduce the likelihood that two stems from the same underground plant system would be exposed to two different treatments.

How many years of control are necessary before re-vegetation projects can begin? The depth and dormancy (apical suppression) characteristics of the rhizomes may make it essentially impossible to completely eradicate Johnson grass. Once Sorghum halepense is eliminated, what are the best ways to rapidly establish native plants in order to prevent the establishment of the remaining Johnson grass fragments and other invasive weeds? Which natives fill the same niche as Johnson grass? Are there any of these natives present in the location? If not, research to determine what plants were originally growing at the site before the land was disturbed must be conducted and then a source of seeds must be located. Determination of the optimal conditions for germination and establishment of seeds is essential in re-seeding experiments (Martin and Cox 1984). The long term survival of the native plants should be analyzed before elaborate re-vegetation projects take place.

Spot herbicide treatments, rather than large scale eradication techniques, may be sufficient for stable areas with small quantities of Johnson grass intermixed with established native plants. The combination of manipulation techniques and maintenance of established native plants must be studied. Will there be deleterious effects on the natives when the Johnson grass is manipulated? Winter burning is detrimental to sacaton (SPOROBOLUS) growth (Cox and Morton 1986). What type of control would aid in reducing the number of Johnson grass plants without disrupting the established native plants?

VII. ADDITIONAL TOPICS

VIII. INFORMATION SOURCES

Bibliography:

Alex, J., R. McLaren, and A. Hamill. 1979. Occurence and winter survival of Johnson grass (Sorghum halepense) in Ontario 59:1173-1176.

Barrentine, W. and C. McWhorter. 1988. Johnson grass (Sorghum halepense) control with herbicides in oil diluent. Weed Science 36: 102-110.

Bock, J. 1989. Professor, Department of Environmental Population and Organismal Biology, University of Colorado. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 29.

Brookbank, G. 1989. Extension Urban Horticulturist, Cooperative Extension Center, University of Arizona. Conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 1, 1989.

Brown, S., J. Chandler and J. Morrison. 1987. Weed control in a conservation tillage rotation in the Texas Blacklands. Weed Science 35:695-699.

Cox, J. 1989. Range Scientist, USDA, Aridland Watershed Management Research Unit. Conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 30.

Cox, J. and H. Morton. 1986. Big sacaton (SPOROBOLUS WRIGHTII) riparian grassland management: annual winter burning, annual winter mowing, and spring-summer grazing. Applied Agricultural Research 1(2): 105-111.

Diamond, D. 1989. Prairie Specialist, Texas National Heritage Program. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 12.

Friedman, T., and M. Horowitz. 1970. Phytotoxicity of subterranean residues of three perennial weeds. Weed Research 10: 382-385.

Ghersa, C., E. Satorre and M. Van Esso. 1985. Seasonal patterns of Johnson grass seed production in different agricultural systems. Ireal Journal of Botany 34:24-30.

Gould, F. 1951. Grasses of southwestern United States. University of Arizona, Tucson, Arizona. 343 pp.

Hamilton, B. 1989. Director of Stewardship, The Nature Conservancy, Oklahoma. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. September 12.

Hamilton, K. 1989. Professor, Department of Plant Science, University of Arizona. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 18.

Heathman, S. 1989. Weed Specialist, Department of Plant Science, University of Arizona. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, Arizona. September 12.

Heathman, S., K. Hamilton, and J. Chernicky. 1986. Control weeds in urban areas. Cooperative Extension Service 8653. University of Arizona, Tucson, Arizona. 4 pp.

Holm, L. G., P. Donald, J. V. Pancho, and J. P. Herberger. 1977. The World's Worst Weeds: Distribution and Biology. The University Press of Hawaii, Honolulu, Hawaii. 609 pp.

Horowitz, M. 1972a. Effects of desiccation and submergence on the viability of rhizome fragments of Bermudagrass and Johnsongrass and tubers of Nutsedge. Israel Journal of Agricultural Research 22(4): 215-220.

Horowitz, M. 1972b. Early development of Johnson grass. Weed Science 20(3):271-273.

Horowitz, M. 1972c. Effects of frequent clipping on three perennial weeds, Cynodondactylon, Sorghum halepense, and Cyperus rotundus. Experimental Agriculture 8:225-234.

Horowitz, M. 1972d. Seasonal development of established Johnson grass. Weed Science 20(4): 392-395.

Horowitz, M. 1973. Spatial growth of Sorghum halepense. Weed Research 13:200-208.

Hull, R. 1970. Germination control of Johnson grass rhizome buds. Weed Science 18:118-121.

Hunter, R., A. Wallace, and E. Romey. 1978. Persistent atrazine toxicity in Mohave desert shrub communities. Journal of Range Management 31 (3):199-203.

Kearney, T.H., and R.H. Peebles. 1951 (with supplement in 1960). Arizona Flora. Univ. California Press, Berkeley. 1085 pp.

Lorenzi, H., and L. Jeffery. 1987. Weeds of the U.S. and their control. Van Nostrand Reinhold Co., New York. 355 pp.

Martin, Martha H., and Jerry R. Cox. 1984. Germination profiles of introduced lovegrasses at six constant temperatures. Journal of Range Management. 37(6):507-509.

McWhorter, C. 1961a. Carbohydrate metabolism of Johnson grass as influenced by seasonal growth and herbicide treatments. Weeds 9: 563-568.

McWhorter, C. 1961b. Morphology and development of Johnson grass plants from seeds and rhizomes. Weeds 9:558-562.

McWhorter, C. 1974. Water-soluble carbohydrates in Johnson grass. Weed Science 22(2): 159-163.

McWhorter, C. 1981. Johnson grass as a weed. USDA Farmers Bulletin 1537: 3-19.

McWhorter, I. 1989. Preserve Manager, Sandylands Preserve, TX. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. September 11.

Monaghan, N. 1979. The biology of Johnson grass (Sorghum halepense). Weed Research 19:261-267.

Newman, D. 1989. Grasslands: history and revegetation projects. Memo in grasslands file, TNC, Tucson AZ.

Odum, E., S. Pomeroy, J. Dickinson and K. Hutcheson. 1973. The effects of late winter litter burn on the composition, productivity, and diversity of a 4-year old fallow-field in Georgia. Tall Timbers Fire Ecology Conference 13: 399-419.

Oyer, E., G. Gries and B. Rogers. 1959. The seasonal development of Johnson grass plants. Weeds 7:13-19.

Parker, K. 1982. An illustrated guide to Arizona weeds. University of Arizona Press, Tucson AZ. 72pp.

Ross, M. 1986. Johnsongrass- two decades of progress in control. Crops and Soils 39: 12-14.

Roundy, B. 1989. Professor, Range Management, University of Arizona. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. August 30.

Sather, N. 1987. Element Stewardship Abstract - POA PRATENSIS and POA COMPRESSA. TNC, Minneapolis, MN. 19pp.

Silberman, R. 1989. Monsanto Representative, Fresno, CA. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. September 28.

Sinha, N., R. Gupta and R. Rana. 1986. Effect of soil salinity and soil water availability on growth and chemical composition of Sorghum halepense. Plant and Soil 95: 411-418.

Warwick, S. and L. Black. 1983. The biology of Canadian weeds - Sorghum halepense. Canadian Journal of Plant Science 63: 997-1014.

Weigel, J. 1989. Director of Stewardship, The Nature Conservancy, Texas. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. September 11.

Williams, R. and B. Ingber. 1977. The effect of intraspecific competition on the growth and developmetn of Johnson grass under greenhouse conditions. Weed Science 25(4): 293-297.

Wood, T. 1989. Preserve Manager, Mile Hi/Ramsey Canyon Preserve, AZ. Telephone conversation with D. Newman, The Nature Conservancy, Tucson, AZ. November 14.

IX. DOCUMENT PREPARATION & MAINTENANCE

Edition Date: 93-03-21

Contributing Author(s): Dara Newman