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TOXICITY, ABSORPTION, TRANSLOCATION, AND METABOLISM OF CHLORIMURON IN YELLOW AND PURPLE NUTSEDGE (CYPERUS ESCULENTUS AND C. ROTUNDUS)

The Ohio State University Ph.D. 1987

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TOXICITY, ABSORPTION, TRANSLOCATION, AND METABOLISM OF CHLORIMURON IN YELLOW AND PURPLE NUTSEDGE (Cyperus esculentus and C. rotundus)

DISSERTATION

Presented in Partial Fulfilment of the Requirements for the Degree Doctor of Philosophy in the Graduate School of The Ohio State University

By

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* * * * *

The Ohio State University
1987

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I express my sincere gratitude to my parents whose love and sacrifices have made my academic career possible.
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Studies in Entomology: Professor Fisher

Studies in Horticulture: Professor Gorski

Studies in Pharmacy: Professors Brueggemeier, Desouza, and Malspeis
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INTRODUCTION

Yellow nutsedge (Cyperus esculentus L.) and purple nutsedge (C. rotundus L.) are troublesome perennial weeds in many parts of the world. Yellow nutsedge is a weed in 21 crops in at least 30 countries while purple nutsedge is a weed in 52 crops in 92 countries (39). Because of their perennial habit, rapid growth, worldwide distribution, and difficulty in control, yellow and purple nutsedge have been ranked as the world's sixteenth and first worst weeds, respectively (39). Yellow nutsedge is found in all states of the United States (10, 56, 94). Purple nutsedge, due to its inability to withstand freezing soil temperature (76), is limited in its distribution to the southern states of the United States (10, 39, 76, 94). Yellow nutsedge is most often found on low, moist areas while purple nutsedge is found more often on well drained soils (39).

Yellow and purple nutsedge possess a number of characteristics that enable them to compete effectively and reproduce rapidly. They are photosynthetically efficient $C_4$ plants (13). They propagate mainly by tubers (39, 65, 71) and rarely by seeds (39, 44, 65, 90). They are major weeds of corn, soybean, cotton, rice, peanuts, sugarcane, tobacco, and several vegetables. Infestations of yellow and purple nutsedge reduce crop yields, lower quality, and harbor
insects and pathogens. Yield losses due to yellow and/or purple nutsedge range from 0 to 89% in various agronomic and horticultural crops (45, 50, 53, 59, 60, 69, 70, 75, 83, 95, 98, 100).

Nonchemical methods have failed to give satisfactory nutsedge control. Several relatively new herbicides have shown promise for effective and consistent nutsedge control. However, effectiveness varies depending upon their chemistry, crops and weeds present, and soil type and climatic factors. Chlorimuron ([ethyl 2-[[([4-chloro-6-methoxypyrimidin-2-yl)amino]carbonyl)amino]sulfonyl]benzoate] is an experimental herbicide. It is selective and systemic, and controls important broadleaf weeds in soybeans. In preliminary studies, it was found to be effective on yellow nutsedge (17, 19, 29). However, systematic studies on the activity of this new herbicide in yellow nutsedge has not previously been reported. Whether this herbicide, which is effective on yellow nutsedge, is also effective on another closely related but more troublesome weed, purple nutsedge, needs to be studied.

The investigations were, therefore, carried out on yellow and purple nutsedge with the objectives: 1) to determine pre- and postemergence lethal rates of chlorimuron; 2) to study parent tuber viability after pre- and postemergence exposure to chlorimuron; 3) to assess the degree of absorption and translocation of chlorimuron in young propagules; 4) to assess the degree of absorption, translocation, and metabolism of foliar-applied chlorimuron; and 5) to determine the efficacy of chlorimuron for yellow nutsedge control in field grown soybeans.
LITERATURE REVIEW

1. Distribution

Yellow and purple nutsedge are often called "nutgrass" or "coffeegrass". These weeds not only infest millions of acres of cropland but also lawns, gardens, flowerbeds, roadsides, neglected areas, banks of irrigation canals, and even paved parking areas (36, 39, 94). They are weeds on all continents. Yellow nutsedge is found in all states of the United States (10, 56, 94). Purple nutsedge, due to its inability to withstand freezing soil temperature (76), is limited in its distribution to the southern states of the United States (10, 39, 76, 94). Yellow nutsedge tolerates high soil moisture while purple nutsedge can survive the highest temperatures known in agriculture. Both species grow very well on all soil types but are quite intolerant of shade (39).

2. Classification

Yellow and purple nutsedge are members of Cyperaceae (sedge family). They are classified in the order Graminales, the class Angiospermae, and the subclass Monocotyledonae. The Cyperaceae family consists of about 75 genera and over 4000 species. In the
genus *Cyperus*, there are over 600 species, most of which occur in the tropics and subtropics (28). Yellow and purple nutsedge are the principal tuber bearing species of the *Cyperus* genus present in the United States (28). Members of Cyperaceae are often confused with members of Gramineae (grass family) and Juncaceae (rush family), but are distinguished by their triangular stems and three-ranked leaves with one-third phyllotaxy. The leaves have closed leaf sheaths, with no ligule, usually solid stems, and each flower is subtended by a single glume or scale (106).

3. Ecotypes

Since yellow and purple nutsedge are distributed widely throughout the world, considerable variation within the species is expected. Some of these ecotypes have been identified by varietal names, while others only by differing physical characteristics (78, 106). However, no systematic classification of ecotypes exists (80). Costa and Appleby (18) described differences in two yellow nutsedge varieties, *C. esculentus* L. var. *esculentus* L. and *C. esculentus* L. var. *leptostachyus* Boeckl. They found that var. *esculentus* had longer, narrower, and more upright leaves and fewer tubers, rhizomes, and seed heads than var. *leptostachyus*. Many other workers also have reported ecotypic differences in yellow nutsedge (55, 84, 86, 109, 110). Wills (105) reported several physical and morphological differences in purple nutsedge collected from 14 states within the United States and from 21 locations around the world.
4. Morphology

Yellow and purple nutsedge are perennial herbs which reproduce extensively by rhizomes and tubers. During the growing season the rhizome may develop into either a new basal bulb with shoot and rhizomes or into a tuber, depending upon environmental conditions. The above ground shoots of yellow and purple nutsedge initially consist of a triangular stem-like fascicle of leaves which later develop into a solid triangular rachis (stem or culm). The rachis extends through the center of the fascicle and terminates as an umbel (inflorescence).

These two species can be distinguished by leaf characteristics, inflorescence color, and tuberization pattern (39, 104, 107, 108). Yellow nutsedge has upright pale green leaves with long needle shaped leaf tips, yellowish brown inflorescence, and rhizomes terminating as single tubers. In comparison, purple nutsedge has prostrate dark green leaves with boat shaped leaf tips, a red, reddish brown, or purplish brown inflorescence, and rhizomes producing tubers in a chain.

a. Leaves. Leaves emerge from the basal bulb in an infolded, triangular fascicle, beginning at the outside, progressing inward and terminate with a seed bearing rachis. A leaf develops from the intercalary meristem at the leaf base in the basal bulb. The upper surface of the leaf is composed of parallel rows of large epidermal cells covered by a prominent waxy cutin. No stomates are present on
the upper surface. The lower surface is composed of smaller, thinly
cutinized epidermal cells along with numerous parallel rows of
stomates and hypodermal fibers (107, 108). Both sedges possess the
highly efficient $C_4$ photosynthetic pathway (13).

b. **Basal bulb.** Basal bulbs are subterranean, initially formed on
the ascending rhizome that originates from the sprouting tuber and
contain meristems for leaves, rhizomes, roots, and flower
structures. They are formed from the meristematic cells of the
rhizome tip and essentially consists of a short acropetal stem with
compact nodes (107, 108). The shoot consists of leaves developed
from primordia at nodes in the basal bulb. Bases of the leaves form
a fascicle as they extend through the soil from their attachment at
the basal bulb.

c. **Rhizomes.** Rhizomes originate as auxiliary buds at the
cotyledonary node of the seedling bulb or at the prophyllar node of
the secondary bulb (42). The rhizome grows as an indeterminate stem
consisting of a series of elongated internodes, nodal cladophylls,
and a pointed terminal. The growing point of an indeterminate
rhizome can differentiate into either a tuber or a basal bulb (42,
108). The rhizomes growing from sprouting tubers, however, are
determinate and transform mostly into basal bulbs. The rhizomes
grow in length primarily by internode elongation.
d. **Tubers.** Tuber formation begins with an abrupt reduction of internode elongation at the rhizome tip and formation of a number of short cladophylls (scale leaves). The entire shoot apex then swells rapidly to form the initial tuber encased in scale leaves. As growth continues the entire structure expands spherically to form a new tuber (42). The newly formed tuber is white, succulent, and turns dark to black in purple nutsedge and brown in yellow nutsedge, as it matures. The papery scale leaves persist for a considerable time and leaf scars are evident when they become detached (39). The tuber consists of several buds which are surrounded by leaf primordia. The oldest bud is the largest and most basipetal (8).

e. **Roots.** Roots originate from endodermal tissues of tubers, basal bulbs, or rhizomes (108). Roots constitute only a small portion of the total plant biomass, whereas rhizomes, basal bulbs, and tubers dominate the mass of the subterranean part of the plant (43).

5. Growth and development

a. **Vegetative development.** A few weeks after emergence, rhizomes develop from the basal bulb. These early season rhizomes elongate nearly horizontally from the bulb. The tips turn upward, differentiating into secondary basal bulbs similar in structure to the primary bulbs. These secondary bulbs produce shoots and then rhizomes. Tertiary bulbs and bulbs of a higher order are possible,
forming a complex system. Vegetative growth is rapid from the time of rhizome initiation until tubers start to form (78, 80). Photoperiod is considered the major known factor in controlling the differentiation of rhizomes (26, 42), but temperature fluctuations, chemicals, and nutrition also affect differentiation (7, 26). Long days promote differentiation into basal bulbs, while short days promote tubers formation (42). Daylength is maximal in early summer, apparently promoting maximum vegetative growth for the first several months of growth.

b. Tuber production. As the day length becomes shorter during the growing season, rhizomes differentiate into tubers instead of basal bulbs (26, 42). In purple nutsedge, the newly formed basal bulb may look very much like a newly formed tuber. It is their activity that distinguishes them; the tuber remains temporarily dormant while the basal bulb differentiates into a new aerial shoot (39). Both yellow and purple nutsedge plants begin to produce new tubers about 8 weeks after the original tubers are planted (33, 91).

A single yellow nutsedge tuber, growing alone, may produce as many as 1900 plants and 7000 tubers in a year (91). However, fewer tubers are produced when crops grow with the yellow nutsedge than when nutsedge grows without competition. Rao (66) in India, found that one purple nutsedge tuber could produce 99 tubers in 90 days. He calculated that, on an area basis, this would mean 8 million tubers per hectare in cultivated areas and 4.8 million per hectare in uncultivated areas. Tuber yields of 5 to 28 ton/ha have been
reported for yellow nutsedge (91). Normally the bulk of the tubers are formed in the top 15 cm of the soil with none below 30 cm (1, 6). However, there are reports that some tubers are produced as deep as 46 cm (78).

Tuber production is reduced greatly when yellow nutsedge plants are shaded by crops, but tubers are still produced under very low light intensity (77). Photoperiod plays an important role in tuber formation. Photoperiods less than 12.5 h promoted tuber formation in yellow and purple nutsedge (42, 103). Garg et al. (26) found that high levels of nitrogen, a long photoperiod (15.5 h), and high levels of gibberellic acid at 12.5 h photoperiod inhibited tuberization in yellow nutsedge, whereas high temperatures of 27 C and 33 C at low nitrogen level favored tuberization.

c. Tuber dormancy. Tuber dormancy is considered one of the major obstacles in controlling these weeds. In yellow nutsedge, maximum tuber dormancy occurs at the end of the growing season (fall), and dormancy is minimum in the spring (86). Tumbleson and Kommedahl (92) reported 12% sprouting in fall-harvested tubers and 95% in spring-harvested tubers. Tubers remain dormant for extended periods, possessing half-lives from 4 months for tubers 10 cm below the soil surface to 6 months for tubers 20 cm below (81). The mechanism of dormancy has been little explored, although there are many experiments on breaking the dormancy. Many workers have reported presence of sprouting inhibitors in yellow and purple nutsedge tubers (41, 85, 92). Dormancy of tubers can be broken by
various chemicals such as ethylene chlorohydrin, thiourea, ethyl ether, KSCN, gibberellic acid, and benzyladenine, or physical actions such as cold storage, desiccation, leaching with water, and tillage (6, 87, 88, 91, 92).

d. Tuber sprouting. The cold and wet conditions of overwintering serve to break tuber dormancy in yellow nutsedge (6, 81, 92). The tubers near the surface could be killed by extreme cold or desiccation (76, 81, 88). Temperatures around -7 to -10 ºC killed yellow nutsedge tubers in laboratory tests, but in the field tubers have survived -10 ºC (81). Sprouting of tubers in yellow nutsedge can occur at temperatures above 12 ºC and proceeds at a maximum rate above 25 ºC (81). In purple nutsedge, Ueki (93) obtained 95% tuber sprouting at 30 to 35 ºC, with no sprouting above 45 ºC or below 10 ºC. Sprouts can emerge from tubers located 30 cm or deeper in the soil (81, 91). However, percentage of tubers sprouting decreases as depth of tuber in the soil increases.

Due to the presence of numerous buds, a tuber can sprout several times, as well as produce several propagules at one time. Apical dominance determines the number of propagules produced per sprouting (6, 8, 42, 65, 79, 89, 91). In yellow nutsedge tubers, buds usually sprout in acropetal order starting with the oldest bud (8). In contrast, the apical bud of purple nutsedge tubers always sprouts first. The terminal tuber exerts apical dominance over all other tubers in a long chain, but this control is not as strong as the dominance within the tuber (39). Separation of a tuber from a
chain releases it from apical dominance. As a rhizome reaches the soil surface, the tip encounters sunlight and diurnal temperature fluctuations, which are the principal factors in stimulating basal bulb formation below the soil surface (78). In a uniform seedbed, all basal bulbs are formed at a comparable distance (1 to 4 cm) from the soil surface, regardless of the depth of the tuber (79).

e. Flowering and seed production. In yellow nutsedge, flowering does not occur regularly in all populations every season, and plants do not produce seeds every year (78, 80). Photoperiod has been considered the major factor that controls flowering in yellow nutsedge (42). In greenhouse studies, flowering occurred at photoperiods from 12 to 14 h (42). In the Corn Belt, however, yellow nutsedge flowers and completes nearly all its growth under days longer than 14 h, indicating perhaps that additional factors affect the flowering process in the field (78). In purple nutsedge flowering is common everywhere. Holm et al. (39) reported that the plant is stimulated to flower in short photoperiods of 6 to 8 h, whereas, Williams (102) reported that flowering occurred only at 12 h photoperiods and concluded that purple nutsedge is intermediate in flowering response to day length. The first evidence of floral development is the appearance of a foliar tube elongating from the fascicle center. The hollow foliar tube is formed by the two most recently differentiated leaves growing as a single unit (42). The flowering structures differentiate from the apical meristem of the basal bulb (107, 108) and elongate inside the foliar tube. The well
developed inflorescence bursts from the foliar tube as it protrudes from the fascicle. The inflorescence continues to develop, sometimes forming mature, viable achenes.

Seed production in both yellow and purple nut sedge is possible (2, 37, 44, 65, 90). Yellow nut sedge seed has greater probability of germination than purple nut sedge (90). The seeds are dormant when produced but dormancy is easily broken by several months of storage at room temperature or at 10 C (44). Bell et al. (6) stated that yellow nut sedge seed did not germinate at a constant temperature of 24, 29, or 35 C but did germinate if the night temperature was less than 21 C for 15 h. Holm et al. (39) stated that light has little effect on germination and Bell et al. (6) believed that darkness was not inhibitory if the temperature was favorable for germination. Ranade and Burns (65) obtained considerable increase in germination of purple nut sedge seeds by heating seeds at 59 C for 1 h. Justice and Whitehead (44) reported that the heaviest seed of purple nut sedge germinated up to 7 % without special treatment. With heat treatment, 59 to 60 C for 3 h, 9 to 17 % of the seeds germinated. Holm et al. (39) reported that purple nut sedge seed seldom germinates more than 1 to 5 %. However, the role of seed in the establishment and spread of nut sedges is of little significance to agriculture (39, 56).
6. Interference with crops

a. Competition. Yellow and purple nutsedge possess the highly efficient $C_4$ photosynthetic pathway. Black et al. (13) proposed that plants which fix CO$_2$ at high rates have an initial advantage which makes them either potentially high yielding crops or serious weeds. If aggressive weeds gain an early competitive advantage over crops, it is difficult for crops to overcome this disadvantage (45). Elmore et al. (23) in their greenhouse studies found that purple nutsedge was initially very aggressive and the most competitive weed of the five species they studied. Prickly sida was least competitive and cotton was intermediate and approximately equivalent in competitive ability to large crabgrass and velvetleaf. This early aggressiveness of nutsedges appears to be a major reason accounting for their ability to compete with even tall growing crops (45).

Since yellow and purple nutsedge plants are relatively short stature, one may not expect them to compete for light, yet yellow nutsedge which was 11 cm tall soon after cotton emergence reached 44 cm in cotton plots within 8 weeks and yellow nutsedge was as tall or taller than cotton for most of the growing season (48). William and Warren (100) reported that purple nutsedge competed for light in slow growing noncompetitive vegetable crops and competition was severe in crops such as garlic, okra, and carrot. Okafor and DeDatta (59) reported that purple nutsedge competition for light was a contributing factor to poor yields in rice.
Severe losses in yield could occur under conditions where yellow nutsedge competes with corn for moisture (83). Often soil moisture is not limiting on the heavy textured soils in the Corn Belt, rendering any competition from yellow nutsedge insignificant. The most potential for crop losses occurs on light textured soils or under drought conditions (78). Keeley and Thullen (48) reported that dense populations of yellow nutsedge (100 shoots/m²) in cotton may deplete soil moisture to the extent that maintenance of proper cotton stands is jeopardized. Okafor and DeDatta (59) found that competition of purple nutsedge for soil moisture contributed to poor yields in rice and the competition became more serious with increased nitrogen fertilization.

The nutsedges also compete with crops for nutrients. When these were grown in association with crops, both sedges were found to reduce the nutrient content of some crops. Volz (95) reported that yellow nutsedge decreased the total nitrogen content of silage corn, tomato fruit, and soybean grain. The decrease was 33, 18, and 5% in corn, tomato, and soybean, respectively, when crops were grown with nutsedge. Okafor and DeDatta (59) reported that with increases in purple nutsedge densities, total nitrogen increased in nutsedge and decreased in rice and that nitrogen uptake by nutsedge was negatively correlated with rice yield.

b. Allelopathy. Allelopathy of yellow and purple nutsedge has been reported under greenhouse conditions (22, 40, 85). Tests have shown that dried yellow nutsedge tuber tissues are allelopathic to other
plants. Soybean growth was inhibited more than corn when tuber residues were present in the rooting medium (22). The allelopathic substances were found to be phenolic in nature (25, 85). Generally, those that studied inhibitory effects of nutsedge residues in soil reported that allelopathic substances may decrease over a period of time but were still appreciable even after 4 months (24, 40).

c. Host plants. Yellow and purple nutsedge serve as alternative hosts for several plant pathogens, such as lucerne dwarf virus, Ascochyta sp., Fusarium sp., Puccinia canaliculata, and root knot nematodes (9, 39, 56).

7. Crop losses

Some crops tolerate low to moderate infestations of yellow nutsedge without substantial yield losses. Yellow nutsedge densities of 40 shoots/m² did not reduce yields of cotton, whereas 70 shoots/m² reduced yields by 12 to 36% (50). In corn, moderate infestations of yellow nutsedge reduced yields only 17%, whereas heavy infestations reduced yields 41%. There was an 8% reduction in corn yield for every 100 shoots/m² of yellow nutsedge (83). In soybeans, moderate infestations of yellow nutsedge did not reduce yields, whereas 128 shoots/m² remaining at harvest reduced yields 29% (98). Yield losses due to yellow and/or purple nutsedge vary considerably depending upon weed density, time of competition, crops, cultivars, and environmental
factors. Yield losses of 17 to 79% in corn (69, 83), 0 to 87% in soybeans (69, 98), 0 to 36% in cotton (50), and 34 to 40% in rice (59) have been reported. In horticultural crops, yield losses range from 35% in cabbage, 41% in beans, 43% in cucumber, 50% in carrot, 53% in tomato, 62% in okra to 89% in garlic (100). Aside from reducing yields these weeds may lower crop quality as well. In some badly infested potato fields in the United States, every potato tuber was found to have a rhizome running through or into it (39).

8. Control

a. Biological. Although considerable effort has been made to develop biocontrol agents for both yellow and purple nutsedge, these control methods currently are not successful enough for field use (78). Several insects, fungi, and nematodes attack yellow nutsedge (56). Some promising results have been reported for control of yellow nutsedge with the insect Bactra verutana (51). However, long periods of weed growth before the insects became plentiful may have limited their usefulness for biological control. The insect Bactra truculenta, which bores into the stems of purple nutsedge, showed promise for biological control in Hawaii in the early years after its introduction from the Philippines in 1925. As the populations of insect increased, the population of Trichogramma minutum, which parasitizes insect eggs, also increased. Consequently, many of the eggs were killed and biological control of purple nutsedge was never attained (39).
b. **Management.** The nutsedges cannot effectively compete with crops once crops form a canopy. The lack of aggressiveness of nutsedges in crops that quickly form a shade canopy is attributed to their sensitivity to shade (43, 49, 62). Therefore, good management practices help crops gain an early competitive advantage over nutsedges. The time required to produce canopies which reduce growth and reproduction of yellow nutsedge varies with each crop but generally is between the first 3 to 16 weeks (49, 50, 100). Cotton plots maintained free of yellow nutsedge for 2 to 6 weeks resulted in yields comparable to weed-free plots (50). William and Warren (100) found that periods of purple nutsedge-free maintenance had to be extended for noncompetitive crops. Garlic had to be maintained free of nutsedge for 13 weeks as compared to 7 weeks for okra and cucumber, 5 weeks for carrot, and 3 weeks for tomato.

Bendixen and Stroube (11) reported that fertilization increased competitiveness of aggressive crops, but when crops are not aggressive, fertilization promoted nutsedge growth. Okafor and DeDatta (59) observed that application of nitrogen benefited purple nutsedge more than rice. Narrow row spacings of crops have been shown to be more effective than wide row spacings in suppressing nutsedges. Row spacings of 45 to 60 cm for corn and 19 to 38 cm for soybeans resulted in better control of nutsedges and increased yield, compared to 90 and 76 cm row spacings, respectively (15, 16). Growing competitive crops and using selective herbicides gradually reduces populations of yellow nutsedge tubers with time. Two years of either growing alfalfa treated with EPTC or double
cropping barley with corn treated with butylate preceding cotton treated with MSMA were more effective in reducing viable tubers than continuous cotton treated with MSMA (52).

c. Mechanical. Tillage is an important part of nutsedge control, but alone will not provide satisfactory control. Preplant tillage can be beneficial in stimulating tuber sprouting, moving tubers to the surface for kill by desiccation, and killing propagules already established (27, 72, 73, 86). Maximum benefits in nutsedge control are observed if tillage is delayed until substantial tuber sprouting has occurred. However, control with preplant tillage is diminishing because of the increasing trend among growers to plant early in the season before the tubers sprout (78). Cultivation after planting is an effective control measure, especially when combined with preplant herbicides. Cultivation effectively controls the weeds between the rows, and the crops can compete effectively with the nutsedges left in the row.

d. Chemical. The use of herbicides appears to be by far the most effective method for nutsedge control. Some soil-applied herbicides are effective in killing the emerging shoots, while others applied to foliage inhibit growth and rhizome production and thus reduce tuber formation. EPTC is one of the oldest herbicides used for yellow nutsedge control in corn (6). Although effective, EPTC causes excessive injury to corn. Soil-incorporated EPTC appears to be effective in controlling purple nutsedge (35, 101). Butylate,
chemically similar to EPTC, is also an effective herbicide for control of yellow nutsedge in corn. However, although butylate is less effective than EPTC it has less potential for causing crop injury. Presently, these herbicides are formulated with antidotes to reduce the possibility of crop injury. These herbicides do not control yellow nutsedge as long as some of the other herbicides (78). Vernolate, similar in chemical properties to EPTC and butylate, effectively controls yellow nutsedge in soybeans. This herbicide sometimes injures soybeans but seldom reduces yield (78).

Alachlor and metolachlor selectively control yellow nutsedge in both corn and soybeans (3, 20, 21, 58, 98). They apparently do not kill tubers, but act by delaying sprouting of tubers and killing young nutsedge shoots (3, 20, 47, 98). These herbicides perform best on silt loam and silty clay loam soils with medium to high organic matter. On coarse textured soils, however, their performance is often erratic. Metolachlor provides better control with more residual activity than alachlor (21, 58), but has somewhat more potential for injuring corn than alachlor (21). Alachlor controls purple nutsedge also but it is somewhat less effective on purple compared to yellow nutsedge (46).

Atrazine is widely used for weed control in corn and may provide some control of yellow nutsedge (61). Control is only fair and often erratic. Atrazine in combination with alachlor, EPTC, or butylate may provide better control (78). Perfluidone, either as soil-incorporated or postemergence treatment, controls yellow nutsedge in cotton (82). Dichlobenil has been reported to inhibit
tuber sprouting in both sedges (32). Fluridone controlled yellow nutseedge in cotton. Control with fluridone was better when soil-incorporated than in preemergence applications (5).

Foliar application of bentazon selectively controls yellow nutseedge in soybeans (82). Control generally is better with split applications 10 to 14 days apart with complete foliar coverage than with single application. As it frequently kills only the foliage contacted by the spray, yellow nutseedge sometimes recovers (78, 97). Several sequential applications of 2,4-D are often required for yellow and purple nutseedge control. Hauser (35) reported excellent purple nutseedge control from 9 applications of 2,4-D initiated 1 or 2 weeks after shoot emergence. In yellow nutseedge, a single application of 2,4-D reduced growth of shoots and rhizomes to less than 60% of the control (12). Two applications of amitrole during the first 4 weeks after shoot emergence gave good control of purple nutseedge (34, 35). Purple nutseedge control with paraquat is inconsistent. By killing an emerged shoot with paraquat, dormant buds on the basal bulb or tuber may produce shoots as a result of released apical dominance (31). Glyphosate is a highly active herbicide with excellent potential for controlling a large number of annual and perennial weeds, including nutseedges (82). Since glyphosate lacks selectivity its use is limited to small, heavily infested areas as a spot treatment.
9. Absorption and translocation of soil-applied herbicides

The site of uptake is an important factor in determining whether a particular herbicide is more phytotoxic when incorporated into the soil or when applied to the soil surface. Herbicides, whose main site of uptake is roots, may have little effect when applied to the soil surface unless they are leached to the root zone. Conversely, herbicides whose main site of uptake is shoots may have greater effect when applied to the soil surface, as the emerging shoot passes through the higher concentration of herbicide present close to the soil surface. Absorption and translocation studies help locate the possible sites of herbicide uptake and its movement within the plant to the sites of action. In nutsedges, these sites could be tuber, emerging shoots, or roots.

Generally, the sites of herbicide uptake depend on the nature of the herbicide and the plant species involved. In corn and peas, 2,4-D, naptalam, simazine, diuron, and dalapon entered primarily through the roots while uptake of EPTC, chloropropham, and trifluralin was primarily through the shoots (64). Exposing the roots of barley, oats, barnyardgrass, annual ryegrass, wheat, rice, cotton, and yellow nutsedge to EPTC caused more injury than from shoot exposure (30). Walker (96) reported that in turnip, lettuce, and ryegrass, uptake of atrazine, simazine, and linuron was by both shoot and root. Absorption of herbicides by seeds of many plant species also has been reported (6, 30, 67, 68).
In yellow nutsedge propagules, when metolachlor and alachlor were applied to shoots and rhizomes (below the basal bulb), the plants absorbed much greater amounts than when they were applied to roots (4, 20). Translocation of metolachlor was both acropetal and basipetal (20, 58). Alachlor applied to shoots and rhizomes translocated to the growing points, but the majority of alachlor applied to roots did not move (4). Both yellow and purple nutsedge absorbed greater quantities of imazaquin from shoot application than from root application (57).

10. Absorption, translocation, and metabolism of foliar-applied herbicides

In nutsedges, the effectiveness of a foliar-applied herbicide depends on rapid absorption and basipetal translocation of biologically active compounds into the underground storage organs in sufficient quantities to kill the entire plant before it is degraded to an inactive form. Absorption and translocation studies help elucidate how well the compound is absorbed and moved within the plant. Often, such studies also help to explain herbicidal selectivity among plant species.

The absorption and translocation of several foliar-applied herbicides in both sedges has been extensively studied (12, 54, 57, 74, 82, 111). In yellow nutsedge, less than 19% of the applied 2,4-D was absorbed and less than 8% was translocated one day after application. Translocation continued up to 12 days without any
degradation (12). Glyphosate applied to yellow nutsedge at the 10-leaf stage moved acropetally in treated shoots and basipetally into the untreated shoots and developing tillers (74). When bentazon was applied to 3 weeks old yellow nutsedge plants very little was translocated to other plant parts and none to the parent tuber (82). Perhaps this pattern can explain bentazon effectiveness in killing only the foliage contacted by the spray. Hill et al. (38) reported that when amitrole was applied to the yellow nutsedge bract leaf, it moved basipetally to the umbel and then acropetally to the seed, resulting in reduced seed germination.

In purple nutsedge, foliar-applied dicamba moved both acropetally and basipetally. It was distributed widely throughout the aerial parts of the plant and accumulated in the regions of meristematic activity. It was barely detectable in the underground organs, although it passed through the rhizomes and tubers into daughter plants. There was no degradation of dicamba during 10 days following treatment (54). Glyphosate translocation from treated purple nutsedge leaves to other plant parts increased from 5% of the amount applied at 1 day to 19% at 4 days after application. Tubers accumulated more glyphosate than leaves of 2 to 6 weeks old plants. As the plant age increased, accumulation decreased in both tubers and leaves. No evidence of glyphosate metabolism was observed (111). In another study, foliar-applied imazaquin translocated basipetally in both yellow and purple nutsedge but translocation was greater in purple than in yellow nutsedge (57).
Brown and Ray (14) studied the biochemical basis for soybean tolerance to chlorimuron. Soybeans rapidly metabolized chlorimuron in a half-life of 1 to 3 h, while sensitive weed species such as cocklebur and redroot pigweed metabolized chlorimuron much more slowly, in half-lives greater than 30 h. The metabolites were identified and they are herbicidally inactive.

11. Chlorimuron-technical data


![Chemical structure of chlorimuron](attachment:image.png)

Common name: Chlorimuron (Formerly DPX-P6025).

Herbicide group: Sulfonylurea.

Trade name: Classic™.
Molecular weight: 414.83.
Melting point: 186 ± 1 C.
Dissociation constant $K_a$: 4.2 at 25 C.
Partition coefficient: 2.3 at pH 7.0 (octanol/water).
Solubility at 25 C: High solubility in organic solvents.
Water solubility is 1200 ppm at pH 7.0.
Formulated product: Dispersible granules.

Chlorimuron is a new herbicide developed by Du Pont. It selectively controls many broadleaf weeds in soybeans and suppresses certain annual grasses. It can be applied preplant, preemergence, or postemergence (63). Preliminary trials conducted in several states of the United States have shown acceptable preemergence control of cocklebur, velvetleaf, morningglory, pigweed, ragweed, and other broadleaf weeds in soybeans at rates of 35 to 210 g/ha, depending upon soil type (17, 19, 29). Chlorimuron applied postemergence was effective at much lower rates than those used for preemergence. Postemergence applications at rates of 4 to 17.5 g/ha gave acceptable control of cocklebur, ragweed, velvetleaf, sunflower, pigweed, nutsedge (19, 29), burcucumber (99), and suppression of morningglory, sicklepod, and Florida beggarweed (19, 29). Soybean tolerance to chlorimuron is due to rapid metabolic inactivation (14, 17). Homoglutathione conjugates and deesterified free acids of chlorimuron are the inactive metabolites. The herbicidal activity of chlorimuron is due to its potent inhibition of acetolactate synthase, an enzyme on the biosynthetic pathway to valine and isoleucine.
TOXICITY, ABSORPTION, AND TRANSLOCATION OF SOIL-APPLIED CHLORIMURON IN YELLOW AND PURPLE NUTSEDGE (Cyperus esculentus and C. rotundus)

INTRODUCTION

Yellow nutsedge and purple nutsedge are persistent perennial weeds in many parts of the world (7, 11). They reproduce mainly by tubers and very rarely by seeds (11, 24). Generally, the tubers are produced on rhizomes present in the top 15 cm of the soil but some tubers are found as deep as 46 cm (1, 5, 19). Each tuber has several buds. The tuber can sprout several times and produce several shoots at one time (6, 20, 23). Most tubers exhibit dormancy and remain viable in soil for nearly 2 years (5, 21, 22). The longevity of tubers, the ability of tubers to sprout several times, and lack of herbicides which can kill dormant tubers have made these weeds difficult to control.

Several soil-applied herbicides have shown promise for control of yellow and purple nutsedge (2, 4, 5, 9, 10, 12, 13, 16, 25). However, the performance of soil-applied herbicides depends on several factors. The site of herbicide uptake by the plant plays an important role in determining whether a particular herbicide would
be more phytotoxic when incorporated into the soil or applied to the soil surface. Therefore, the site of herbicide uptake and lethal action should be a primary concern when deciding where the herbicide should be placed in the soil for maximum effectiveness (14, 17, 18). Absorption and translocation studies help to locate possible sites of herbicide uptake and movement within the plant to sites of action. In nutsedge propagules, these sites could be the tuber, emerging shoots, or roots. Generally, the site of herbicide uptake depends on the nature of the herbicide and plant species involved. Yellow nutsedge propagules absorbed much greater amounts of metolachlor and alachlor when they were applied to shoots and rhizomes (below the basal bulb) than when they were applied to roots (3, 9). Absorption of imazaquin was greater from shoot application than from root application in both sedges (15).

Chlorimuron is a new selective herbicide effective on yellow nutsedge as well as many broadleaf weeds (8). However, there are no detailed reports of its activity on yellow and purple nutsedge. Thus, this research was carried out on yellow and purple nutsedge: 1) to determine preemergence lethal rates and tuber viability after exposure to chlorimuron; and 2) to study the pattern of absorption and translocation of chlorimuron in young propagules.
MATERIAL AND METHODS

Celina silt loam with a pH of 6.3 and consisting of 15.7% clay, 63.3% silt, 21% sand, and 3.5% organic matter was used in these experiments. It was collected from the upper 15 cm of the soil profile at the Agronomy farm, Columbus, Ohio, air-dried, then sieved to get a particle size of less than 2 mm. Yellow nutsedge tubers were collected locally from the field, washed with water, and stored in plastic bags at 3 C until used. Purple nutsedge tubers were harvested from greenhouse grown plants and used without incubation. The forms of chlorimuron used were the commercial formulation and $^{14}$C-[U]-phenyl labeled chlorimuron (specific activity of 3.47 mCi/mM and over 99% purity).

Herbicide rate studies. Five uniform tubers of either yellow or purple nutsedge were planted 4 cm deep in 15 cm diameter plastic pots containing about 950 g of soil. After planting, herbicide rates of 0, 10, 20, 40, and 60 g ai/ha were applied to the soil surface with a pot sprayer calibrated to deliver 236 l/ha. The pots were maintained in the greenhouse at 28/20 C day:night temperatures and a 14 h photoperiod and watered as required to keep the soil moist. Four weeks after planting, the number and dry weight of emerged shoots were recorded. The parent tubers were recovered and the number of sprouts were counted, regardless of shoot emergence, and expressed as percent sprouting. The experiment was conducted in a randomized complete block design with four replications. Each
experiment was conducted twice and the data were combined for analysis. Means were separated at the 5% level of significance using Fisher's LSD test.

**Tuber viability studies.** The five tubers per treatment recovered after 4 weeks of exposure to chlorimuron in the herbicide rate studies were washed with water and replanted in 15 cm diameter plastic pots containing herbicide-free soil, then put in the greenhouse. After 3 weeks, resprouting of tubers was evaluated as described in the herbicide rate studies. The experiment was conducted twice in a randomized complete block design with four replications. Statistical analysis was performed as described in the herbicide rate studies.

**Absorption and translocation studies.** Plants for the isotope studies were cultured by soaking tubers in water for 8 h, then placing them on a tray lined with paper towels moistened with modified Hoagland's solution (Appendix). After covering the trays with aluminum foil to exclude light, a few small holes were made in the foil to permit gas exchange. The trays were then placed in a darkened germinator adjusted to give 30 C and 100% relative humidity for 3 days. At this stage, the propagules had shoots 2 cm long and three to five roots 2 to 5 cm in length. Propagules of uniform size and with a single shoot were selected for treatment.

Each propagule was transferred to a 10 cm diameter petri dish which contained 20 ml of modified Hoagland's solution. The shoot
tips were guided through a small hole of mesh to prevent the shoot tissue from contacting the nutrient solution. The petri dishes were placed in a tray lined with moistened paper towels. For shoot treatment a 3 ul drop containing 0.036 uCi of $^{14}\text{C}$-chlorimuron in water was placed on the shoot 5 mm below the tip. For 'roots and tuber' treatment the same amount of $^{14}\text{C}$ was dissolved in the nutrient solution where both the roots and tuber were in contact with the nutrient solution. During the absorption period, the propagules were placed in a growth chamber set for continuous darkness and 25/15 C temperatures for 14/10 h cycle, and 85% relative humidity. The propagules were harvested at 12, 24, and 48 hours after treatment initiation. The treated part was washed with water to remove unabsorbed $^{14}\text{C}$. The propagule was fractionated into shoot, roots, and tuber. Fractions were then lyophilized and weighed.

The lyophilized samples were combusted in a Model 306 Tri-carb sample oxidizer. The evolved $^{14}\text{CO}_2$ was trapped in a mixture of 7 ml of CO$_2$ absorbent (Carbo-sorb) and 11 ml of scintillation solution (Permafluor V). Two 0.5 ml aliquots of the treated part washes or the nutrient solutions were pipeted into scintillation vials, then 15 ml of scintillation solution (Formula 963) was added to each vial. Radioactivity from washes, nutrient solutions, and oxidations was quantified by liquid scintillation spectrometry (Beckman LS 8000). Quenching was corrected by H-number. The counts per minute were corrected for background and counting efficiency, then converted to disintegrations per minute (dpm). The dpm data
are presented as percent of total $^{14}$C recovered (Absorption) and percent of total $^{14}$C absorbed (Translocation).

**RESULTS**

**Herbicide rates and tuber viability.** Chlorimuron had a significant effect on tuber sprouting, shoot emergence, and shoot dry weight (Tables 1 and 2). Sprouting was inhibited by chlorimuron in both species when compared to the untreated controls. The decrease was significant at all rates in yellow nutsedge, while the decrease was significant only at the highest rate in purple nutsedge. At 10 g/ha, chlorimuron was effective in reducing shoot emergence as well as shoot dry weight in both species. Shoot emergence in both nutsedges decreased with increases in herbicide rate. Chlorimuron treatments reduced shoot dry weight by 99.3 to 100% in yellow nutsedge and by 84.7 to 98.3% in purple nutsedge. The differences in shoot emergence and shoot dry weight were not significant among the rates from 20 to 60 g/ha in yellow nutsedge and between the rates of 40 and 60 g/ha in purple nutsedge.

Chlorimuron was not effective in reducing parent tuber resprouting in yellow nutsedge (Table 3), while in purple nutsedge resprouting was reduced at 60 g/ha (Table 4). Previous exposure to chlorimuron had no effect on shoot emergence regardless of rates. However, shoot dry weight was significantly reduced at the highest rate in both species.
Table 1. Response of yellow nutsedge to chlorimuron 4 weeks after preemergence application.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>Sprouting (%)</th>
<th>Emerged (no./pot)</th>
<th>Dry weight (g/ha)</th>
<th>% control (X control)</th>
</tr>
</thead>
<tbody>
<tr>
<td>0</td>
<td>92.5 a</td>
<td>17.9 a</td>
<td>100.0 a</td>
<td></td>
</tr>
<tr>
<td>10</td>
<td>67.5 b</td>
<td>3.1 b</td>
<td>0.7 b</td>
<td></td>
</tr>
<tr>
<td>20</td>
<td>67.5 b</td>
<td>0.6 c</td>
<td>0.2 c</td>
<td></td>
</tr>
<tr>
<td>40</td>
<td>32.5 c</td>
<td>0.3 c</td>
<td>0.1 c</td>
<td></td>
</tr>
<tr>
<td>60</td>
<td>12.5 c</td>
<td>0.0 c</td>
<td>0.0 c</td>
<td></td>
</tr>
</tbody>
</table>

*Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.

bFive tubers were planted per pot.

cUntreated shoot dry weight was 4.16 g/pot.
Table 2. Response of purple nutsedge to chlorimuron 4 weeks after preemergence application.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>Sprouting (%)</th>
<th>Emerged (no./pot)</th>
<th>Dry weight (% control)</th>
</tr>
</thead>
<tbody>
<tr>
<td>0</td>
<td>95.0 a</td>
<td>14.9 a</td>
<td>100.0 a</td>
</tr>
<tr>
<td>10</td>
<td>95.0 a</td>
<td>7.0 b</td>
<td>15.3 b</td>
</tr>
<tr>
<td>20</td>
<td>92.5 a</td>
<td>6.4 b</td>
<td>7.6 c</td>
</tr>
<tr>
<td>40</td>
<td>87.5 a</td>
<td>3.3 c</td>
<td>1.9 d</td>
</tr>
<tr>
<td>60</td>
<td>65.0 b</td>
<td>2.3 c</td>
<td>1.7 d</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.

Five tubers were planted per pot.

Untreated shoot dry weight was 3.73 g/pot.
Table 3. Effect of chlorimuron on yellow nutsedge tuber viability 3 weeks after replanting.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>Resprouting (%)</th>
<th>Emerged (no./pot)</th>
<th>Dry weight (g/pot)</th>
</tr>
</thead>
<tbody>
<tr>
<td>0</td>
<td>60.0 ab</td>
<td>4.5 a</td>
<td>0.53 ab</td>
</tr>
<tr>
<td>10</td>
<td>60.0 ab</td>
<td>5.8 a</td>
<td>0.50 ab</td>
</tr>
<tr>
<td>20</td>
<td>72.5 a</td>
<td>7.3 a</td>
<td>0.56 a</td>
</tr>
<tr>
<td>40</td>
<td>40.0 b</td>
<td>5.0 a</td>
<td>0.32 bc</td>
</tr>
<tr>
<td>60</td>
<td>40.0 b</td>
<td>4.6 a</td>
<td>0.21 c</td>
</tr>
</tbody>
</table>

*Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.

bFive tubers were planted per pot.
Table 4. Effect of chlorimuron on purple nutsedge tuber viability 3 weeks after replanting.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>Resprouting (%)</th>
<th>Emerged (no./pot)</th>
<th>Dry weight (g/pot)</th>
</tr>
</thead>
<tbody>
<tr>
<td>0</td>
<td>55.0 a</td>
<td>3.0 a</td>
<td>0.27 a</td>
</tr>
<tr>
<td>10</td>
<td>57.5 a</td>
<td>2.9 a</td>
<td>0.24 a</td>
</tr>
<tr>
<td>20</td>
<td>40.0 ab</td>
<td>2.3 a</td>
<td>0.17 ab</td>
</tr>
<tr>
<td>40</td>
<td>50.0 a</td>
<td>2.5 a</td>
<td>0.16 ab</td>
</tr>
<tr>
<td>60</td>
<td>30.0 b</td>
<td>1.9 a</td>
<td>0.05 b</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.

Five tubers were planted per pot.
Absorption and translocation. Over 86% of the total applied radioactivity was recovered in these studies. Application of $^{14}$C-chlorimuron to shoots resulted in rapid absorption in both species (Tables 5 and 6). The species differed considerably in $^{14}$C absorption when applied to shoot than to roots and tuber. Shoots of both yellow and purple nutsedge absorbed more $^{14}$C than did roots and tubers. Shoot absorption at 12 h was less than at 24 and 48 h in both species. There were no differences through time in either species, however, in the amount of $^{14}$C absorbed by roots and tuber.

Unlike absorption, translocation of $^{14}$C (out of the treated part) was greater from roots and tuber than from shoots in both species (site means of Tables 5 and 6). Furthermore, the amount translocated from roots and tuber increased through time while there were no differences in amounts translocated from shoots of either species. The pattern of radioactivity distribution shows that most of the shoot-absorbed $^{14}$C in both species remained in the shoot (Tables 7 and 8). Of the $^{14}$C absorbed by the roots and tuber, the amount retained by the tuber seemed to be quite static while that absorbed by the roots was translocated in essentially equivalent amounts to the shoots.

The tissue concentration of $^{14}$C (dpm/mg) was significantly higher in the shoot than in other fractions when applied to the shoot in both species (Table 9), while there were no differences in $^{14}$C concentration among the fractions when applied to roots and tuber. Relatively, the tuber had the least $^{14}$C concentration in both species.
Table 5. Effect of site of application on absorption and translocation of $^{14}C$-chlorimuron in yellow nutsedge propagules over a period of 48 hours.

<table>
<thead>
<tr>
<th>Site of application</th>
<th>Time (hours)</th>
<th>Absorption (% of recovered)</th>
<th>Translocation (% of absorbed)</th>
<th>Site mean</th>
<th>Absorption Site mean</th>
<th>Translocation Site mean</th>
</tr>
</thead>
<tbody>
<tr>
<td>Shoot</td>
<td>12</td>
<td>57.78 b</td>
<td>1.39 c</td>
<td>1.39 c</td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>63.64 a</td>
<td>1.56 c</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>64.98 a 62.13 a</td>
<td>3.06 c 2.00 b</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td>Roots and tuber</td>
<td>12</td>
<td>0.68 c</td>
<td>13.86 b</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>1.42 c</td>
<td>13.94 b</td>
<td></td>
<td></td>
<td></td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>2.74 c 1.61 b</td>
<td>23.49 a 17.10 a</td>
<td></td>
<td></td>
<td></td>
</tr>
</tbody>
</table>

*a* Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.
Table 6. Effect of site of application on absorption and translocation of ^14C-chlorimuron in purple nutsedge propagules over a period of 48 hours.

<table>
<thead>
<tr>
<th>Site of Application</th>
<th>Time (hours)</th>
<th>Absorption Site mean (%)</th>
<th>Translocation Site mean (%)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Shoot</td>
<td>12</td>
<td>40.73 b</td>
<td>1.66 c</td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>51.20 a</td>
<td>2.64 c</td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>57.67 a</td>
<td>4.22 bc 2.84 b</td>
</tr>
<tr>
<td>Roots and tuber</td>
<td>12</td>
<td>1.34 c</td>
<td>3.00 c</td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>1.80 c</td>
<td>8.77 ab</td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>5.46 c 2.87 b</td>
<td>13.29 a 8.35 a</td>
</tr>
</tbody>
</table>

*Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.*
Table 7. Distribution of radioactivity in various fractions of yellow nutsedge propagules over a period of 48 hours.

<table>
<thead>
<tr>
<th>Fraction</th>
<th>Time</th>
<th>Shoot</th>
<th>Roots and tuber</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>(hours)</td>
<td>(%) of absorbed</td>
<td>(%) of absorbed</td>
</tr>
<tr>
<td>Shoot</td>
<td>12</td>
<td>98.61</td>
<td>13.86</td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>98.44</td>
<td>13.94</td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>96.94</td>
<td>23.49</td>
</tr>
<tr>
<td>Root</td>
<td>12</td>
<td>0.71</td>
<td>19.32</td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>0.78</td>
<td>18.33</td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>1.65</td>
<td>8.48</td>
</tr>
<tr>
<td>Tuber</td>
<td>12</td>
<td>0.68</td>
<td>66.82</td>
</tr>
<tr>
<td></td>
<td>24</td>
<td>0.78</td>
<td>67.73</td>
</tr>
<tr>
<td></td>
<td>48</td>
<td>1.41</td>
<td>68.03</td>
</tr>
</tbody>
</table>

LSD (0.05)\(^a\) 7.33

\(^a\)For comparing means of Site X Time X Fraction.
Table 8. Distribution of radioactivity in various fractions of purple nutsedge propagules over a period of 48 hours.

<table>
<thead>
<tr>
<th>Site of application</th>
<th>Fraction</th>
<th>Time (hours)</th>
<th>Shoot (% of absorbed)</th>
<th>Roots and tuber (% of absorbed)</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td></td>
<td>12</td>
<td>24</td>
<td>48</td>
</tr>
<tr>
<td>Shoot</td>
<td></td>
<td>98.34</td>
<td>97.36</td>
<td>95.78</td>
</tr>
<tr>
<td>Root</td>
<td></td>
<td>0.65</td>
<td>1.02</td>
<td>1.47</td>
</tr>
<tr>
<td>Tuber</td>
<td></td>
<td>1.01</td>
<td>1.62</td>
<td>2.75</td>
</tr>
<tr>
<td>LSD (0.05)(^a)</td>
<td></td>
<td></td>
<td></td>
<td>8.15</td>
</tr>
</tbody>
</table>

\(^a\)For comparing means of Site X Time X Fraction.
Table 9. Tissue concentration of radioactivity in yellow and purple nutsedge propagules.

<table>
<thead>
<tr>
<th>Site of application</th>
<th>Fraction</th>
<th>Yellow nutsedge (dpm/mg)</th>
<th>Purple nutsedge (dpm/mg)</th>
</tr>
</thead>
<tbody>
<tr>
<td>Shoot</td>
<td>Shoot</td>
<td>8022 a</td>
<td>5230 a</td>
</tr>
<tr>
<td></td>
<td>Root</td>
<td>189 b</td>
<td>118 b</td>
</tr>
<tr>
<td></td>
<td>Tuber</td>
<td>4 b</td>
<td>2 b</td>
</tr>
<tr>
<td>Roots and tuber</td>
<td>Shoot</td>
<td>52 b</td>
<td>61 b</td>
</tr>
<tr>
<td></td>
<td>Root</td>
<td>103 b</td>
<td>276 b</td>
</tr>
<tr>
<td></td>
<td>Tuber</td>
<td>10 b</td>
<td>7 b</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.
DISCUSSION

Chlorimuron treatments decreased tuber sprouting in both yellow and purple nutsedge. The increased availability of herbicide for absorption by the tuber with increased rates resulted in a gradual decrease in tuber sprouting. It is assumed that such a decrease was in part due to the toxicity of chlorimuron in the meristematic region of buds in the tuber. In contrast with these findings, alachlor (2) and metolachlor (9) have been reported to have no effect on tuber sprouting in yellow nutsedge. Perhaps this may be due to the differences in herbicidal activity.

Many shoots from the tubers in chlorimuron-treated soil failed to emerge. Those which did emerge were apparently weak and growth was severely inhibited as illustrated by shoot dry weight data. Similar findings have been reported by other workers for alachlor (2, 25), metolachlor (9), vernolate (25), and imazaquin (15) in yellow and purple nutsedge. Often the emerged shoots died, especially at the higher rates. The observed growth inhibition and death of shoots is assumed to be due to the toxicity of the chlorimuron that was absorbed and accumulated in shoots in greater quantities (Tables 5 and 6). Although both species were susceptible to chlorimuron, the results indicated some differences in susceptibility between species. Yellow nutsedge was relatively more susceptible than purple nutsedge at all rates. This difference may be due, in part, to differential absorption during emergence as revealed from the $^{14}$C-chlorimuron absorption studies (Tables 5
and 6). Apparently, absorption of shoot applied $^{14}$C-chlorimuron was greater in yellow nutsedge than in purple nutsedge.

Soil-applied herbicides, to be effective, must kill parent tubers as well as young emerging shoots. Presently available herbicides such as alachlor (2, 25), metolachlor (9), and vernolate (25) have been reported to be ineffective in killing parent tubers. In the present study previous exposure to chlorimuron at 60 g/ha had some toxic effect on parent tubers in both species (Tables 3 and 4). At the 60 g/ha rate, it is assumed that a sufficient amount of herbicide was either absorbed by tubers or moved into tubers by shoot absorption and caused toxic effects when replanted. The decrease in shoot dry weight suggests carryover herbicide injury in plants.

Both species absorbed greater amounts of $^{14}$C when applied to the shoot than to roots and tuber. Similar results have been reported in nutsedges by other workers for metolachlor (9), alachlor (3), and imazaquin (15). Apparently chlorimuron must have been able to penetrate the shoots more readily than roots and tuber. The differences in absorption between sites may be due, partly, to the differences in $^{14}$C application method. For lack of a better method, $^{14}$C was applied to roots and tuber through the nutrient solution, which resulted in a differential concentration of radioactivity between sites. However, the treatment method of this study mimics the exposure of emerging shoot to higher concentrations of soil-applied herbicide present close to the soil surface under field conditions. Despite greater shoot absorption in
both species, very little was moved out of the shoot, in part, perhaps due to the absence of a strong source to sink translocation at this early stage. The apparent basipetal movement of $^{14}$C from the shoot into other parts was most likely through the symplastic system. At the same time, $^{14}$C uptake by roots and tuber was very limited, but a considerable amount moved acropetally into the shoot, probably through the apoplastic system. However, due to the limited transpiration of the shoot, $^{14}$C movement into shoots was not as great as expected in either species.

The tissue concentration (dpm/mg) was highest in shoots in both species when the herbicide was applied to shoots, due to greater shoot absorption and limited translocation. The concentrations in roots and tuber were not different from either site of application. This suggests that the amounts translocated to roots and tuber from shoots were similar to the amounts absorbed and retained by roots and tuber. Overall, the percent distribution of radioactivity was relatively higher in tubers than in roots for both species with either site of application. However, in terms of tissue concentration, the roots had higher concentrations due to smaller mass and tubers had least due to larger mass.
LITERATURE CITED


INTRODUCTION

Yellow and purple nutsedge are troublesome perennial weeds in many parts of the world (3, 8). They propagate mainly by tubers. Tubers have several buds and can sprout several times, often producing more than one shoot at a time (2, 14, 16). The sprouting tuber produces one or more rhizomes which usually grow upward. As the rhizome emerges from the soil, a swelling develops on the rhizome below the soil surface, leading to formation of the basal bulb. The basal bulb contains meristems for leaves, rhizomes, roots, and flower structures. Rhizomes produced from the basal bulb terminate in either basal bulbs or tubers, forming a complex system (9, 13, 18, 19). A single yellow nutsedge tuber growing alone can produce about 1900 plants and 7000 tubers in a year (17).

Control methods, such as frequent tillage and mowing designed to starve underground organs, have not been effective in reducing nutsedge populations. Several new postemergence herbicides have shown promise for effective nutsedge control. However,
effectiveness of a foliar-applied herbicide depends upon its rapid absorption and translocation in biologically active form to the meristematic regions of the basal bulb, rhizomes, and tubers in sufficient quantities to kill the entire plant (12). The movement of foliar-applied herbicides within the plant is often determined by relative activity of 'source and sink' (7). The initial underground centers of high metabolic activity (parent tubers) are replaced by the new centers of actively growing roots, rhizomes, and tubers (1). Consequently, some foliar-applied herbicides which are very effective in inhibiting further growth and reproduction often fail to kill the parent tubers attached to those plants because of the inactivity of the 'metabolic sinks' in these organs to which herbicides could translocate (14).

Stoller et al. (15) reported that effectiveness of bentazon in controlling yellow nutsedge was restricted mainly to the foliage contacted by the spray, due to the absence of basipetal translocation to parent tubers and very little acropetal translocation to other plant parts. The inhibition of rhizome production and growth in yellow nutsedge by 2,4-D was due to its accumulation in rhizome tips which develop into either new shoots or tubers (4). Sprankle et al. (12) reported that glyphosate moved basipetally into the untreated shoots and developing tillers. In purple nutsedge, glyphosate translocation increased from 5% at 1 day to 19% at 4 days after application and accumulation was greater in tubers than in leaves (20). Magalhaes et al. (10) reported that movement of dicamba in purple nutsedge was both acropetal and
basipetal. Dicamba was barely detectable in the underground organs, although it passed through the rhizomes and tubers into daughter plants. The greater toxicity of imazaquin to purple nutsedge than to yellow nutsedge was partly due to greater translocation and accumulation in rhizomes of purple nutsedge (11).

Chlorimuron is a new herbicide effective on yellow nutsedge as well as many broadleaf weeds (6). However, detailed studies of the toxicity, absorption, translocation, and metabolism of chlorimuron in yellow or purple nutsedge have not been reported. Thus, this research was carried out on yellow and purple nutsedge: 1) to determine postemergence lethal rates and tuber viability after exposure to chlorimuron; and 2) to assess the degree of absorption, translocation, and metabolism of foliar-applied chlorimuron.

MATERIALS AND METHODS

Yellow nutsedge tubers were collected from the field, washed with water, and stored in plastic bags at 3°C until used. Purple nutsedge tubers were harvested from greenhouse grown plants and used without incubation. The soil used in the study was collected from the upper 15 cm of the soil profile at the Agronomy farm, Columbus, Ohio. The soil type was Celina silt loam with 3.5% organic matter. The forms of chlorimuron used were the commercial formulation and 14C-[(U)-phenyl labeled chlorimuron (specific activity of 3.47 mCi/mM and over 99% purity).
Herbicide rate studies. Five uniform tubers were planted in soil in 15 cm diameter plastic pots. The pots were watered regularly to keep the soil moist. Plants were grown in the greenhouse at 28/20 C day:night temperatures and a 14 h photoperiod. After emergence, plants were thinned to five uniform plants per pot. Herbicide rates of 0, 5, 10, 20, and 30 g ai/ha were applied to plants at the 4 to 5 leaf stage using a pot sprayer calibrated to deliver 236 l/ha. The surfactant X-77 was added to the herbicide solution at 0.2% (v/v). The control treatments consisted of the same surfactant concentration in water. Prior to spraying, the soil in the pots was covered with charcoal to limit the herbicidal activity to foliar aspects. The charcoal was removed after spraying. Visual injury ratings on a scale of zero = no injury and 100 = complete kill were made at 1, 2, 3, and 4 weeks after application (WAA). Dry weight of shoots which received spray was recorded at 4 WAA. The experiment was conducted in a randomized complete block design with four replications. Each experiment was conducted twice and the data were combined for analysis. Means were separated at the 5% level of significance using Fisher's LSD test.

Secondary shoot production and tuber viability studies. Yellow and purple nutsedge plants were cultured and treated with herbicide as described in the herbicide rate studies, except that only one tuber was planted in each 9 by 9 by 8 cm plastic pot. An experimental unit consisted of five such pots. Before treatment, the plants were thinned to one shoot per pot. Thereafter, the secondary shoots were
allowed to grow. At 4 WAA, the secondary shoots were counted, harvested, dried, and weighed. The original tubers attached to sprayed plants were recovered and replanted in herbicide-free soil in pots as before. At 3 weeks after replanting, the number of resprouted tubers, and the number and dry weight of emerged shoots were recorded. The experiment was conducted twice in a randomized complete block design with four replications. Statistical analysis was performed as described in the herbicide rate studies.

Absorption and translocation studies. The test species (one plant per pot) were grown in a soil:sand (1:1) medium in 9 by 9 by 8 cm plastic pots. Uniform plants, with at least four fully expanded leaves, were selected for treatment. A 1 cm$^2$ area in the middle of the 4th fully expanded leaf was marked on the upper surface of the leaf by placing two lanolin bands perpendicular to the midrib. A 15 ul solution containing 0.18 uCi of $^{14}$C-chlorimuron (in water) and 0.2% (v/v) of X-77 was applied within the marked area as several small droplets. The experiment was conducted in a growth chamber with 28/20 C day:night temperatures, 14 h photoperiod at 28 klux light intensity, and 80% relative humidity.

The plants were harvested at 1, 2, 4, and 8 days after application (DAA). Lanolin was removed by wiping with ashless filter paper. The treated area was cut and washed by gentle shaking for 1 minute in 10 ml of 10% methanol and then rinsed again with 5 ml of 10% methanol to remove herbicide surface residues. The plant was fractionated into treated area of treated leaf, treated leaf
above treated area, treated leaf below treated area, other leaves, basal bulb, roots, rhizomes, and tuber, then lyophilized and weighed. The lyophilized samples and lanolin wipes were combusted in a Model 306 Tri-carb sample oxidizer. The evolved $^{14}$CO$_2$ was trapped in a mixture of 7 ml of CO$_2$ absorbent (Carbo-sorb) and 11 ml of scintillation solution (Permafluor V). Two 0.5 ml aliquots of each leaf wash were taken and 15 ml of scintillation solution (Formula 963) was added to each aliquot. Radioactivity from leaf washes and oxidations was quantified by liquid scintillation spectrometry (Beckman LS 8000). Quenching was corrected by H-number. The counts per minute (cpm) were corrected for background and counting efficiency, then converted to disintegrations per minute (dpm). The dpm data are presented as percent of total $^{14}$C recovered (Absorption) and percent of total $^{14}$C absorbed (Translocation).

Metabolism studies. Plants were cultured and treated as described in the absorption and translocation studies. At 1 and 8 DAA, the plants were harvested, fractionated into treated area and remainder of the plant, then frozen in liquid nitrogen and extracted within 3 weeks. Radiolabeled material was extracted by grinding (pestle and mortar) in three 4 ml volumes of 90% acetone. The extractions were pooled and centrifuged. A 200 ul sample of extract was spotted on a silica gel (250 um) precoated, 20 by 20 cm TLC sheet. Labeled chlorimuron was also spotted on each TLC sheet as a standard. The chromatograms were developed in dichloromethane : acetone : 1%
acetic acid (95:4:1, v/v/v). After development, the TLC sheet was divided into 1 cm horizontal bands and each band cut directly into scintillation vials and counted. Radioactivity was quantified by liquid scintillation spectrometry. Since the objective was to determine whether the parent compound was degraded, the metabolite was identified only by its Rf value. The results are presented as percent of 14C chromatographed.

RESULTS

Herbicide rates. Both yellow and purple nutsedge were sensitive to chlorimuron and exhibited similar injury symptoms. The injury was evidenced by stunted growth followed by leaf discoloration and death. Discoloration began along the midrib and later extended to either side of the midrib. Severe injury resulted in complete leaf discoloration and death beginning at the leaf tip and progressing towards the leaf base. The symptoms first appeared on young leaves and later spread to older leaves. In purple nutsedge, the dead tissue around the leaf base turned black forming a characteristic band.

Chlorimuron treatments caused 18 to 31% injury in yellow nutsedge (Figure 1) and 10 to 24% injury in purple nutsedge (Figure 2) at 1 WAA. Increased injury paralleled herbicide rate. However, the increase was greater between 5 to 10 g/ha than 10 to 20 g/ha. The increase between 20 to 30 g/ha was not significant. A similar
Figure 1. Response of yellow nutsedge to given rates of chlorimuron at 1, 2, 3, and 4 weeks after application (WAA).
Figure 2. Response of purple nutsedge to given rates of chlorimuron at 1, 2, 3, and 4 weeks after application (WAA).
trend was noticed at 2, 3, and 4 WAA in both species. At 4 WAA, even the lowest rate of 5 g/ha caused considerable injury in both species. An application of 20 g/ha gave complete kill of purple nutsedge and 84% injury in yellow nutsedge. Yellow and purple nutsedge growth was significantly reduced at all rates as compared to control (Figures 3 and 4), but differences among the rates were not significant in either species.

Secondary shoot production and tuber viability. Chlorimuron treatments, regardless of rates, inhibited secondary shoot production during 4 weeks after postemergence application and also inhibited resprouting of parent tubers when replanted (Tables 10 and 11). In purple nutsedge, although some secondary shoots were produced at 5 and 10 g/ha, they were not significant compared to the untreated control. About 13% of the parent tubers did resprout at the lowest rate in both species; however, the shoots exhibited stunted growth and had significantly lower dry weight as compared to control.

Absorption and translocation. Over 92% of the total applied radioactivity was recovered in these studies. The means for absorption and translocation data were described by a general linear model of the form \( Y = a + bx \). The reported goodness of fit (\( R^2 \)) for these regression models was defined as 1.0 minus the ratio of the residual sum of squares to the total sum of squares. Absorption of \(^{14}C\)-chlorimuron was rapid and increased from 13% at
Figure 3. Effect of given rates of chlorimuron on yellow nutsedge shoot dry weight at 4 weeks after application.
Figure 4. Effect of given rates of chlorimuron on purple nutsedge shoot dry weight at 4 weeks after application.
Table 10. Effect of postemergence application of chlorimuron on yellow nutsedge secondary shoot production and tuber viabilitya.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>4 weeks after postemergence application</th>
<th>3 weeks after replanting parent tubers</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Secondary shoots</td>
<td>Shoots</td>
</tr>
<tr>
<td></td>
<td>Emerged (no/tuber)</td>
<td>Resprouting (%)</td>
</tr>
<tr>
<td>0</td>
<td>7 a</td>
<td>70 a</td>
</tr>
<tr>
<td>5</td>
<td>0 b</td>
<td>13 b</td>
</tr>
<tr>
<td>10</td>
<td>0 b</td>
<td>0 b</td>
</tr>
<tr>
<td>20</td>
<td>0 b</td>
<td>8 b</td>
</tr>
<tr>
<td>30</td>
<td>0 b</td>
<td>0 b</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.
Table 11. Effect of postemergence application of chlorimuron on purple nutsedge secondary shoot production and tuber viability.

<table>
<thead>
<tr>
<th>Rate (g/ha)</th>
<th>4 weeks after postemergence application</th>
<th>3 weeks after replanting parent tubers</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Secondary shoots</td>
<td>Shoots</td>
</tr>
<tr>
<td></td>
<td>Emerged (no/tuber)</td>
<td>Re-sprouting (no/tuber)</td>
</tr>
<tr>
<td></td>
<td>Dry weight (mg/tuber)</td>
<td>Emerged (no/tuber)</td>
</tr>
<tr>
<td></td>
<td></td>
<td>Dry weight (mg/tuber)</td>
</tr>
<tr>
<td>0</td>
<td>3.8 a</td>
<td>90 a</td>
</tr>
<tr>
<td>5</td>
<td>0.4 b</td>
<td>13 b</td>
</tr>
<tr>
<td>10</td>
<td>0.3 b</td>
<td>0 b</td>
</tr>
<tr>
<td>20</td>
<td>0.0 b</td>
<td>0 b</td>
</tr>
<tr>
<td>30</td>
<td>0.0 b</td>
<td>0 b</td>
</tr>
</tbody>
</table>

*aMeans within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.*
1 DAA to 27% at 8 DAA in yellow nutsedge (Figure 5). A similar trend was noticed in purple nutsedge, except that absorption was slightly higher at 2, 4, and 8 DAA (Figure 6). $^{14}$C-chlorimuron translocated rapidly and the amount that moved out of the treated area increased with time in both species (Figures 7 and 8). At 8 DAA, the amount translocated was 31% in yellow nutsedge and 36% in purple nutsedge.

Distribution of radioactivity in different plant fractions revealed that translocation was both acropetal and basipetal (Tables 12 and 13). Movement of $^{14}$C into the leaf above the treated area represents acropetal translocation. Movement into all other fractions constitutes basipetal translocation. Over 68% of the absorbed $^{14}$C in yellow nutsedge and 63% in purple nutsedge remained in the treated area at 8 DAA. In both species, the distribution of radioactivity in the treated area and above the treated area differed significantly from each other. Furthermore, they accumulated significantly greater amounts of $^{14}$C than all other fractions. Overall, the basal bulb, rhizomes, and tuber had accumulated the least amounts of $^{14}$C in both species. The amounts of $^{14}$C decreased in the treated area, and increased in the above the treated area and below the treated area from 1 DAA to 8 DAA. Based on tissue concentration (dpm/mg tissue dry weight) the treated area and the above the treated area differed significantly from each other (Table 14). Further they had significantly greater tissue concentration than the rest of the fractions. In general, the tuber had the least concentration in both species.
Figure 5. Absorption of $^{14}$C-chlorimuron in yellow nutsedge over a period of 8 days.
Figure 6. Absorption of $^{14}$C-chlorimuron in purple nutsedge over a period of 8 days.
Figure 7. Translocation of $^{14}$C-chlorimuron in yellow nutsedge over a period of 8 days.
Figure 8. Translocation of $^{14}$C-chlorimuron in purple nutsedge over a period of 8 days.
Table 12. Distribution of radioactivity in various fractions of yellow nutsedge at 1, 2, 4, and 8 days after application.

<table>
<thead>
<tr>
<th>Fraction</th>
<th>days after application (%) of absorbed</th>
<th>1</th>
<th>2</th>
<th>4</th>
<th>8</th>
</tr>
</thead>
<tbody>
<tr>
<td>Treated area</td>
<td></td>
<td>84.09</td>
<td>79.54</td>
<td>75.58</td>
<td>68.71</td>
</tr>
<tr>
<td>Above treated area</td>
<td></td>
<td>5.17</td>
<td>12.77</td>
<td>16.27</td>
<td>19.08</td>
</tr>
<tr>
<td>Below treated area</td>
<td></td>
<td>1.72</td>
<td>3.05</td>
<td>4.17</td>
<td>5.94</td>
</tr>
<tr>
<td>Other leaves</td>
<td></td>
<td>2.96</td>
<td>1.62</td>
<td>1.35</td>
<td>5.42</td>
</tr>
<tr>
<td>Basal bulb</td>
<td></td>
<td>0.19</td>
<td>0.46</td>
<td>0.13</td>
<td>0.23</td>
</tr>
<tr>
<td>Roots</td>
<td></td>
<td>4.91</td>
<td>2.37</td>
<td>2.08</td>
<td>0.46</td>
</tr>
<tr>
<td>Rhizomes</td>
<td></td>
<td>0.44</td>
<td>0.09</td>
<td>0.22</td>
<td>0.09</td>
</tr>
<tr>
<td>Tuber</td>
<td></td>
<td>0.52</td>
<td>0.10</td>
<td>0.20</td>
<td>0.07</td>
</tr>
</tbody>
</table>

LSD (0.05)

6.98

\(^a\)For comparing means of Day X Fraction.
Table 13. Distribution of radioactivity in various fractions of purple nutsedge at 1, 2, 4, and 8 days after application.

<table>
<thead>
<tr>
<th>Fraction</th>
<th>1</th>
<th>2</th>
<th>4</th>
<th>8</th>
</tr>
</thead>
<tbody>
<tr>
<td>Treated area</td>
<td>81.75</td>
<td>73.76</td>
<td>69.53</td>
<td>63.76</td>
</tr>
<tr>
<td>Above treated area</td>
<td>9.94</td>
<td>15.70</td>
<td>19.81</td>
<td>27.40</td>
</tr>
<tr>
<td>Below treated area</td>
<td>3.30</td>
<td>4.72</td>
<td>5.14</td>
<td>6.36</td>
</tr>
<tr>
<td>Other leaves</td>
<td>1.58</td>
<td>1.71</td>
<td>2.31</td>
<td>0.95</td>
</tr>
<tr>
<td>Basal bulb</td>
<td>0.25</td>
<td>0.34</td>
<td>0.21</td>
<td>0.29</td>
</tr>
<tr>
<td>Roots</td>
<td>2.10</td>
<td>2.68</td>
<td>1.88</td>
<td>0.51</td>
</tr>
<tr>
<td>Rhizomes</td>
<td>0.36</td>
<td>0.12</td>
<td>0.27</td>
<td>0.40</td>
</tr>
<tr>
<td>Tuber</td>
<td>0.72</td>
<td>0.97</td>
<td>0.85</td>
<td>0.33</td>
</tr>
</tbody>
</table>

LSD (0.05)\textsuperscript{a} 5.92

\textsuperscript{a}For comparing means of Day X Fraction.
Table 14. Tissue concentration of radioactivity in various fractions of yellow and purple nutsedge.

<table>
<thead>
<tr>
<th>Fractions</th>
<th>Yellow nutsedge</th>
<th>Purple nutsedge</th>
</tr>
</thead>
<tbody>
<tr>
<td>(dpm/mg)</td>
<td></td>
<td></td>
</tr>
<tr>
<td>Treated area</td>
<td>8200 a</td>
<td>7064 a</td>
</tr>
<tr>
<td>Above treated area</td>
<td>1401 b</td>
<td>1139 b</td>
</tr>
<tr>
<td>Below treated area</td>
<td>196 c</td>
<td>251 c</td>
</tr>
<tr>
<td>Other leaves</td>
<td>14 c</td>
<td>4 c</td>
</tr>
<tr>
<td>Basal bulb</td>
<td>19 c</td>
<td>7 c</td>
</tr>
<tr>
<td>Roots</td>
<td>12 c</td>
<td>8 c</td>
</tr>
<tr>
<td>Rhizomes</td>
<td>10 c</td>
<td>5 c</td>
</tr>
<tr>
<td>Tuber</td>
<td>5 c</td>
<td>1 c</td>
</tr>
</tbody>
</table>

Means within a column followed by the same letter are not significantly different at the 5% level as determined by Fisher's LSD test.
Metabolism. Recovery of the applied label by extraction of the plant tissue was over 95% for the treated area and over 60% for the rest of the plant. Thin layer chromatographic analysis revealed two spots, with Rf values of 0.00 and 0.57. The spot at Rf 0.57 was unaltered chlorimuron. The metabolite(s) at Rf 0.00 appears to be polar. Yellow and purple nutsedge seem to be slow in metabolizing chlorimuron since less than 4% was degraded in yellow nutsedge and less than 13% in purple nutsedge within 1 DAA (Table 15). Degradation continued in both species, but did not exceed 15% in yellow nutsedge or 26% in purple nutsedge by 8 DAA.

DISCUSSION

The toxicity of chlorimuron became slowly apparent in yellow and purple nutsedge following foliar application. The plants exhibited stunted growth and leaf discoloration within 1 WAA, but complete kill of plants generally took 2 to 4 WAA regardless of rates. The relationship between chlorimuron rates and nutsedge injury followed a typical dose-response curve (Figures 1 and 2). The injury increased markedly with herbicide rates up to 20 g/ha. Shoot dry weight reductions remained quite static at all rates in both species (Figures 3 and 4). Apparently, chlorimuron treatments, regardless of rates, disrupted normal plant growth as evident by stunted plant growth, leaf discoloration, and death of shoot apex. Chlorimuron injury was similar in both species at 1 WAA, but
Table 15. Chlorimuron metabolism in yellow and purple nutsedge at 1 and 8 days after application.

<table>
<thead>
<tr>
<th>Fraction</th>
<th>Time (day)</th>
<th>Yellow nutsedge Rf 0.00</th>
<th>Yellow nutsedge Rf 0.57</th>
<th>Purple nutsedge Rf 0.00</th>
<th>Purple nutsedge Rf 0.57</th>
</tr>
</thead>
<tbody>
<tr>
<td>Treated area</td>
<td>1</td>
<td>3.0</td>
<td>97.0</td>
<td>10.6</td>
<td>89.4</td>
</tr>
<tr>
<td></td>
<td>8</td>
<td>11.8</td>
<td>88.2</td>
<td>22.9</td>
<td>77.1</td>
</tr>
<tr>
<td>Rest of the plant</td>
<td>1</td>
<td>3.9</td>
<td>96.1</td>
<td>12.6</td>
<td>87.4</td>
</tr>
<tr>
<td></td>
<td>8</td>
<td>14.4</td>
<td>85.6</td>
<td>25.3</td>
<td>74.7</td>
</tr>
</tbody>
</table>

*Rf 0.57 corresponds to chlorimuron ethyl and Rf 0.0 a metabolite(s).*

*Values are means of three plant samples.*
thereafter injury increased sharply in purple nutsedge. Overall, chlorimuron was relatively more toxic to purple nutsedge than to yellow nutsedge. This may be due, in part, to greater herbicide absorption and translocation in purple nutsedge than in yellow nutsedge.

Foliar-applied chlorimuron, to be effective, must reach the meristems of basal bulb, rhizomes, and tuber (buds) in lethal amounts and prevent further growth. In the present study, it was observed that chlorimuron, regardless of rates, inhibited secondary shoot production and killed parent tubers in both yellow and purple nutsedge.

Results of the absorption and translocation studies demonstrated that the $^{14}$C translocated out of the treated area was proportional to that absorbed into the leaf in both species. This suggests that more herbicide would translocate if it were absorbed. Generally, acropetal movement was higher than basipetal movement in both species. The greater acropetal movement seems to be the reason for discoloration symptoms to begin first at the leaf tip. The lesser basipetal movement may be due, in part, to restricted herbicide movement by the disconnected accessory bundles and/or by immature vascular elements at the leaf base (18, 19).

The radioactivity was distributed within all plant parts. The movement of $^{14}$C into active meristems such as basal bulb, rhizomes, and roots of nutsedges is determined by the relative activity of 'source' and 'sink' (1, 7). Although the basal bulb is the center of high meristematic activity, the $^{14}$C in the basal
bulb of either species was barely detectable, possibly due to $^{14}$C diversion into rhizomes, roots, tuber, and other leaves as well. It is assumed that the inhibition of secondary shoot production (Tables 10 and 11) resulted from the toxicity of chlorimuron that accumulated in the basal bulb and rhizomes, therefore, inhibiting production of new rhizomes from the basal bulbs or growth of existing rhizomes. Accumulation of $^{14}$C in tubers may have been poor due to the inactivity of 'metabolic sinks' (1, 14). The tuber had the least tissue concentration, apparently due to greater tuber mass. However, the herbicide killed the parent tuber of the plant receiving the spray (Tables 10 and 11). This was partly due to the general decline in parent tuber vigor, as it had already sprouted once which depleted its food reserves (14), and coupled with the toxic effect of chlorimuron that accumulated in the tuber. Therefore, chlorimuron appears to have potential for suppressing spread of these species by exerting toxic effects on rhizomes and parent tubers.

Thin layer chromatographic analysis demonstrated that yellow and purple nutsedge were slow in metabolizing chlorimuron. The differences in metabolic inactivation of chlorimuron in other plant species have been reported (5). The sensitive species such as cocklebur and redroot pigweed metabolize chlorimuron slowly (half-lives over 30 h), while the tolerant species such as soybeans metabolize it rapidly (1 and 3 h). Overall susceptibility of yellow and purple nutsedge to chlorimuron can perhaps be attributed to this slow metabolic inactivation.
LITERATURE CITED


YELLOW NUTSEDGE (*Cyperus esculentus*) CONTROL
IN SOYBEANS WITH CHLORIMURON

INTRODUCTION

Yellow nutsedge is a persistent weed in the United States (3, 9, 11, 15) and throughout the world (3, 9). It infests many agronomic and horticultural crops. In the United States, yellow nutsedge together with purple nutsedge rank fifth in seriousness among all weed species and second only to quackgrass as perennial weed problems (9). In 1975, it was estimated that approximately 1.7 million hectares (12.6%) of soybeans were infested with yellow nutsedge in the North Central Region of the United States (1) and the weed is still spreading. The ability to reproduce by tubers and rhizomes, the removal of annual weed competition with increased use of effective herbicides, and erratic performance of herbicides in control of yellow nutsedge, all acting together, probably have contributed to its rapid spread.

Yellow nutsedge infestations can seriously reduce crop yields. In soybeans, yield losses of up to 87% have been reported due to yellow nutsedge alone (10, 12, 17).
Among the soil-applied herbicides, alachlor and metolachlor seem to provide satisfactory control of yellow nutsedge in soybeans (2, 7, 17). However, they are more effective with preplant incorporation than as preemergence applications. On coarse textured soils their performance is often erratic (13). Control of yellow nutsedge in soybeans with foliar-applied herbicides is rather inconsistent. Bentazon selectively controls yellow nutsedge, when applied in split applications 10 to 14 days apart with good coverage of the foliage by the spray solution (14, 16). Yellow nutsedge is sensitive to shade. Nutsedge control was better in soybeans when planted in narrow rows as compared to wide rows (4, 7). Therefore, chemical control coupled with planting soybeans in narrow rows can provide effective nutsedge control.

Chlorimuron is a new herbicide for selective weed control in soybeans. It controls many broadleaf weeds and appears to be effective on yellow nutsedge (5, 6, 8). It is active both in preemergence and postemergence applications. When applied postemergence, chlorimuron is active at much lower rates (4 to 17.5 g/ha) than when applied preemergence (35 to 210 g/ha). The objective of this two year field study was to evaluate efficacy of chlorimuron to control yellow nutsedge in 25 and 76 cm row soybeans.
MATERIALS AND METHODS

Yellow nutsedge control in the field was evaluated at Canal Winchester and Columbus, Ohio, in 1985 and 1986, respectively. The soil type at Canal Winchester (site I) was Brookston silty clay loam with 4.5% organic matter, whereas the soil type at Columbus (site II) was Celina silt loam with 3.5% organic matter. The areas were naturally infested with yellow nutsedge. The land was plowed, disked, and then leveled with an S-tine cultivator during the spring. Soybeans (cv GL-3610) were planted in late May in 25 and 76 cm row spacings using conventional equipment. The plots were 3 m wide and 7 m long. The treatments were replicated three times in a split plot design with row spacing as main plots.

Metolachlor and bentazon were included in the study as standard treatments for comparison. Preplant herbicides were applied just before planting, then all plots, including those not treated, were immediately incorporated with a Danish S-tine cultivator with two rolling basket levelers to a depth of 5 to 7 cm. Preemergence applications were made within a few hours after soybean planting. Postemergence applications were made on June 26, 1985, and June 21, 1986, when soybeans were at the third trifoliate stage and yellow nutsedge at the 4 to 6 leaf stage. A surfactant, X-77, was added to the spray solutions at 0.2% (v/v) for postemergence applications. Metribuzin at 0.28 kg/ha and chloramben at 2.27 kg/ha in a tank mix were applied preemergence to all plots after soybean planting for general grass and broadleaf weed control. All herbicides were
applied broadcast using a CO₂ backpack sprayer calibrated to deliver 236 l/ha. After planting, no further cultivation was employed.

Yellow nutsedge control was rated 2 and 4 weeks after the postemergence application by visually evaluating control in the treated compared to untreated plots. A rating scale of zero = no control and 100 = complete control was used.

RESULTS AND DISCUSSION

Yellow nutsedge control was better in the 1986 season than in the 1985 season (Table 16). The differences in soil type and rainfall probably influenced the degree of yellow nutsedge control. Site I (1985) had Brookston silty clay loam with 4.5% organic matter, whereas site II (1986) had Celina silt loam with 3.5% organic matter. Rainfall during the 7 days after soil application was 1.42 cm in 1985 and 2.97 cm in 1986. Perhaps the lower organic matter and adequate rainfall for herbicide incorporation contributed to better performance of the soil applied herbicides in 1986.

Nutsedge control did not differ significantly between row spacings in either year. Other workers have reported better nutsedge control in narrow row spacing than in wide row spacing (4, 7). This may be due, in part, to the differences in locations, varieties, etc. The nutsedge control in row spacings was rated with

<table>
<thead>
<tr>
<th>Herbicide</th>
<th>Rate</th>
<th>Application method</th>
<th>1985</th>
<th>1986</th>
<th>1985</th>
<th>1986</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>(kg/ha)</td>
<td></td>
<td>2 WAPA</td>
<td>4 WAPA</td>
<td>2 WAPA</td>
<td>4 WAPA</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.040</td>
<td>PRE</td>
<td>63</td>
<td>61</td>
<td>81</td>
<td>81</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.060</td>
<td>PRE</td>
<td>68</td>
<td>67</td>
<td>93</td>
<td>93</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.080</td>
<td>PRE</td>
<td>81</td>
<td>80</td>
<td>98</td>
<td>98</td>
</tr>
<tr>
<td>Metolachlor</td>
<td>3.405</td>
<td>PPI</td>
<td>33</td>
<td>33</td>
<td>63</td>
<td>63</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.005</td>
<td>POST</td>
<td>35</td>
<td>56</td>
<td>47</td>
<td>55</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.015</td>
<td>POST</td>
<td>40</td>
<td>72</td>
<td>65</td>
<td>72</td>
</tr>
<tr>
<td>Chlorimuron</td>
<td>0.025</td>
<td>POST</td>
<td>46</td>
<td>78</td>
<td>66</td>
<td>82</td>
</tr>
<tr>
<td>Bentazon</td>
<td>1.135</td>
<td>POST</td>
<td>26</td>
<td>50</td>
<td>55</td>
<td>60</td>
</tr>
<tr>
<td>Control</td>
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<td></td>
<td>0</td>
<td>0</td>
<td>0</td>
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</tr>
<tr>
<td>LSD (0.05)</td>
<td></td>
<td></td>
<td>22</td>
<td>22</td>
<td>9</td>
<td>8</td>
</tr>
</tbody>
</table>

Row spacing

<table>
<thead>
<tr>
<th></th>
<th>1985</th>
<th>1986</th>
</tr>
</thead>
<tbody>
<tr>
<td>25 cm</td>
<td>41</td>
<td>56</td>
</tr>
<tr>
<td>76 cm</td>
<td>45</td>
<td>54</td>
</tr>
<tr>
<td>LSD (0.05)</td>
<td>NS</td>
<td>NS</td>
</tr>
</tbody>
</table>

---

*a Weeks after postemergence application (WAPA).
reference to their respective controls. A rating with reference to a common control might have revealed the row effects on nutsedge control.

Preemergence applications of chlorimuron gave significantly better nutsedge control than metolachlor, possibly due to differences in herbicidal activity. Among the rates, preemergence application of chlorimuron at 0.080 kg/ha gave over 80% control in both years at both rating dates. In 1986, all three preemergence rates gave over 81% control at both rating dates. Further, differences in control between the 0.060 and 0.080 kg/ha rates were not significant, but both gave significantly better control than the 0.040 kg/ha rate. Chlorimuron at 0.040 kg/ha had no apparent toxic effect on soybeans. At 0.060 kg/ha and above, although initially soybeans were slightly injured, they exhibited ability to recover.

At 4 weeks after postemergence applications, chlorimuron at 0.015 kg/ha and above gave significantly greater control than bentazon in both years. At the second rating, chlorimuron at 0.025 kg/ha was significantly better than 0.005 kg/ha in 1985, whereas all three rates differed significantly from one another in 1986. Chlorimuron at 0.005 kg/ha and bentazon were ineffective in controlling nutsedge in either year. At 0.025 kg/ha, chlorimuron injured soybeans; however, they recovered from apparent injury within a week.
LITERATURE CITED


CONCLUSION

Experiments were conducted in the greenhouse, growth chamber, laboratory, and field to investigate the effects of soil and foliar applied chlorimuron on yellow and purple nutsedge. Soil-applied chlorimuron (10 to 60 g/ha) significantly decreased tuber sprouting, shoot emergence, and shoot growth in both yellow and purple nutsedge in greenhouse studies. Chlorimuron at 20 g/ha on yellow nutsedge and at 40 g/ha on purple nutsedge gave excellent control. Previous exposure to chlorimuron was not effective in reducing parent tuber resprouting at any rate used in yellow nutsedge, while in purple nutsedge parent tuber resprouting was reduced significantly at 60 g/ha. Previous exposure to chlorimuron had no effect on shoot emergence regardless of rates, but shoot dry weight decreased significantly in both species at 60 g/ha rate.

Absorption and translocation of $^{14}$C-chlorimuron in young nutsedge propagules differed between sites of application. Absorption of $^{14}$C was rapid and higher when applied to shoots than when applied to roots and tubers in both species. Shoot absorption at 24 and 48 hours after application was significantly higher than at 12 h in both species. Amounts of $^{14}$C absorbed by roots and tubers did not differ significantly through time in either species. In both yellow and purple nutsedge, translocation
of $^{14}$C was greater from the roots and tuber than from the shoot. Most of the shoot-absorbed $^{14}$C in both species remained in the shoot. Of the $^{14}$C absorbed by roots and tuber, the amount retained by tubers was quite static while that absorbed by the roots was translocated in essentially equivalent amounts to the shoots. The tissue concentration of $^{14}$C (dpm/mg) was significantly greater in the shoot than in other fractions when application was made to the shoot. However, differences among the fractions were not significant when application was made to the roots and tuber.

Foliar-applied chlorimuron (5 to 30 g/ha) was toxic to both yellow and purple nutsedge. Toxicity was evidenced by stunted growth, leaf discoloration, and death. Symptoms first appeared on young leaves and later spread to older leaves. The dead tissue around the leaf base turned black in purple nutsedge, forming a characteristic band. The relationship between chlorimuron rates and injury followed a typical dose-response curve. Injury increased with rates up to 20 g/ha at all evaluation dates. Application of chlorimuron at 20 g/ha gave 84% control in yellow nutsedge and 100% control in purple nutsedge. Chlorimuron treatments reduced shoot dry weight and inhibited secondary shoot production at all rates in both species. Chlorimuron at all rates killed the parent tubers of yellow and purple nutsedge attached to plants receiving the spray.

Foliar-applied $^{14}$C-chlorimuron was rapidly absorbed and translocated in yellow and purple nutsedge. Over 13% of the $^{14}$C
recovered was absorbed and over 15% of the absorbed was translocated at 1 DAA in both species. This increased two-fold by 8 DAA. Translocation was both acropetal and basipetal. Over 68% of the absorbed $^{14}C$ in yellow nutsedge and 63% in purple nutsedge remained in the treated area at 8 DAA. In both species, the distribution of radioactivity as well as tissue concentration in the treated area and above the treated area differed significantly from each other. Further, they had significantly greater amounts of $^{14}C$ than all other fractions. Overall, the basal bulb, rhizomes, and tuber had the least amounts of $^{14}C$ in both species.

Thin layer chromatographic analysis of 90% acetone extracts of plant tissue revealed two spots. The spot at Rf 0.0 appears to be a polar metabolite(s). Yellow and purple nutsedge were slow in metabolizing chlorimuron. Degradation did not exceed 15% in yellow nutsedge or 26% in purple nutsedge at 8 DAA. Overall susceptibility of yellow and purple nutsedge to chlorimuron appears to be due to slow metabolic inactivation.

Under field conditions, postemergence application of chlorimuron was more consistent than preemergence application in yellow nutsedge control. The performance of preemergence application was influenced by soil type and weather conditions. Preemergence application of chlorimuron at 0.080 kg/ha gave at least 80% control and postemergence application at 0.025 kg/ha gave at least 78% control.
The results of these studies demonstrated that soil and foliar applied chlorimuron was toxic to both yellow and purple nutsedge. Differences in susceptibility between species were observed. Yellow nutsedge was relatively more susceptible than purple nutsedge when chlorimuron was applied to soil, while purple nutsedge was more susceptible than yellow nutsedge when applied to foliage. The differences in absorption and translocation of chlorimuron between sites of application in young propagules suggest a need for incorporation of chlorimuron for maximum effectiveness under field conditions. Postemergence application of chlorimuron was more effective in killing parent tubers in both species and consistent in yellow nutsedge control under field conditions than as preemergence application. Thus, postemergence application of chlorimuron can be preferred to preemergence application under certain conditions.

Since chlorimuron appears to be a very promising herbicide for yellow and purple nutsedge control, more research needs to be done to develop a comprehensive nutsedge control program in general. The areas suggested for further research include: 1) effect of soil pH and organic matter on the performance of soil-applied chlorimuron, and herbicide carryover in soil; 2) effect of temperature and relative humidity on the performance of foliar-applied chlorimuron; 3) activity of chlorimuron at different growth stages of yellow and purple nutsedge; and 4) positive identification of metabolite(s) of the parent compound.


## APPENDIX

Composition of the modified Hoagland's nutrient solution used in isotope studies.

<table>
<thead>
<tr>
<th>Stock Solution</th>
<th>Preparation of nutrient solution (ul of stock/1)</th>
<th>Concentration of nutrient solution (uM)</th>
</tr>
</thead>
<tbody>
<tr>
<td>1 M Ca(NO$_3$)$_2$·4H$_2$O</td>
<td>5</td>
<td>5</td>
</tr>
<tr>
<td>1 M KH$_2$PO$_4$</td>
<td>2</td>
<td>2</td>
</tr>
<tr>
<td>1 M MgSO$_4$·7H$_2$O</td>
<td>4</td>
<td>4</td>
</tr>
<tr>
<td>1 M (NH$_4$)$_2$SO$_4$</td>
<td>1</td>
<td>1</td>
</tr>
<tr>
<td>1 M KNO$_3$</td>
<td>5</td>
<td>5</td>
</tr>
<tr>
<td>Iron chelate$^a$</td>
<td>1</td>
<td>-</td>
</tr>
<tr>
<td>Micronutrients$^b$</td>
<td>1</td>
<td>-</td>
</tr>
</tbody>
</table>

$^a$Iron chelate stock contained 72 g/l of sequestrene (Geigy Chemical Corp., Ardsley, New York).

$^b$Micronutrients stock solution contained the following components per liter: 2.86 g H$_2$BO$_3$; 1.81 g MnCl$_2$·4H$_2$O; 0.22 g ZnSO$_4$·7H$_2$O; 0.08 g CuSO$_4$·5H$_2$O; and 0.02 g (NH$_4$)$_2$MoO$_4$·4H$_2$O.