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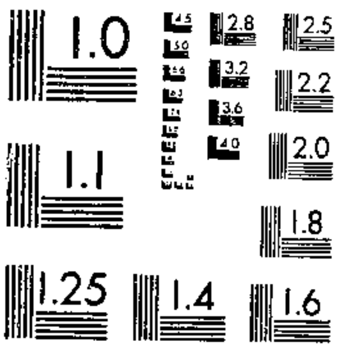

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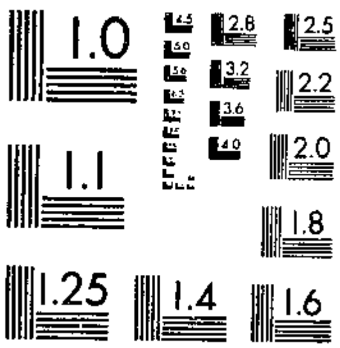

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Yellow Nutsedge

A Menace in the Corn Belt

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Abstract

Yellow nutsedge, considered one of the worst weeds throughout the world, has rapidly spread in the United States. It is a major weed problem in crops. Growing on all soil types, including peat and sand, it infests more than 10 percent of the cropland in the North Central and North East regions. No single measure is available that will control yellow nutsedge for an entire season, but it can be controlled in corn and soybeans better than in most other cultivated crops. This report summarizes the knowledge concerning its growth and control as it affects these crops in the Corn Belt.

Issued December 1981

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Yellow Nutsedge: A Menace in the Corn Belt

E. W. Stoller[1]

Introduction

Yellow nutsedge (*Cyperus esculentus* L.) is a herbaceous perennial that is rapidly spreading as a weedy pest in the United States (*70*).[2] Worldwide, it is considered one of the worst weeds (*28*). It has been troublesome for many decades in various parts of the world and has stimulated much research on its growth and control. Within recent decades, yellow nutsedge has become increasingly troublesome in agronomic crops, grown in the Midwestern United States (the Corn Belt). It is now the most troublesome perennial in most of the Corn Belt. Fortunately, technology now is available to reduce this pest to levels that will not reduce yields of these crops in most situations. This publication summarizes the knowledge concerning yellow nutsedge growth and control as it affects corn and soybean production in the Corn Belt. Much of the information also will apply to other situations where this weed is troublesome.

Distribution

Yellow nutsedge is a weed in both tropical and temperate climates of essentially all the agricultural production areas in the world (*28*). It is present in all States including Hawaii and Alaska (*48, 69*). It also grows in several provinces of southeastern Canada (*48*). Cold soil temperatures are an environmental factor that can limit the range of this weed because extreme cold can kill the tubers (*52*). This species seems to propagate exclusively by tubers, especially in cultivated areas.

Yellow nutsedge grows in a wide variety of habitats and can be found throughout the Corn Belt at various levels of infestation. It grows in both disturbed and undisturbed sites, on all soil types, including peats and sands, and tolerates a wide range of soil moisture conditions (*28, 32, 48*). In cultivated areas, dense stands often are found in low wet areas and in river bottoms. Yellow nutsedge proliferates where crops and annual weeds are sometimes flooded out, leaving a noncompetitive habitat for prolific growth. Infestations in cropland are characterized by numerous small, but densely populated areas, which vary in size from several square feet to several square yards (fig. 1). A majority of cultivated fields in the Corn Belt probably have at least a low level of yellow nutsedge infestation in them. Dense stands sometimes infest several acres or complete fields.

In 1975, estimates showed that 4.2 million acres of soybeans and 6.4 million acres of corn were infested in the North Central Region of the United States (*1*). In the Northeast United States, more than 3.5 million acres of corn were infested in 1974 (*58*). These infestations constitute more than 10 percent of this cropland. Further, the infestation trend increased in these areas from 1970 to 1975 (*1, 58*). Infestation surveys since 1975 for the entire area are not available, but farming practices that could reduce infestation in the Corn Belt have not changed sufficiently since then to halt the spread of yellow nutsedge. Growers increasingly are aware of this weed and are interested in information about its growth and control.

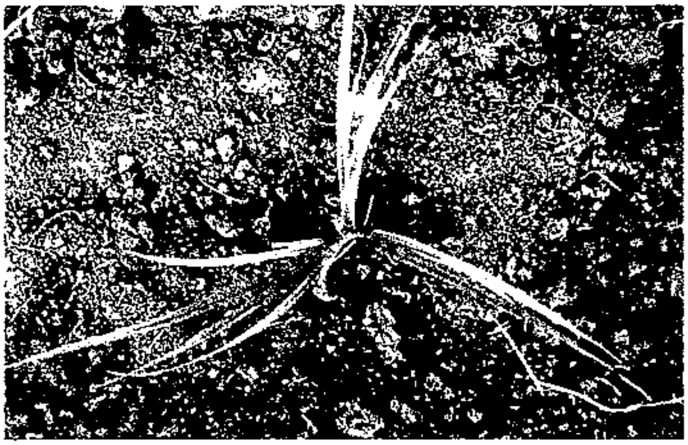

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FIGURE 2.—Top view of a yellow nutsedge plant that shows the triangular arrangement of the leaves.

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FIGURE 1.—Yellow nutsedge infestation in soybeans several weeks after planting.

Plant Description

Classification

Yellow nutsedge is a member of the Cyperaceae family (*4, 14, 20*). In addition, the plant is classified in the order Graminales, the subclass Monocotyledoneae, and the class Angiospermae. The Cyperaceae is a large family, consisting of about 75 genera and perhaps 4,000 species, most of them inhabiting the tropics and subtropics (*4, 20*). Over 450 species in the family inhabit Central-Northeastern United States and adjacent Canada (*14*). Only a few species are weeds. Yellow nutsedge and purple nutsedge (*Cyperus rotundus* L.) are the principal tuber-bearing species of the *Cyperus* genus present in the United States (*20*). The range of purple nutsedge, however, does not extend into the cold winter climates like yellow nutsedge and, currently, purple nutsedge does not grow in the Corn Belt (*52, 69*).

Members of the Cyperceae are characterized by three-ranked leaves with one-third phyllotaxy (fig. 2), and the leaves have closed sheaths around the triangular fascicle (fig. 3). Yellow nutsedge has rhizomes, tubers, leaves, and inflorescences (seed heads) that are distinctive to the species and can afford positive identification.

Taxonomically, species identification usually requires

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[2] Italic numbers in parentheses refer to Literature Cited, page 10.

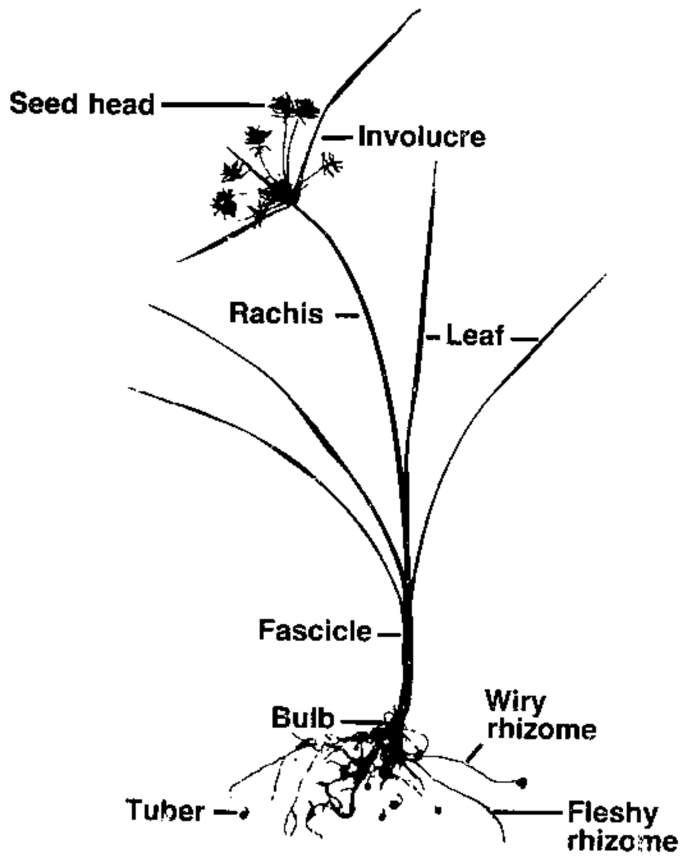

FIGURE 3.—A mature yellow nutsedge plant with the major plant parts identified (*74*).

a plant with a mature inflorescent seed head and mature achenes (seeds). The inflorescence, an umbel, is borne on a triangular scape, or stalk (fig. 4). The yellowish-brown umbel consists of several short rays and two to nine longer ones, all springing from the base at a common point. Below the umbel are three to nine involucral leaves (bracts) with characteristic one-third phyllotaxy, but they lack sheathing bases like vegetative leaves. Most of the involucral leaves are much longer than the rays of the umbel. The spikelets are strongly flattened, golden brown, about 0.3 to 1 in (1 to 3 cm) long and 0.06 to 0.12 in (1.5 to 3 mm) wide. They also are mostly four-ranked but occasionally are two-ranked (*14, 20*).

Seeds are borne individually in the form of achenes, often not well developed. Achenes are yellowish-brown, triangular, and about 0.06 in (1.5 mm) long. Small, dark spots are scattered on the glossy surface of the achenes. Each achene grows sessile (attached to the stem) on a spikelet and is covered by a thin scale. These scales are distinctly nerved, obtusely shaped, and about 0.1 in (2.5 to 3 mm) long (*74*).

Obtaining positive identification often is desirable or necessary at an early stage of growth, as well as when plants are mature but without seed heads. Fortunately, positive identification of yellow nutsedge usually can be made by observing tuber (p. 4) or leaf (p. 3) characteristics that are unique to this species.

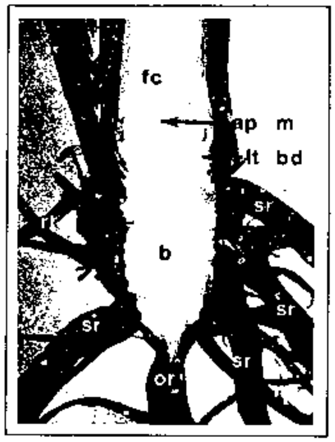

FIGURE 5.—Longitudinal section (approximately 3×) of a basal bulb (*74*) shows bulb (b), fascicle leaves (fc), original rhizome (or), secondary rhizomes (sr), apical meristem (ap m), roots (rt), and lateral bud (lt bd).

Chromosome numbers in this species have been reported as n equaling about 48 for plants from southern Canada (*48*), but 2n equaling about 108 for plants from Massachusetts (*26*).

Morphology and Anatomy

Basal bulbs. The basal bulb is considered the principal site of vegetative activity and propagation (*5, 18, 28, 31*). Basal bulbs are subterranean and contain meristems for leaves, rhizomes, roots, and flower structures. An initial basal bulb forms on the rhizome that originates from the germinating tuber. Secondary basal bulbs then develop from rhizomes that grow from the initial bulb. As the plant continues growth, bulbs of a higher order form on rhizomes from the secondary bulbs, giving rise to a complex system of rhizomes and basal bulbs that eventually produce tubers.

Basal bulbs are formed from meristematic cells of the rhizome tip and essentially consist of a short acropetal stem with compact nodes (*74*). The shoot consists of leaves developed from the 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 (fig. 5). New rhizomes originate from meristematic areas in the basal bulb at the parenchyma cells back of the tissues that develop new leaves (leaf primordia) (*74*).

Rhizomes. Rhizomes arise

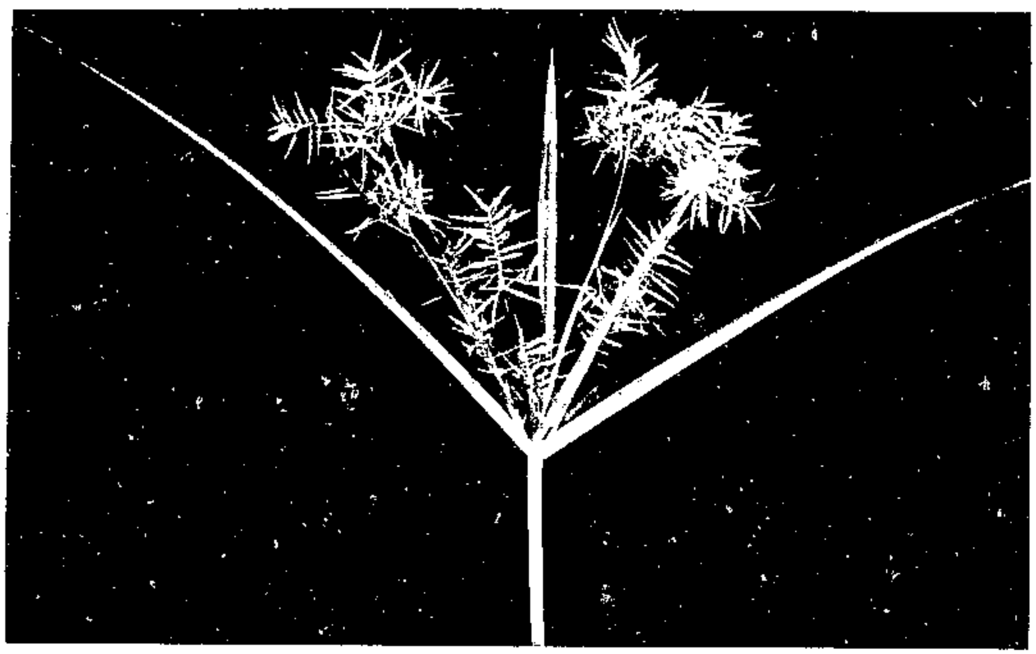

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FIGURE 4.—The inflorescence of yellow nutsedge, showing the basal attachment of the rays and involucral leaves.

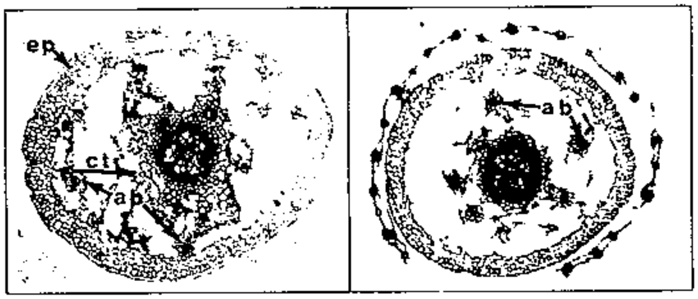

FIGURE 6.—Cross sections of young (left) and old (right) rhizomes with epidermis (ep), cortex (ct), and accessory bundle (ab). The darkened portion of the outer cortex surrounding the central vascular bundle becomes lignified as the rhizome ages. The outer cortical cells and epidermis completely slough away from the lignified cylinder that encloses the central vascular system (*74*).

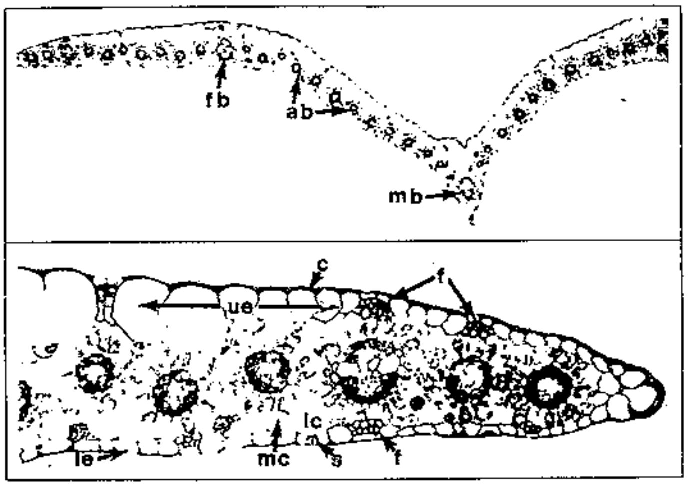

FIGURE 7.—Cross sections (upper, approximately 87×, lower, approximately 226×) of a yellow nutsedge leaf showing flank bundle (fb), median bundle (mb), and accessory bundles (ab) of vascular tissue, cuticle (c), fibers (f), upper epidermis (ue), lower epidermis (le), mechanical cells (mc), stomate (s), and lacunae (lc) (*74*).

from basal bulbs during vegetative propagation or from tubers during germination (*31, 53, 74*). The rhizomes that develop from germinating tubers appear structurally similar to those that develop from basal bulbs. A new rhizome grows as an indeterminate stem with nodes and internodes. Scarious, veined, scale-leaves about 0.1 to 0.3 in (0.5 to 1.0 cm) long are at each of the nodes, posterior to the basal bulbs. The rhizome terminal end is sharp and pointed as a result of the plicate vernation (lengthwise, fanlike folds) of the extended scale-leaves. The growing point of an indeterminate rhizome is enclosed by the terminal scale-leaves and can transform into either a basal bulb or a tuber (*74*). Rhizomes growing from germinating tubers, however, are determinate and transfer only into basal bulbs. Rhizomes grow in length primarily by internode expansion. Because branching at nodes rarely occurs, rhizomes normally terminate in either basal bulbs or tubers.

In cross section, a young rhizome has an epidermis, a cortex, and an endodermis, which surrounds the vascular cylinder (fig. 6). The vascular cylinder contains most of the vascular tissue, with the xylem outside of the phloem in the vascular bundles (*74*). Some of the xylem is scattered throughout the cortex in small auxiliary bundles. Bundles within the vascular cylinder appear to be continuous between basal bulbs and tubers (*74*).

As the rhizome matures, secondary thickening occurs within the walls of the central vascular bundles and in several layers of cortical cells located next to the endodermis (fig. 6). The thickening consists of a partial filling of the cells surrounding the vascular cylinder with lignin. Deterioration of the exterior tissues leaves a ligneous rhizome, which surrounds the vascular tissues (*74*). These rhizomes resist degradation but can transport food to developing tubers through the enclosed vascular system.

Young rhizomes are light colored with a fleshy appearance (fleshy rhizomes). As they mature, they become dark brown and wiry as they slough off their epidermal cells (wiry rhizomes) (*73*).

Leaves. Photosynthetically active leaves emerge from the basal bulb in an infolded, triangular fascicle, beginning at the outside and progressing inward (*74*). Leaves can emerge from the fascicle until the scape emerges from the center of the basal bulb inside the foliar tube (*31*). The two leaves forming the foliar tube can remain enmeshed until forced apart by the developing scape.

A leaf develops from the intercalary meristem at the leaf base in the basal bulb (*74*). The initial vascular system of the developing leaf arises from the extension of three vascular strands into the leaf primordia. Accessory vascular bundles are formed from anticlinal divisions of the initial vascular strands (*74*).

The upper leaf surface is composed of parallel rows of epidermal cells covered with a waxy cutin layer (fig. 7). Nearly all stomates are present on the lower leaf surface; those on the upper surface are near the leaf margins (*74*). Stomates on the lower surface are arranged in rows parallel with the hypodermal fibers and vascular bundles (*74*).

The vascular bundles lie adjacent, with the xylem above the phloem (*74*). Bundles are surrounded by a layer of fibers, then a layer of chlorophyllous cells. The largest vascular bundles have some specialized "Kranz" cells inside the fiber bundle (*74*). Most of the photosynthetic activity probably occurs in this outer layer of cells in the bundle sheath, if yellow nutsedge is typical of many other plants that also possess the C_4-dicarboxylic-acid-CO_2-fixation pathway.

A single layer of large and highly vacuolated epidermal cells is directly above the vascular bundles (fig. 7). A large fiber bundle that spans the length of the leaf supports these vacuolated cells (*74*). Below the bundle is another layer of highly vacuolated cells, which are interspersed with large lacunae. The collateral vascular arrangement of the leaf changes to the amphivasal arrangement of the xylem as it passes through the basal bulb (*74*).

Morphologically, the shape of the leaf tip is distinctive and often offers a means for differentiation from closely related species (*49*). In yellow nutsedge, the leaf has a "shoulder" 0.4 to 0.8 in (1 to 2 cm) from the tip, which tapers to an attenuated, needlelike point. This compares to purple nutsedge leaves, for example, which taper rather abruptly to a blunt tip (*49*).

Roots. Roots originate from endodermal tissues of tubers, basal bulbs, or rhizomes (*74*). They usually originate from all these structures but constitute only a small portion of the total plant biomass (*33*). Rhizomes, basal bulbs, and tubers dominate the subterranean part of the plant (*33*).

A root cross section reveals four large and several small, ligneous xylem vessels surrounded by three to four layers of phloem vessels (fig. 8). This vascular cylinder is sheathed by the pericycle consisting of a single layer of

FIGURE 8.—Cross section of a yellow nutsedge root (left, approximately 130×, right, approximately 560×), showing cortex (ct), endodermis (en), pericycle (pc), xylem (xy), and phloem (ph) (*74*).

parenchyma cells. The pericycle is surrounded by lignified endodermal cells. A further outer layer of cortical cells is covered by a layer of epidermal cells. As the root matures, the cortical and epidermal cells slough away and the root changes from a light fleshy color to dark brown (*74*).

Tubers. A tuber is essentially an enlarged section of rhizome with terminal buds. These buds become new plants as the tubers germinate. Mature tubers are brown to dark brown and clearly display their nodes, internodes, roots, and scale leaves (fig. 9). These features also are useful in species identification in that tubers of closely related species (purple nutsedge) differ.

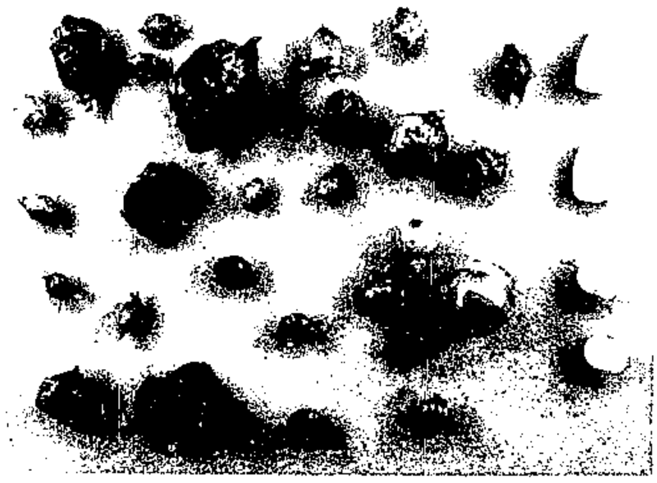

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FIGURE 9.—Various shades, shapes, and sizes of mature yellow nutsedge tubers. These tubers include those from plants found outside the Corn Belt. Corn and soybean seeds are on the right.

Under appropriate environmental conditions, tubers are differentiated from meristems at the rhizome tips. The internodes at the tip become increasingly shorter, resulting in a group of appressed leaves to form a cone (*7*). Buds are present at the bases of the terminal leaves under the canopy of this cone, one bud per node. Characteristic one-third phyllotaxy is displayed by the leaves and buds in the tuber apex (fig. 10). The buds are surrounded by leaf primordia and root initials. Five to seven buds are formed. The oldest bud is the largest and most basipetal (*7*).

Tuber formation begins in the meristematic region behind the leaf primordia (*31*, *74*). The leaf primordia remain dormant as the parenchyma cells, both inside and outside the endodermis, enlarge and accumulate starch. As the tuber enlarges, the rhizome vascular system disperses into the tuber matrix (*74*).

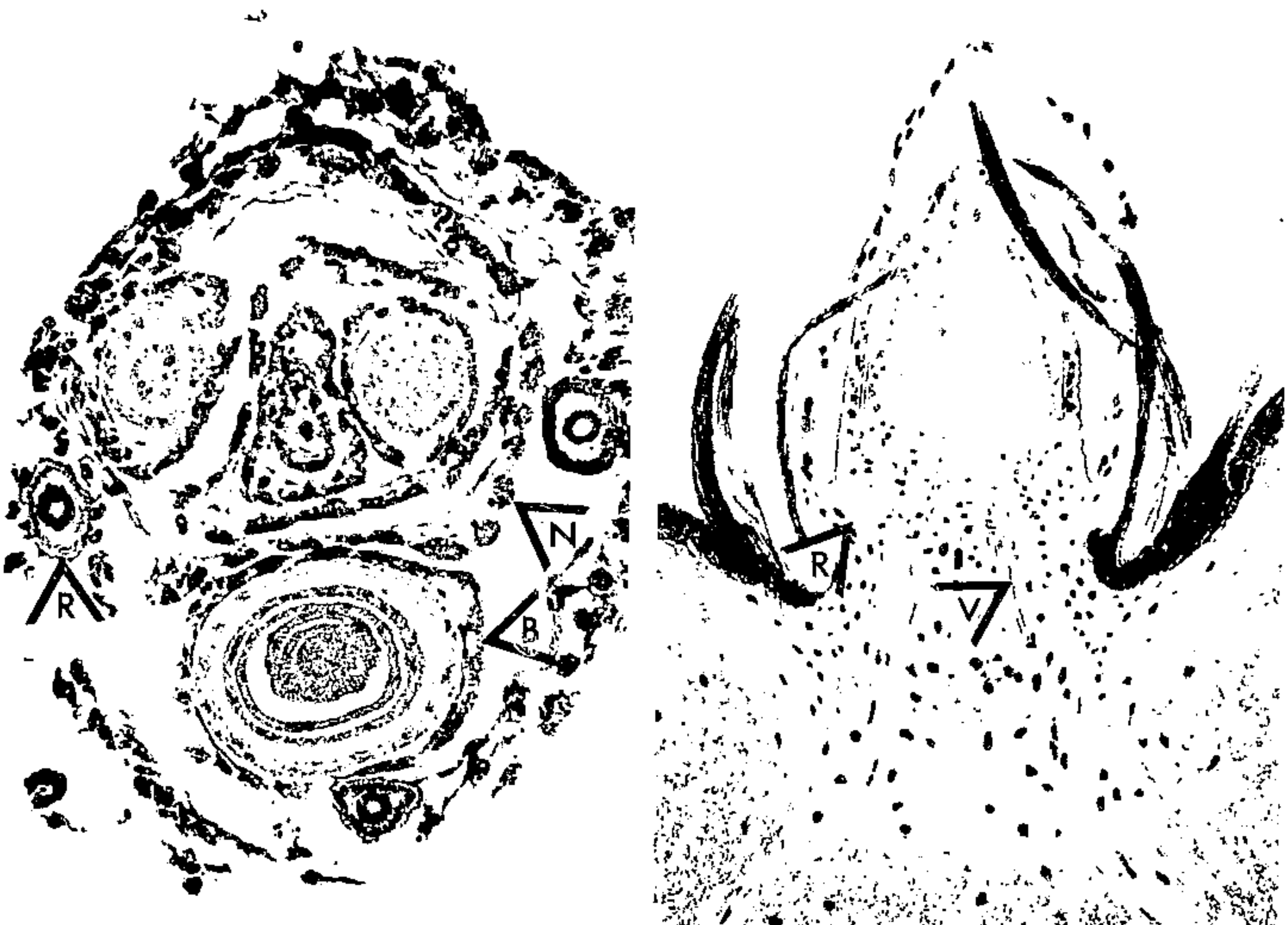

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FIGURE 10.—Cross section of tuber apex (left) and lontigudinal section of a mature bud (right) on the tuber. Nodal tissues (*N*), roots (*R*), vascular tissues (*V*), and the oldest and largest bud (*B*) are indicated (*7*).

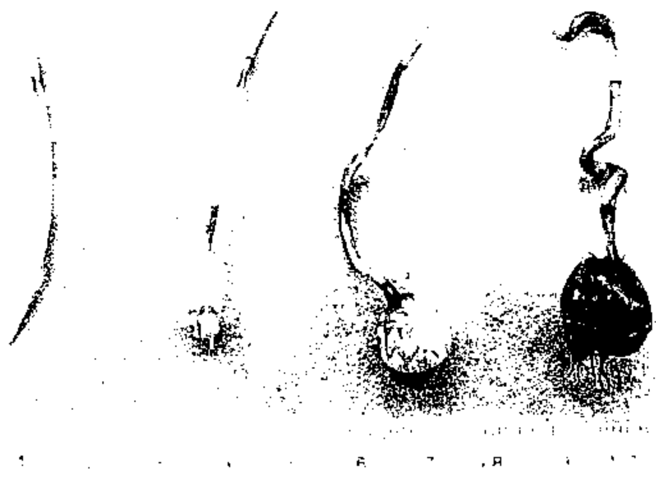

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FIGURE 11.—Successive tuber development (left to right) on a rhizome. Note nodes and scale leaves on both the rhizome and the tuber.

A newly differentiated tuber is white and fleshy, changing to tan and brown as it matures (fig. 11). During maturation, the epidermal layer becomes hard and tough as the epidermis lignifies. Mature tubers are dark brown to brownish black and tend to be spherical. Several morphological types, however, based primarily on tuber color, have been described (*61, 62*). Distorted shapes are common, resulting from physical constraints during development. Small, thick-veined leaves are compressed against the tuber surface (*7*). Adventitious roots also protrude randomly from the tuber.

Variability

Since yellow nutsedge has been distributed widely throughout the world, appreciable variability within the species is expected. Certain taxonomic characteristics, namely the size of spikelet components, form the bases for naming the varieties *angustispicatus, macrostachyus, esculentus,* and *leptostachyus* (*14, 46*). The nonweed variety *sativus,* commonly called chufa (*4, 14*), is grown for its edible tubers. Additional varieties undoubtedly exist, but no systematic classification of them exists (*28*). The varieties *esculentus* and *leptostachyus* also differ in certain morphological characteristics, such as leaf size and tuber weight (*10, 46*). They also differ in response to herbicides (*10*).

Other populations, which vary in morphological characteristics, have been called variants, selections, or ecotypes (*9, 44, 75, 76*); all these populations have not been characterized taxonomically, but foundations for distinguishing varieties exist. For six different variants originating from diverse areas within the United States, large differences were shown in shoot density, tuber depths, flowering, tuber size, tuber survival over winter, and other morphological features (*44*). Furthermore, the tubers differed in their contents of total carbohydrate, saccharides, starch, lipid, and protein, as well as in their component fatty acids (*45*). Six additional biotypes also differed in their morphological and growth characteristics (*75, 76*).

Life Cycle

Yellow nutsedge is a perennial but grows much like an annual in that the vegetative propagules, the tubers, serve nearly the same function as the seeds of annuals. Viable seeds (achenes) often are produced (*5, 27, 34, 66*), but they do not play a role in propagating the species in cultivated fields because the small and nonvigorous seedlings do not survive (*59*). The difficulty of seedling survival from seeds in purple nutsedge, even in undisturbed sites, was noted before the 20th century (*43*) and has recently been expressed for yellow nutsedge (*66*). Thus, attention should be focused on the tuber as the principal propagule in this species. Although figure 12 is a simplified representation of the life cycle of this weed, the details of seasonal growth and development are given below.

Tuber germination and seedling emergence. The tubers lie dormant in the soil at various depths during the winter. In most soils, more than 75 percent of the tubers are produced in the "plow layer" or upper 6 in (15 cm), but their distribution is affected by tillage and soil type (*5, 67*). The cold and wet conditions of overwintering serve to break tuber dormancy (*5, 54, 67*). Germination inhibitors are present in tubers (*30, 60, 68*), and water movement through the soil during the dormant period aids in leaching inhibitors out of the tubers. Those tubers near the surface can be killed by extreme cold or desiccation, or both (*54, 63*); this effect of cold and drought probably influences only those tubers in the top 2 to 3 in (5 to 8 cm) of soil in Corn Belt climates.

In the spring when the soil warms, some tubers are stimulated to germinate. Temperatures about 54° F are required to initiate tuber germination (*54*). In central Illinois, seedling emergence begins near May 1 and continues for as long as 60 days (fig. 13). Seedlings readily emerge from tubers 12 in (30 cm) deep, and some emerge through 18 in (46 cm) of soil (*59, 67*), but emergence is delayed as the tuber depth in the soil increases.

When a tuber germinates, one or more slender rhizomes elongate vertically from the buds at the terminal end of the tuber (*53*). The rhizome is a continuation of the growth of the rhizome that formed the tuber, with the new rhizome expressing a negatively geotropic (upward) response upon germination (fig. 14). Because the end of the leaf at the tip of the rhizome is sharp and strong, seedlings can emerge through hard and tough substrates. Because of the presence of numerous buds, a tuber can germinate several times, as well as produce several seedlings at one time (*7, 53, 65*). The first germination consumes most of the food reserves, leaving the subsequent seedlings with reduced vigor (*53*). Apical dominance in tuber buds is expressed inasmuch as the

LIFE CYCLE OF YELLOW NUTSEDGE

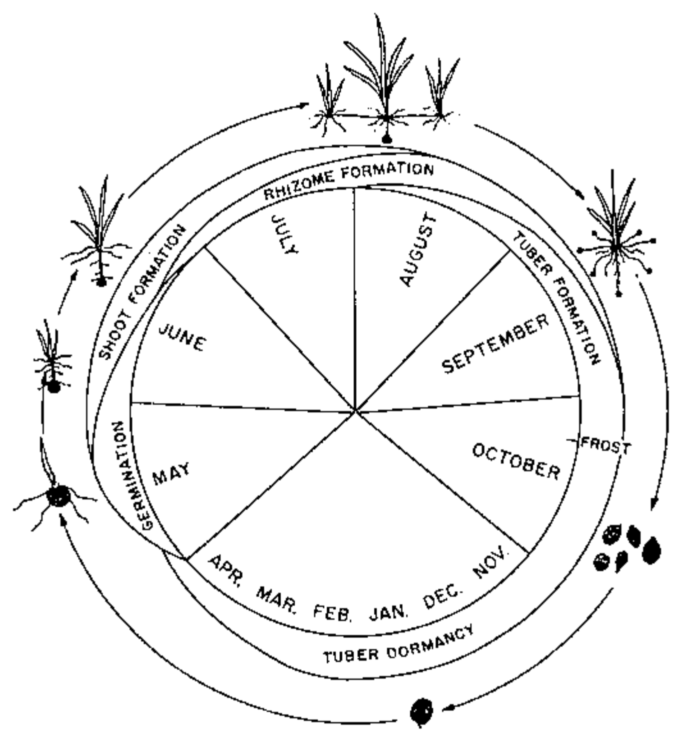

FIGURE 12.—A diagram of yellow nutsedge life cycle in the Corn Belt. The width of the band enclosing the five major processes indicates the intensity of the process. Tuber dormancy is depicted only during the nongrowing season but occurs throughout the year.

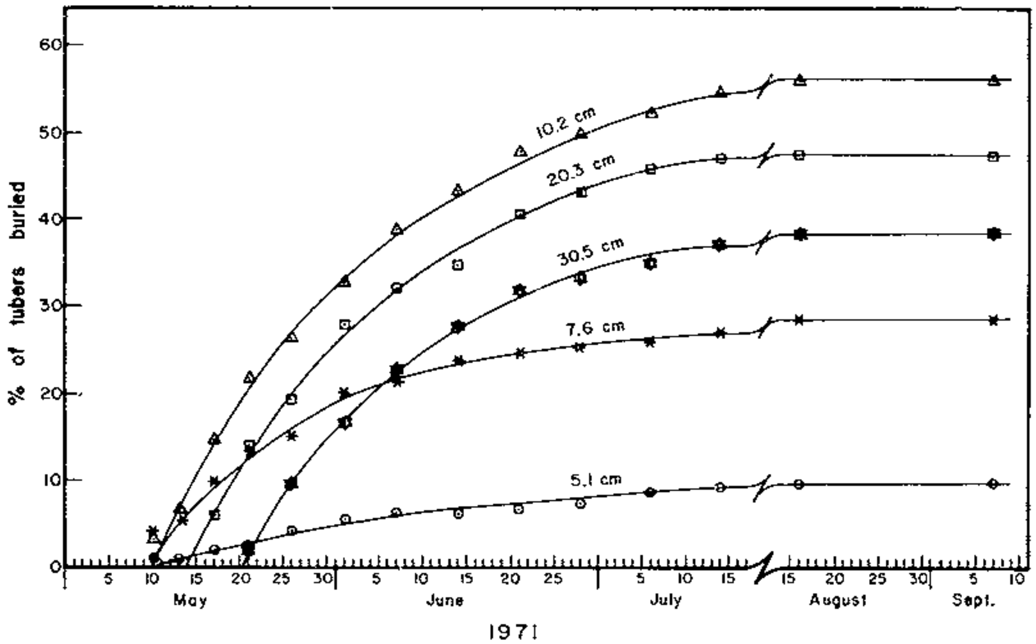

FIGURE 13.—Cumulative emergence of yellow nutsedge seedlings from tubers buried at various depths the previous fall. Emergence is low from tubers at 2 in (5.1 cm) and 3 in (7.6 cm) because the tubers winterkilled.

attached seedling inhibits germination in the other buds. Dominance is released for additional buds to germinate when the seedlings are detached from the tuber. The plant evolved from the seedling remains attached to the tuber by the vertical rhizome, provided it is not physically detached.

As the rhizome reaches the soil surface, the rhizome tip encounters sunlight and diurnal temperature fluctuations, which are the principal factors in stimulating the basal bulb to form under the soil. In a uniform seedbed, all basal bulbs are at a comparable distance from the soil surface, regardless of depth of the tuber. As a basal bulb forms, the nodes in the top of the vertical rhizome become compacted leaving very short internodes. Leaf length and function are dependent on internode length: short, scalelike leaves develop at nodes attached to long internodes, and long, photosynthetically active leaves develop at nodes attached to short internodes at the basal bulb.

The basal bulb region contains meristems for roots, secondary rhizomes, leaves, and the flower stalk. Its size increases as these structures develop. Since nodes and internodes of the stem are compacted into the basal bulb, all leaves of a mature plant extend to nodes below the soil surface. Leaves grow out of the bulb from a plicate, triangular fascicle, beginning with the outermost leaf and the fascicle terminates, under proper conditions, in a seed-bearing rachis. One leaf grows at each node. In early shoot development, each successive photosynthetically active leaf tends to be longer than the previous leaf (*31*).

Vegetative development. Several 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 bulb. These secondary bulbs produce shoots, then rhizomes, as described above for primary bulbs. Tertiary bulbs and bulbs of a higher order are possible, forming a complex system of subterranean, vegetative growth. Under ideal conditions, a single tuber can proliferate into a dense stand of shoots covering several square meters in a single season. Vegetative growth is rapid from the time of rhizome initiation until tubers start to form (fig. 12).

Photoperiod is considered the major known factor controlling the differentiation of rhizomes (*18, 31*), but temperature fluctuations, chemicals, and nutrition also affect the differentiation (*6, 18*). Long days promote differentiation into basal bulbs; short days enhance differentiation into tubers (*31*). Day length is maximal in early summer, apparently promoting maximum vegetative growth for the first several months of

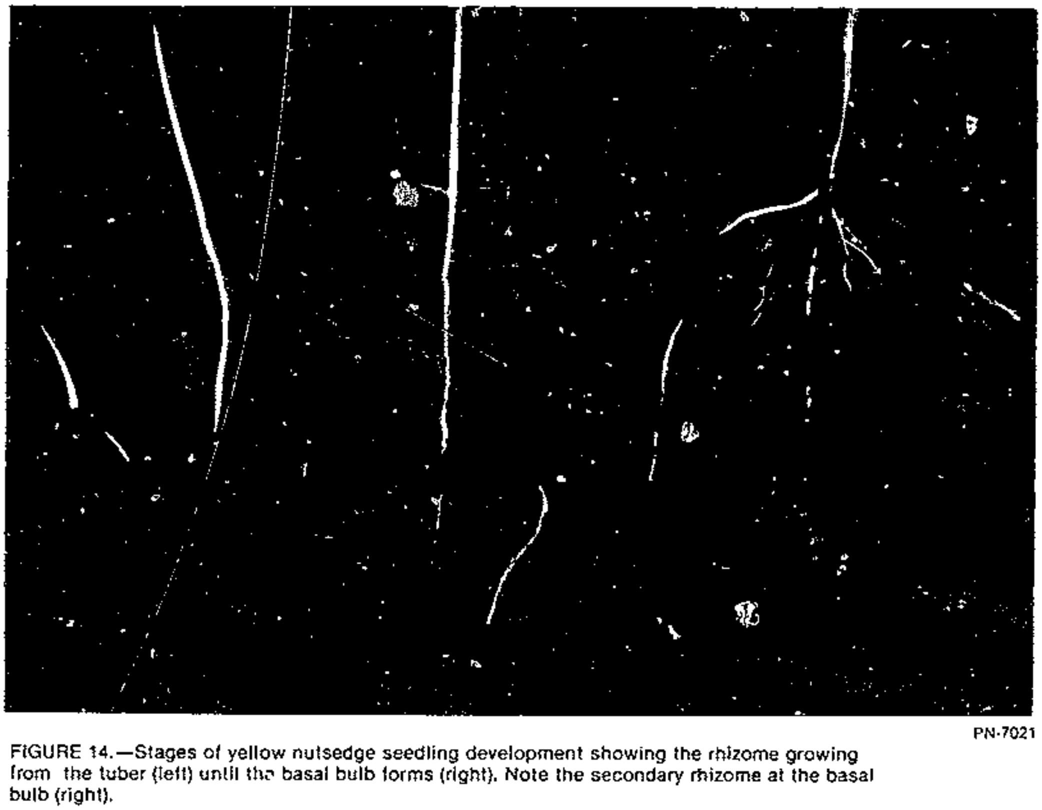

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FIGURE 14.—Stages of yellow nutsedge seedling development showing the rhizome growing from the tuber (left) until the basal bulb forms (right). Note the secondary rhizome at the basal bulb (right).

growth. An extensive vegetative system then is operative to form the tubers late in the season.

Tuber development. As day length shortens late in the season, rhizomes differentiate into tubers instead of basal bulbs (*18, 31*). In the Corn Belt, tubers are produced from August until the season ends (*33*, fig. 12). Using its complex system of shoots and rhizomes, the plant is efficient at fixing dry matter in tubers. At the end of the season, more than one-third of the total plant dry weight is commonly in tubers (*33*).

A single plant, growing alone, could produce 9,000 tubers in a season (*67*); however, fewer tubers are produced when crops grow with the yellow nutsedge than when nutsedge grows without competition. For in corn, tuber density in the soil increases rapidly in one season when control is inadequate, then levels off at about 45/ft^2 (1,000 tubers/m^2) for at least 3 years (*56*). No experimental evidence is available to explain why this tuber density does not increase in crops; however, allelopathy may be a factor.

Flowering. From the standpoint of propagation, flowering is a rather insignificant event in the life cycle of this plant, mostly because of the apparent inability of seeds to establish plants in cultivated fields. Since flowering does not occur regularly in all populations every season, plants do not produce seeds every year. A plant that survives until fall, however, almost always will produce tubers before it senesces.

Photoperiod has been considered the major factor that controls flowering in yellow nutsedge (*31*). In greenhouse studies, flowering occurred at photoperiods from 12 to 14 h (*31*). 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.

One of the first superficial evidences of floral development is the appearance of the foliar tube elongating from the fascicle center (*31*). The foliar tube is a hollow tube formed by the two most recently differentiated leaves growing as a single unit (*31*). The flowering structures differentiate from the apical meristem of the basal bulb (*74*) and elongate inside the foliar tube. The well-developed inflorescence bursts from the foliar tube as it protrudes the fascicle (*31*). The inflorescence continues to develop, sometimes forming mature, viable achenes.

Nutsedge Uses

The beneficial uses of yellow nutsedge seem to be confined to the nonweedy variant called chufa. This plant is the same species as yellow nutsedge (variety *sativus*, *4, 14*) but lacks the characteristics that would allow it to compete aggressively as a pest in most cropping environments (*48*). Chufa has been cultivated since earliest times in Italy and Northern Africa (*42, 50*) and still is grown in different parts of the world for its edible tubers.

Chufa tubers are used as feed unaltered as forage crop for animals (*42*) and as an ingredient for "horchata" (a beverage for humans). They also are extracted for vegetable oil, ground and roasted for coffee substitute (although they contain no caffeine) (*50*), and roasted as "earth almonds" for human food. Because of their low protein (*16*) and high fiber content, chufa tubers have relatively low nutritional value (*47*). The major differences between chufa and yellow nutsedge tubers are that chufa tubers are larger and have a higher oil content (*42, 50*). They have from 20 to 29 percent fatty oil and 30 to 50 percent carbohydrates (*16, 50*).

Tuber Characteristics

Size. Tubers tend to be spherical (fig. 9), although other distorted shapes occur because of obstructions during development. Mature tubers vary greatly in size within a local population, as well as among various populations (*44, 45, 65, 67*). For example, tubers gathered from Illinois ranged from 0.1 to 0.4 in (3 to 11 mm) in diameter with an average of 0.3 in (7 mm), while those from Minnesota ranged from 0.08 to 0.3 in (2 to 7 mm) with a one-fifth in (5 mm) average diameter (*44, 67*). The fresh weight of the Illinois tubers averaged 0.005 oz (150 mg) while the Minnesota tubers averaged 0.002 oz (70 mg). Tubers from Maryland were one-half in (12 mm) in diameter and weighed 0.03 oz (710 mg) (*44*).

Distribution in the soil. In most undisturbed soils, more than 75 percent of the tubers are produced in the surface 6-in (15 cm) layer with some tubers being produced as deep as 18 in (46 cm) in dense populations of yellow nutsedge. In Minnesota, for example, the percentages of tubers in successive 6-in (15 cm) layers of a peat soil were 85, 14 and 1, with none below 18 in (46 cm) (*67*). Similarly, an average 80, 20, and 0 percent of the tubers were found in these respective increments in two soils in Delaware (*5*).

Tuber density in the soil is affected somewhat by the yellow nutsedge density. In dense, pure stands, 500 to 1,500 tubers/ft^2 (5,000 to 16,000/m^2) commonly yield 9 to 15 tons/acre (*67*). In cultivated fields where this weed competes with a crop, tuber densities on the order of 100/ft^2 (1,100/m^2) have been observed (*57*), which is much less than those in pure stands of yellow nutsedge.

Temperature and moisture. Tubers can be killed relatively easy by extreme cold or heat as well as by desiccation (*11, 52, 53, 57, 63*). Temperatures of −7°C killed 50 percent of the tubers in the laboratory, while some tubers survived −20°C in the field (*52, 54*). Low temperature apparently causes tubers near the soil surface to be killed during the winter (*54*), but the cold does not greatly affect those tubers deep in the soil because the soil surface insulates them from exposure to the severe cold. A combination of low temperature and low humidity is more effective together than either one alone in causing tubers to lose their viability (*63*). Tubers can withstand 63°C in water without loss in viability (*71*). Variants differ in their responses to both temperature and desiccation, and some survive desiccation (*12, 44, 64*).

Longevity. Although tubers of yellow nutsedge resemble seeds of other plants in many functional characteristics, tubers are not as long-lived as many weed seeds. Over 80 percent of the tubers in the soil will decay in less than 3 years (*54, 64, 67*). Tuber longevity, of course, is dependent on tuber depth (*54*). Half-lives are expected of about 4 months for tubers 4 in (10 cm) below the surface and 6 months for tubers 8 in (20 cm) below the surface (fig. 15) (*54*). In corn moderately infested with yellow nutsedge, at least 2 years of season-long control treatments were necessary to reduce tubers to 20 percent of the original density and 3 years of the treatments to reduce them to 15 percent (*56*). The time required for 100 percent of the tubers to die is not known but is of interest if eradication of this species is desired.

Composition. Considerable diversity exists among the

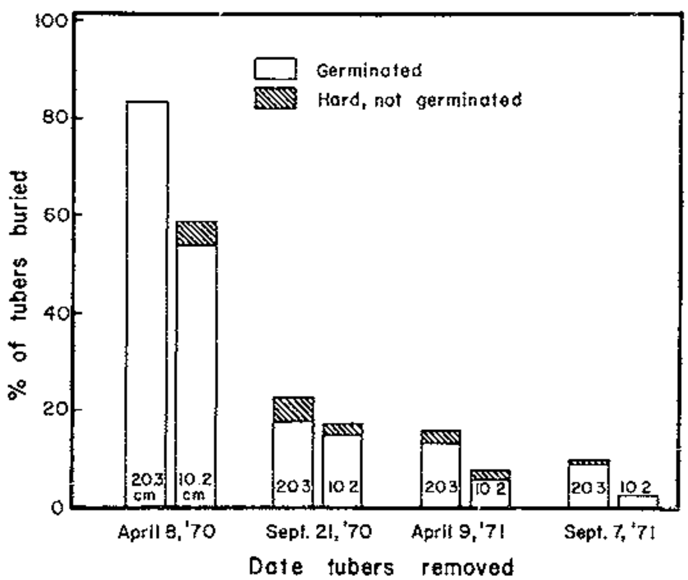

FIGURE 15.—Change in tuber viability at two depths after burial at Urbana, Ill., November 1969.

weedy yellow nutsedge variants regarding their tuber composition (*45*). The major differences seem to be that they contain varying proportions of constituents, rather than different constituents. Carbohydrates are the most abundant component in tubers, constituting from 45 to 75 percent of tuber dry weight (*45*). While starch is the major carbohydrate present, also present are small quantities of fructose, glucose, sucrose, melibiose, and other polysaccharides (*45*).

Lipids constitute from 5 to 14 percent of tuber dry weight (*45, 57*). The storage lipids, triglycerides, are present in the largest quantity, while polar lipids make up only about 1 percent of tuber dry weight. Oleic and linoleic are the principal fatty acids in tubers; other acids present are palmitic, linolenic, stearic.

Protein content varies from 5 to 10 percent of tuber weight (*45*). The constituent amino acids of these proteins have not been characterized.

Tubers contain a variety of phenolic compounds. Ferulic and *p*-coumaric acids are the most abundant, but vanillic, *p*-hydroxybenzoic, syringic, protocatecheric, and caffeic acids, as well as eugenol and others, are also present (*30, 60*). Most of these compounds also are present in the foliage of the plant (*30*) and may contribute to the apparent allelopathic action of yellow nutsedge tissues on other plants (*13*), since these compounds are known inhibitors of plant growth. In addition, these phenolics are probably responsible for the observed germination inhibition of seeds of other species by extracts from yellow nutsedge tubers (*60, 67*). These compounds also may inhibit the tubers themselves from germinating, causing the observed dormancy.

Dormancy and germination. Tuber dormancy is considered one of the major obstacles in controlling this weed because a population of tubers is always dormant and therefore resists control methods. If all the tubers in the soil could be stimulated to germinate at one time, the subsequent plants could be killed and yellow nutsedge could be eliminated.

Tubers are most dormant near the end of the growing season and least dormant in the spring and early summer (*5, 61*), which contributes to the observed emergence pattern in the field (*68*). Many tubers that lie dormant in the field can be stimulated to germinate by washing the soil from them and placing them in a suitable environment (*53*).

A number of chemicals or physical actions can break tuber dormancy and cause them to germinate. Such chemicals as ethylene chlorohydrin, thiourea, ethyl ether, ethephon, KSCN, H_2O_2, O_2, gibberellic acid, and benzyladenine have been reported to break dormancy when applied at the appropriate concentration (*5, 62, 68*). Such physical actions as cool, 2° to 5°C storage or stratification (*5, 68*), scarification (*62*), desiccation (*64*), and leaching with H_2O (*68*) are also effective in breaking tuber dormancy.

Tillage operations can stimulate tubers to germinate in the field (*11, 12*). Cool winter temperatures and leaching with H_2O are natural actions that promote spring germination. Leaching apparently washes away inhibitors that prevent germination. Diurnal alternating temperatures, which occur in the soil in the spring, also promote germination. Promoting tuber germination in the field as a prelude to control has been proposed but getting the germination-promoting substances into the soil to contact tubers and influence germination would be difficult. Applying a herbicide to kill the plants as they grow through the herbicide-treated soil is much more practical.

Most soil-applied herbicides that now are used widely for selective yellow nutsedge control in crops kill by acting on the newly formed seedling before emergence, but they do not kill the tuber or inhibit initial germination (*3, 5, 19, 38, 55, 73*). These herbicides seem to make the tubers dormant. When tubers are removed from treated soil, they are attached to the seedling that was killed by herbicide action, but the tubers readily germinate again when placed in fresh media (*3, 19, 73*). Tubers germinate when exposed to herbicides in nutrient culture (*3*), which supports evidence that these herbicides do not kill the tubers. On the other hand, some of the nonselective, soil-applied herbicides can kill the tubers (*21, 37*).

Foliar-applied herbicides that are applied during tuber formation translocate in the symplast and can kill the tubers that are being produced (*8, 36*); the foliar herbicides, however, that transport in the apoplast do not translocate to the tubers and kill them (*51*). Bentazon, applied several weeks after emergence, apparently can kill parent tubers without being translocated into the tuber (*55*).

Competitive Characteristics

Yellow nutsedge generally lacks the competitive traits that would cause it to reduce yields of corn or soybeans drastically in the Corn Belt. Growers who attain control will experience minimal yield losses, even in dense stands. This weed does not compete vigorously in these crops because the crops grow much taller than yellow nutsedge, resulting in a shade canopy for weed growth. Because yellow nutsedge growth is reduced greatly under low light, this weed does not compete well with crops that grow taller than the weed and provide a shaded canopy (*11, 39*).

Even with its lack of competitiveness, yellow nutsedge infestations have spread rapidly in the last several decades in the Corn Belt. It now is considered the most widespread and troublesome perennial weed in the region (*1, 72*). A number of factors, all acting together, probably have contributed to its spread during the last 20 years (*1*). The increased use of effective herbicides that control annual weeds, principally foxtails, but not yellow nutsedge, has reduced the population of plants that compete with and reduce the spread of yellow nutsedge (*72*). As the use of these herbicides increased and the control of annual weeds improved, mechanical cultivation in corn and soybeans was performed less frequently. Since cultivation is an effective weed control measure, reduced cultivation allowed more yellow nutsedge to grow.

Associated with reduced crop cultivation was the increased use of large-horsepower tractors, which pulled such implements as discs and field cultivators through fields at greater depths than the smaller tractors, thereby spreading tubers to the noninfested areas. While herbicides that effectively kill yellow nutsedge have been developed, their performance has been somewhat erratic because of environmental and soil conditions. These herbicides lack the residual activity to control the late-emerging yellow nutsedge plants, which then produce tubers late in the growing season (*33*).

Season-long competition from yellow nutsedge will result in maximum crop yield losses, compared with competition for part of the season, if the research found from another crop is expanded to corn and soybeans. In cotton, for example, no yield losses were found during 4 weeks of competition, but 6 weeks or longer of competition reduced yield more than 15 percent (*38*).

Research is lacking on the economic losses caused by yellow nutsedge infestations in corn and soybeans in the Corn Belt. Reports vary considerably on the losses caused by this weed, probably as a result of differences in weed density and environmental conditions. Greatest losses in yield occur under conditions where yellow nutsedge competes with crops for moisture (*56*). 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 droughty conditions.

In soybeans, yields were reduced 30 percent in Minnesota where no control measures were used in a dense population of the weed (*73*), but no reduction occurred in Illinois where yellow nutsedge was not controlled (*73*). For 4 years, no significant loss in soybean yields occurred in dense yellow nutsedge infestations where no control measures were used, and a good soybean stand was obtained. Generally, soybeans will compete with this weed better than corn because soybeans make the most dense canopy for shading the yellow nutsedge.

Reports of yield losses in corn also show variability. Where the yellow nutsedge was not controlled, corn yields were reduced 66 percent in Georgia (*24*) and from 0 to 47 percent in Illinois (*56*). While the losses averaged 10 percent in the Illinois experiments, losses ranged to 47 percent, and the research showed that there would be an expected 8 percent reduction in yield for every 10 yellow nutsedge plants per square foot (100/m²) (fig. 16). The extreme variability in yield losses at low tuber densities demonstrates the environmental variation in the effect of yellow nutsedge and stresses the economic need for controlling the weed because there can be significant yield losses, especially under droughty situations.

Tests have shown that dried yellow nutsedge tissues, especially tubers, are allelopathic to other plants in greenhouse experiments. Soybean growth was inhibited more than corn by the presence of 1 percent (by weight) of ground tubers in the rooting medium (*13*). The phenolic components probably are responsible for the allelopathic action of the tuber tissues since these compounds are known to inhibit growth and to be present in tubers (*30, 60, 68*). The significance of this allelopathic action under field conditions, however, is not yet known.

The response of yellow nutsedge to reduced irradiance has been studied to simulate the plants' response to shade under crop canopies. The season-long growth of a California variant was a linear function of sunlight intensity (*39*), but for an Illinois variant, dry matter production was the same at both 70 percent of full sunlight and at full sunlight (*33*). Although growth of yellow nutsedge is reduced greatly at 20 to 30 percent of full sunlight (*33, 39*), tuber production occurs at this low level of irradiance (*33*).

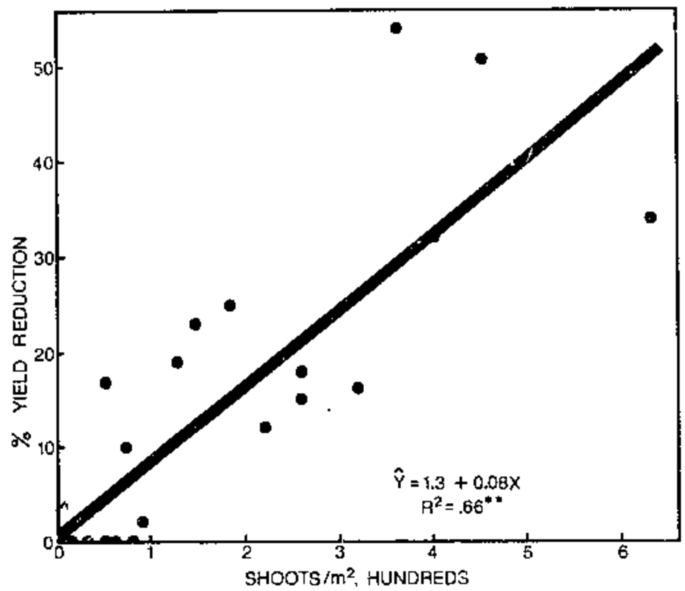

FIGURE 16.—The relationship between corn yield loss and yellow nutsedge shoot density (shoots evaluated in July) at Urbana, Ill. (3 years of data).

Control

No single measure is available that will control yellow nutsedge for the entire season in corn or soybeans. As with most weeds, an integrated program involving several methods is the most effective. Various strategies are available that can normally prevent yield losses, even though season-long control is not attained. Yellow nutsedge can be controlled selectively in corn and soybeans better than in most other cultivated crops.

Mechanical and cultural control. Tillage is an important part in a yellow nutsedge control program, but tillage alone will not provide satisfactory control. Preplant tillage can stimulate tuber germination (*73*), move tubers to the surface for kill by desiccation, and kill seedlings already germinated (*73*). Maximum benefit is obtained if tillage is delayed until substantial tuber germination occurs; however, control with preplant tillage is diminishing because of the increasing trend for growers to plant early in the season, before the tubers germinate.

Tillage after planting (cultivation) is an effective control measure, especially when combined with preplant herbicides. Cultivation effectively removes the weeds between the rows, and the crops can compete effectively with the yellow nutsedge left in the row. Two cultivations were as effective in yellow nutsedge control in corn as postemergence herbicides (*56*). Narrow-row culture probably should not be used in heavy yellow nutsedge infestations unless long-lasting control from soil treatments is assured.

Crop rotation is usually an excellent practice to follow in reducing troublesome weeds in cultivated crops. This is also true for yellow nutsedge, as the cropping system affects the long-term tuber population (*41*). Rotating to corn or soybeans from other crops is advisable, however, since chemical control and competition from these crops will reduce the infestation. Season-long control of yellow nutsedge for at least 2 years will reduce tuber numbers to at least 20 percent of the original density (*56*).

Soil-applied herbicides. The most effective and consistent control of yellow nutsedge in corn or soybeans is achieved by applying preplant-incorporated herbicides. An effective herbicide, applied to the soil near planting time, in conjunction with crop cultivation, seems to afford acceptable yellow nutsedge control without yield losses in corn (*56*) or soybeans. When applied preemergence at planting time, herbicides require adequate rainfall within several days to achieve control as good as that with equivalent treatments applied by preplant incorporation. To be effective, the herbicides have to be placed where they can readily be absorbed by the emerging shoot and roots (*2, 29, 72, 73*). Since roots are present on the stem from the tuber to the basal bulb on young emerging plants (fig. 14), herbicides need to be incorporated into the soil to facilitate uptake.

Alachlor and metolachlor are acetanilide herbicides that selectively control yellow nutsedge in both corn and soybeans (*2, 35, 72, 73*). These herbicides perform best on the silt loam and silty clay loam soils, which are typical of many soils in the Corn Belt, with medium to high organic matter (up to about 6 pct). On coarse-textured soils, however, performance is often erratic. Metolachlor lasts longer than alachlor but also has somewhat more potential for injuring corn than alachlor.

EPTC and butylate are chemically similar herbicides for use in corn (*5, 22, 72*). Both herbicides commonly are formulated with a safening agent to reduce the possibility of crop injury. At equivalent rates, EPTC has more activity on yellow nutsedge than butylate but also has more potential for crop injury. These herbicides do not control yellow nutsedge as long as some of the other herbicides.

The herbicide, vernolate, effectively controls yellow nutsedge in soybeans. This herbicide is similar in chemical properties to EPTC and butylate. Vernolate sometimes injures soybeans but seldom reduces yields. Subsurface placement techniques of vernolate were developed and shown to be effective for yellow nutsedge control in peanuts, especially on sandy soils in the Southeastern United States (*23, 25*). These techniques have not yet been shown satisfactory in the Corn Belt, however.

Atrazine, used for many years for weed control in corn, provides only fair and often erratic yellow nutsedge control (*22, 72*). This herbicide is used most often with other herbicides, principally alachlor, EPTC, or butylate, so it may contribute to the control when used in these combinations.

A herbicide currently under development, S-734, gives good yellow nutsedge control in soybeans. This herbicide sometimes acts slowly but gives good late-season control. Rates need to be adjusted for different soil types since the activity seems to be influenced by soil texture and organic matter.

Postemergence herbicides. Foliar-applied herbicides are usually not as effective or consistent as soil-applied herbicides in a yellow nutsedge control program (*22, 72*). Postemergence treatments are most effectively used when applied as spot treatments or when soil-applied herbicides fail to give adequate control. Cultivation is often an acceptable alternative to postemergence herbicides.

Bentazon selectively controls yellow nutsedge in both corn and soybeans (*55*). It gives maximum control when applied with nonphytotoxic oil in split applications 10 to 14 days apart. In addition, complete coverage of the foliage is necessary. Yellow nutsedge sometimes recovers from bentazon sprays, resulting in poor control.

Atrazine, applied postemergence, can control yellow nutsedge in corn. It is most effective when applied at high rates with crop oil or surfactant. Ametryn can be used as a postemergence-directed spray in corn, provided sufficient height differential is allowed for the spray to cover the yellow nutsedge but not the corn.

Glyphosate kills yellow nutsedge very effectively (*55*). Since it lacks selectivity, however, it should be considered for use only on small, heavily infested areas as a spot treatment.

Biological control. Despite considerable effort to develop biocontrol agents for both yellow and purple nutsedges, these control methods currently are not successful enough for field use. Several insects, fungi, and nematodes attack yellow nutsedge (*48*), and insects currently offer the best chance for biocontrol of this species (*15*). Numerous insects from several genera specifically attack both nutsedges (*15*), but none are able to suppress early shoot growth satisfactorily to prevent crop losses and tuber production. The larvae and pupae of *Bactra verutana* (Zeller), which feed within the fascicle, represent the best effort yet to achieve biological control for their species (*15, 40*). Augmentation of larvae early in the season, however, may be required to achieve acceptable control (*15, 17*). Even if biological control is successful, it will have to compete economically with the mechanical, cultural, and chemical controls now widely used.

Literature Cited

(*1*) Armstrong, T. F. 1975. The problem: Yellow nutsedge. Proceedings of the North Central Weed Control Conference 30:120-121.

(*2*) _____ W. F. Meggitt, and D. Penner. 1973. Absorption, translocation, and metabolism of alachlor by yellow nutsedge. Weed Science 21:357-360.

(*3*) _____ W. F. Meggitt, and D. Penner. 1973. Yellow nutsedge control with alachlor. Weed Science 21:354-357.

(*4*) Bailey, L. H., and E. Z. Bailey. 1976. Hortus third: A Concise dictionary of plants cultivated in the United States and Canada. MacMillan Publishing Co., Inc., New York.

(*5*) Bell, R. S., W. H. Lachman, E. M. Rahn, and R. D. Sweet. 1962. Life history studies as related to weed control in the northeast. 1. Nutgrass. Rhode Island Agricultural Experiment Station Bulletin No. 364.

(*6*) Bendixen, L. E. 1970. Altering growth form to precondition yellow nutsedge for control. Weed Science 18:599-603.

(*7*) _____ 1973. Anatomy and sprouting of yellow nutsedge tubers. Weed Science 21:501-503.

(*8*) Bhan, V. M., E. W. Stoller, and F. W. Slife. 1970. Toxicity, absorption, translocation, and metabolism of 2,4-D in yellow nutsedge. Weed Science 18:733-737.

(*9*) Boldt, P. F., C. P. Yip, and R. D. Sweet. 1976. Phenotypic studies of five New York yellow nutsedge selections. Abstract of the Weed Science Society of America 155.

(*10*) Costa, J., and A. P. Appleby. 1976. Response of two yellow nutsedge varieties to three herbicides. Weed Science 24:54-58.

(*11*) Davis, C. H., and R. S. Hawkins. 1943. Eradication and control of nutgrass. Arizona Agricultural Experiment Station Bulletin No. 189.

(*12*) Day, B. E., and R. C. Russell. 1955. The effect of drying on survival of nutgrass tubers. California Agricultural Experiment Station Bulletin No. 751.

(*13*) Drost, K. C., and J. D. Doll. 1980. The allelopathic effect of yellow nutsedge (*Cyperus esculentus*) on corn (*Zea mays*) and soybeans (*Glycine max*). Weed Science 28:229-333.

(*14*) Fernald, M. L. 1950. Gray's manual of botany. Eighth ed. American Book Co., New York. 1,632 p.

(*15*) Frick, K. E. 1978. Purple nutsedge (*Cyperus rotundus* L.): A case for augmentation. p. 145-151. *In* T. E. Freeman, ed., Proceedings of the IV International Symposium on Biological Control of Weeds.

(*16*) Gad, A. M., and F. Osman. 1961. Studies on the chemical constitution of Egyptian chufa (*Cyperus esculentus*). United Arab Republic Journal of Chemistry 4(1):119-132.

(*17*) Garcia, C., Jr., and K. E. Frick. 1975. *Bactra verutana*, a possible biological control agent of purple and yellow nutsedge: Large scale rearing on artificial diet. Annals of the Entomological Society of America 68(1):15-18.

(*18*) Garg, D. K., L. E. Bendixen, and S. R. Anderson. 1967. Rhizome differentiation in yellow nutsedge. Weeds 15(2):124-128.

(*19*) Gentner, W. A. 1973. Yellow nutsedge control with MBR-8251. Weed Science 21:122-124.

(*20*) Gleason, H. A. 1963. Illustrated flora of the Northeastern United States and adjacent Canada. Vol. 1. Hofner Publishing Co., Inc., New York.

(*21*) Hardcastle, W. S., and R. E. Wilkinson. 1968. Response of purple and yellow nutsedge to dichlobenil. Weeds 16(3):339-340.

(*22*) Hauser, E. W. 1968. Yellow nutsedge—Problems, research trends and outlook. Proceedings of the Northeastern Weed Control Conference, p. 37-48.

(*23*) _____ J. L. Butler, J. L. Shepherd, and S. A. Parham. 1966. Response of yellow nutsedge, Florida pusley, and peanuts to thiocarbamate herbicides as affected by method of placement in soil. Weed Research 6:338-345.

(*24*) _____ C. C. Dowler, M. D. Jellum, and S. R. Cecil. 1974. Effects of herbicide-crop rotation on nutsedge, annual weeds, and crops. Weed Science 22:172-176.

(*25*) _____ L. E. Samples, and S. A. Parham. 1969. Incorporated versus subsurface vernolate for weed control in peanuts. Weed Research 9:173-184.

(*26*) Hicks, G. C. 1929. Cytological studies in *Cyperus, Eleocharis, Dulichium,* and *Eriophorum*. Botanical Gazette 88:132-150.

(*27*) Hill, E. R., W. H. Lachman, and D. N. Maynard. 1963. Reproductive potential of yellow nutsedge by seed. Weeds 11(2):160-161.

(*28*) Holm, L. G., D. L. Plucknett, J. V. Pancho, and J. P. Herberger. 1977. The world's worst weeds. Distribution and biology. University Press of Hawaii, Honolulu. p. 125-133.

(*29*) Ingle, E. L., and A. D. Worsham. 1971. Effect of soil placement of several herbicides on yellow nutsedge (*Cyperus esculentus*) tubers. Proceedings of the Southern Weed Science Society 24:188-192.

(*30*) Jangaard, N. O., M. M. Sckerl, and R. H. Schieferstein. 1971. The role of phenolics and abscisic acid in nutsedge tuber dormancy. Weed Science 19:17-20.

(*31*) Jansen, L. L. 1971. Morphology and photoperiodic responses of yellow nutsedge. Weed Science 19(3):210-219.

(*32*) Jenkins, L., and E. R. Jackman. 1941. Yellow nutgrass and sedge. Oregon Cooperative Extension Service Bulletin No. 568.

(*33*) Jordan-Molero, J. E., and E. W. Stoller. 1978. Seasonal development of yellow and purple nutsedges (*Cyperus esculentus* and *C. rotundus*) in Illinois. Weed Science 26:614-618.

(*34*) Justice, O. L., and M. D. Whitehead. 1946. Seed production, viability, and dormancy in the nutgrasses *Cyperus rotundus* and *C. esculentus*. Journal of Agricultural Research 73:303-318.

(*35*) Keeley, P. E., C. H. Carter, and J. H. Miller. 1972. Evaluation of the relative phytotoxicity of herbicides to cotton and nutsedge. Weed Science 20:71-74.

(*36*) _____ and R. J. Thullen. 1970. Vitality of tubers of yellow nutsedge treated with arsenical herbicides. Weed Science 18:437-439.

(*37*) _____ and R. J. Thullen. 1974. Yellow nutsedge control with soil-incorporated herbicides. Weed Science 22:378-383.

(*38*) _____ and R. J. Thullen. 1975. Influence of yellow nutsedge competition on furrow-irrigated cotton. Weed Science 23:171-175.

(*39*) _____ and R. J. Thullen. 1978. Light requirements of yellow nutsedge (*Cyperus esculentus*) and light interception by crops. Weed Science 26:10-16.

(*40*) _____ R. J. Thullen, and J. H. Miller. 1970. Biological control studies on yellow nutsedge with *Bactra verutana* Zeller. Weed Science 18:393-395

(*41*) _____ R. J. Thullen, J. H. Miller, and C. H. Carter. 1979. Comparison of four cropping systems for yellow nutsedge (*Cyperus esculentus*) control. Weed Science 27:463-467.

(*42*) Killinger, G. B., and W. E. Stokes. 1946. Chufas in Florida. University of Florida Agricultural Experiment Station Bulletin No. 419.

(*43*) MacMahon, P. 1899. Nut-grass (*Cyperus rotundus*). Queensland Agricultural Journal 5:460-465.

(*44*) Matthiesen, R. L. 1976. Plant development and tuber composition of six biotypes of yellow nutsedge (*Cyperus esculentus* L.). Ph. D. thesis, University of Illinois, Urbana.

(*45*) Matthiesen, R. L., and E. W. Stoller. 1979. Tuber composition in yellow nutsedge (*Cyperus esculentus* L.) variants. Weed Research 18:373-377.

(*46*) Mohlenbrock, R. H. 1960. The *Cyperaceae* of Illinois. I. *Cyperus*. American Midland Naturalist 63:270-306.

(*47*) Mokady, S. H., and A. Dolev. 1970. Nutritional evaluation of tubers of *Cyperus esculentus* L. Journal of the Science of Food and Agriculture 21:211-214.

(*48*) Mulligan, G. A., and B. E. Junkins. 1976. The biology of Canadian weeds. 17. *Cyperus esculentus* L. Canadian Journal of Plant Science 56:339-350.

(*49*) Parker, C. 1972. Distinguishing *Cyperus rotundus* from some related species. Pesticide Articles and News Summaries 18:477-482.

(*50*) Power, F. B., and V. K. Chestnut. 1923. Chemical examination of "chufa," the tubers of *Cyperus esculentus* Linne. Journal of Agricultural Research 26:69-75.

(*51*) Saidak, W. J. 1961. Translocation of dalapon in yellow nutsedge (*Cyperus esculentus*). Weeds 9(4):626-633.

(*52*) Stoller, E. W. 1973. Effect of minimum soil temperature on differential distribution of *Cyperus rotundus* and *C. esculentus* in the United States. Weed Research 13:209-217.

(*53*) _____ D. P. Nema, and V. M. Bhan. 1972. Yellow nutsedge tuber germination and seedling development. Weed Science 20:93-97.

(*54*) _____ and L. M. Wax. 1973. Yellow nutsedge shoot emergence and tuber longevity. Weed Science 21:76-81.

(*55*) _____ L. M. Wax, and R. L. Matthiesen. 1975. Response of yellow nutsedge and soybeans to bentazon, glyphosate, and perfluidone. Weed Science 23:215-221.

(*56*) _____ L. M. Wax, and F. W. Slife. 1979. Yellow nutsedge (*Cyperus esculentus*) competition and control in corn (*Zea mays*). Weed Science 27:32-37.

(*57*) _____ and E. J. Weber. 1975. Differential cold tolerance, starch, sugar, protein, and lipid of yellow and purple nutsedge tubers. Plant Physiology 55:859-863.

(*58*) Sweet, R. D. 1974. Yellow nutsedge workshop. Proceedings of the Northeast Weed Science Society 28:20-34.

(*59*) _____ 1975. Control of nutsedge in horticultural crops. Proceedings of the North Central Weed Control Conference 20:129-130.

(*60*) Tamés, R. S., M. D. V. Gesto, and E. Vieitez. 1973. Growth substances isolated from tubers of *Cyperus esculentus* var. *aureus*. Physiology Plantarium 28:195-200.

(*61*) Taylorson, R. B. 1967. Seasonal variation in sprouting and available carbohydrate in yellow nutsedge tubers. Weeds 15(1):22-24.

(*62*) Thomas, P. E. L. 1967. A preliminary study on the dormancy of *Cyperus esculentus* tubers. Pesticide Articles and News Summaries (C) 13(4):329-333.

(*63*) _____ 1969. Effects of desiccation and temperature on survival of *Cyperus esculentus* tubers and *Cynodon dactylon* rhizomes. Weed Research 9:1-8.

(*64*) _____ and I. E. Henson. 1968. Influence of climate and soil moisture on tuber dormancy of *Cyperus esculentus*. Pesticide Articles and News Summaries (C) 14(3):271-276.

(*65*) Thullen, R. J., and P. E. Keeley. 1975. Yellow nutsedge sprouting and resprouting potential. Weed Science 23:333-337.

(*66*) _____ and P. E. Keeley. 1979. Seed production and germination in *Cyperus esculentus* and *C. rotundus*. Weed Science 27:502-505.

(*67*) Tumbleson, M. E., and T. Kommedahl. 1961. Reproductive potential of *Cyperus esculentus* by tubers. Weeds 9(4):646-653.

(*68*) _____ and T. Kommedahl. 1962. Factors affecting dormancy in tubers of *Cyperus esculentus*. Botanical Gazette 123(3):186-190.

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(*69*) U.S. Department of Agriculture. 1970. Selected weeds of the United States. U.S. Department of Agriculture, Agric ..lture Handbook No. 366. p. 98.

(*70*) ...... 1972. Extent and cost of weed control with herbicides and an evaluation of important weeds. 1968. U.S. Department of Agriculture, Agricultural Research Service. Headquarters, ARS-H-1. [Series discontinued; Agricultural Research Service is now Science and Education Administration-Agricultural Research.]

(*71*) Waters, W. E. 1965. Influence of hot-water on viability of *Cyperus esculentus* and *Cyperus rotundus* tubers. Proceedings of the Florida State Horticulture Society 78:437-440.

(*72*) Wax, L. M. 1975. Control of yellow nutsedge in field crops. Proceedings of the North Central Weed Control Conference 30:125-128.

(*73*) ...... E. W. Stoller, F. W. Slife, and R. N. Andersen. 1972. Yellow nutsedge control in soybeans. Weed Science 20:194-201.

(*74*) Wills, G. D., R. E. Hoagland, and R. N. Paul. 1980. Anatomy of yellow nutsedge (*Cyperus esculentus*). Weed Science 28:432-437.

(*75*) Yip, C. P., and R. D. Sweet. 1978. Biotypes of yellow nutsedge (*Cyperus esculentus* L.): I. Morphological characteristics. Abstract of the Weed Science Society of America 127.

(*76*) ...... and R. D. Sweet. 1978. Biotypes of yellow nutsedge (*Cyperus esculentus* L.): II. Tuber dormancy and responses to photoperiod and herbicides. Abstract of the Weed Science Society of America 128.

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