INVESTIGATIONS ON YELLOW NUTSEDGE

BY

RON KEHLER

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Abstract

Yellow nutsedge (*Cyperus esculentus* L.) is a perennial weed that was first identified in Manitoba in 1979. Red River floodwaters carried the tubers of yellow nutsedge northward and deposited them when the floodwaters receded. To date 21 infestations have been identified in Manitoba, all infestations bordering the Red River. In consideration of the economic importance of this weed, research into its growth and reproductive potential in Manitoba was initiated.

Growth analysis was carried out on two ecotypes of yellow nutsedge, one from Harrow, Ontario and the other from Manitoba. The Manitoba ecotype produced 3 400 tubers in a single growing season compared with 1 450 from the Ontario ecotype. On the contrary, the Manitoba plants only produced about one third the number of shoots that the Ontario ecotype produced. In addition to these morphological differences, physiological differences were also discovered. The critical freezing temperatures for the Manitoba and Ontario ecotypes were determined to be -15.0 and -7.3 C, respectively.

Cultivation is an important component of perennial weed control since it exposes underground plant tissue to desiccation conditions. Tubers of yellow nutsedge were evaluated for desiccation tolerance by allowing them to equilibrate with a wide range of different relative humidities. Yellow nutsedge tubers were determined to be tolerant to desiccation. The rate of desiccation and tuber viability were strongly related.

Yellow nutsedge is sensitive to shading; therefore it was grown together with barley to determine the effect of crop competition on its growth and reproductive

potential. Crop competition resulted in a 7-fold reduction in total nutsedge biomass accumulation. Similarily, there was a 18 - fold difference in tuber production between nutsedge grown with and without a barley crop resulting in a net increase of 168 and 3151 tubers m^{-2}, respectively, by the end of the growing season.

These results show that yellow nutsedge is a pernicious weed that will increase rapidly if left unchecked. Control of this weed will require implementation of several different control practices over an extended period.

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I. Introduction

Yellow nutsedge (*Cyperus esculentus* L.) is one of approximately 600 species in the genus *Cyperus* (Cyperaceae). Of these, purple nutsedge or *Cyperus rotundus*, is ranked as the world's worst weed and yellow nutsedge, the 16th worst weed (Holm et al., 1977). The former occurs throughout tropical and subtropical regions and is a major problem in rice, sugarcane, cotton, maize and vegetable crops, whereas yellow nutsedge occurs in both the tropical and more northerly latitudes and is a problem in sugarcane, maize, potato, cotton and soybean. In North America, yellow nutsedge occurs in Nova Scotia, New Brunswick, Quebec, Ontario, and Manitoba and throughout the United States (Bendixen and Nandihalli, 1987).

Yellow nutsedge is a perennial weed that proliferates primarily by tubers which form at the ends of rhizomes. The leaves follow a one-third phyllotaxy, have a definite midvein, are 10 to 80 cm long and up to 1 cm wide. The rachis protrudes from the fascicle and bears the inflorescence. The inflorescence is a yellowish to brown umbel that is subtended by 3 to 9 involucral leaves or bracts. Varieties of *C. esculentus* are distinguished by inflorescence characteristics since there is considerable variation in spikelet morphology (Yip, 1978).

Interference from yellow nutsedge results in significant losses in both crop yield and quality in many parts of the world. The most severe losses are incurred in crops such as cotton and horticultural row crops that offer little competition to the weed. For example cotton yields were reduced by up to 41% from yellow nutsedge competition in Alabama (Patterson et al., 1980). In Illinois, a density of

300 yellow nutsedge shoots per square meter reduced corn yields by 25% (Stoller, 1981).

Yellow nutsedge-growing in a potato crop can reduce both yield and quality of the crop. In New York up to 29% of the potato tuber harvest was damaged as a result of yellow nutsedge rhizomes growing through the tubers (Yip et al., 1974).

The prolific growth and perennial nature of yellow nutsedge make it difficult to control (Friesen, 1986). Few selective herbicides are available that provide effective control of this weed. In addition, nutsedge tubers that are dormant or sprout late may escape chemical control applied early in the growing season, but these tubers can be disturbed by tillage. Shallow cultivation provides the most effective control of the underground structures of yellow nutsedge since deep tillage places the tubers deeper in the soil profile where tuber longevity is enhanced (McDonald, 1986). Tubers on, or near, the soil surface may be killed by desiccation or freezing (Stoller and Wax, 1973; Lanini, 1987).

In Manitoba, yellow nutsedge was first reported in 1979 near Emerson (Sturko, 1983) and has since been identified at a number of additional sites adjacent to the Red River. Because of the recent introduction of yellow nutsedge into Manitoba and the potential for this weed to spread, this research was initiated with the following objectives:

1. Determine the geographical distribution of yellow nutsedge in Manitoba.

2. Characterize the growth, reproductive potential, critical tuber freezing temperature and tuber desiccation tolerance of two yellow nutsedge

ecotypes, one from Aubigny, Manitoba and the other from Harrow, Ontario, under Manitoba growing conditions.

3. Determine the influence of barley crop competition on the growth and reproductive potential of yellow nutsedge (Manitoba ecotype).

2. Literature Review

2.1. Yellow Nutsedge - Life Cycle

Yellow nutsedge reproduces both asexually by tubers and sexually by seed. A brief review of the general life cycle of the weed follows. For more comprehensive reviews, see Mulligan and Jenkins (1976), Stoller (1981), and Stoller and Sweet (1987).

Yellow nutsedge, is a perennial weed regenerating each spring from underground tubers that overwinter in the soil. Multiple rhizomes commonly emerge simultaneously from a single tuber on the first sprouting (Bendixen, 1973; Thullen and Keeley, 1975). Primary rhizomes are negatively geotropic and when exposed to light differentiate to form a basal bulb, usually just below the soil surface. The basal bulb contains meristems for the inflorescence, leaves, rhizomes and roots, (Stoller et al., 1972), and as such facilitates the conversion from heterotrophism to autotrophism in new shoots. As aerial growth proceeds, the size of the basal bulb increases and more indeterminate rhizomes are formed (Mulligan and Junkins, 1976). Differentiation of these indeterminate rhizomes into tubers or more basal bulbs is affected by photoperiod, with long days promoting shoot growth, and short days tuber production (Garg et al., 1967; Jansen, 1971). Tuber production continues until freezing temperatures kill the above-ground plant parts. Normally flowering is induced by a short photoperiod (Williams, 1982) but can also occur under long photoperiods (Jordan-Molero and Stoller, 1978).

Yellow nutsedge plants are capable of producing large numbers of seeds with varying degrees of dormancy (Thullen and Keeley, 1979). However, seeds are

prone to decay in the seedbed and results from Zimbabwe showed that seedlings rapidly desiccated even under irrigated conditions where less than 1% emerged after 16 weeks (Lapham and Drennan, 1990).

In order to determine the importance of sexual reproduction in the nutsedge lifecycle, Horak and Holt (1986) sampled 20 nutsedge plants from 10 different locations in California and analyzed them for genotypic variability. Plants from each location were considered as individual populations. Sexual reproduction by an outcrossing species implies that the population should consist of a wide range of genotypes. Isozyme analysis was used as an indicator of genetic diversity within and among the populations. Initially twelve loci were evaluated for variability but only four of these were not monomorphic, therefore in total 81 isozyme genotypes were possible (3^n where n is the number of loci and 3 the number of possible genotypes per locus). Among the 200 nutsedge plants that were sampled, only 9 of the possible 81 genotypes were identified; some populations included up to four genotypes while others showed no genotypic variability. Based on these results, sexual reproduction is uncommon and therefore insignificant in the spread and persistence of the weed (Mulligan and Junkins, 1976; Holt, 1987; Horak et al., 1987). Because yellow nutsedge is self-incompatible and seedling survival is rare, asexual reproduction is the predominant means of propagation.

2.2. Yellow Nutsedge - Tuber Biology

Since tubers are the primary reproductive propagules, a large amount of literature exists on the biology, morphology, and physiology of nutsedge tubers. Consolidation of all the available information is beyond the scope of this review

and the following discussion touches only on the most pertinent information as it relates to the objectives of the project.

The initiation and production of tubers is a function of photoperiod, nutrient levels and temperature (Garg et al., 1967). In Ohio, temperature variation is the determining stimulus for the onset of tuberization when the day/night temperature variation is greater than 10 C. Otherwise, photoperiod is more important (Stoller and Woolley, 1983). Jorden-Molero and Stoller (1978) determined that a 14 hour photoperiod was critical for the initiation of tuberization. Friesen and Hamill (1977) reported that in Ontario tuberization was optimal when the photoperiod was 12 hours or less. Plants grown under 16 hour daylengths produced twice the number of shoots but only one-eighth the amount of tuber dry matter compared with those grown under 10-12 hour daylengths.

Differences in the photoperiod required to initiate tuberization may be attributable to ecotypic differences. Yip (1978) determined a correlation between photoperiod responses and the geographical origin of the plants. Under a 12 hour day, tuber formation by northern ecotypes was greater than it was for southern ecotypes. Additionally, tubers of the northern ecotypes were more mature at any given time than those of the southern ecotypes.

One other factor that is important in the initiation of tuberization is the level of nitrogen (N) in the soil. High N levels inhibit tuber production and promote shoot growth. In contrast, low N levels enhance tuber production (Garg et al., 1967).

Tubers are formed at the tips of indeterminate rhizomes through a process that involves a concomitant reduction in rhizome internode length, increase in

rhizome diameter, and accumulation of cellular starch (Bendixen, 1973). Initially tubers are soft and white, becoming darker and harder with increased maturation (Jansen, 1971). The apical end of the tuber is formed into a cone shape as a result of the clustering of cladophylls at this end (Jansen, 1971). Within this conical region, four to five buds develop (Thullen and Keeley, 1975) in an equilateral triangle formation (Lorougnon, 1969). Buds normally sprout from the apical end of the tuber with one to three buds sprouting at a time. The initial sprouts are apically dominant and inhibit the development of other buds (Stoller et al., 1972). The buds of purple nutsedge also sprout from the apical end of the tuber but only one at a time. In this species the most terminal bud exhibits apical dominance over the other buds (Bayer, 1987; Lorougnon, 1969).

Tumbleson and Kommedahl (1962) observed that 12% of the tubers harvested in fall and 95% of those harvested in spring sprouted. The authors concluded that the most newly formed tubers were dormant. Exposure to prolonged cool temperatures and washing with cold water released tubers from dormancy (Tumbleson and Kommedahl, 1961; Thomas, 1967; Stoller and Wax, 1973; Lapham, 1985). It is probable that these processes remove a sprouting inhibitor since an extract from dormant tubers inhibited sprouting when it was applied to non-dormant tubers (Tumbleson and Kommedahl, 1962).

A second type of dormancy, known as conditional dormancy, occurs when the environment around the tuber is not conducive for the tuber to sprout despite the physiological readiness of the buds (Vegis, 1964). Environmental parameters responsible for conditional tuber dormancy include cool soil temperatures (Thullen and Keeley, 1987), low oxygen levels (Bayer, 1987) and low moisture conditions (Friesen and Hamill, 1977). These conditions vary with soil depths

and consequently tuber placement in the soil profile has important ramifications for tuber longevity (Bayer, 1987; van Groenendael and Habekotte, 1988).

In Ontario, tubers formed under moist fall conditions sprouted in spring when soil temperatures warmed, however, if they were formed under dry conditions, tubers became dormant and sprouted erratically (Friesen and Hamill, 1977). Dormant tubers remain inaccessible to most nutsedge control practices and thereby prolong the infestation period. Of those tubers left undisturbed in the soil for three years, 80% decayed (Stoller and Wax, 1973).

2.3. Yellow Nutsedge - Ecotypic Variability

Several publications have mentioned the need for research in characterizing the ecotypic differences within the species *Cyperus esculentus* (Matthiesen and Stoller, 1978; Stoller and Sweet, 1987). Ecotypic variability has been observed in all biological parameters measured on yellow nutsedge (Stoller and Sweet, 1987). Hauser (1968) compared nutsedge plants from Georgia and Delaware and determined that the plants from Georgia grew taller, flowered later, spread further from the parent tuber and produced larger tubers than those from Delaware. Boldt et al., (1976) selected five different populations of yellow nutsedge from New York and grew them at two locations that had a 65 day difference in growing season length. Populations differed in morphological parameters but these were not consistent across locations, therefore the populations were ecotypically different. Locational variability included differences in flower production, numbers of shoots per plant and shoot dry weight per plant. Ecotypic variability has also been observed in nutsedge tuber composition (Stoller and Weber, 1975; Matthiesen an Stoller, 1978).

In a large study conducted by Yip (1978), growth parameters such as rate of shoot production and shoot dry matter accumulation varied among ecotypes. The number of shoots per plant was inversely related to the distance between the primary and higher order shoots. More tubers were produced by ecotypes that produced more shoots, although these tubers were smaller than those produced by ecotypes with fewer shoots (Yip, 1978).

Yellow nutsedge has been categorized into different varieties based on mophological and inflorescence characteristics, primarily spikelet morphology. According to Kuekenthal's descriptions, two main varieties exist in North America: var. *leptostachyus* and var. *esculentus* (Yip, 1978). The main distinguishing characteristics are as follows:

var. *leptostachyus* - tubers large, up to 2 cm long; spikes branched at the base, spikelets rather long (15-20 mm) with narrowed to acute tip.

var. *esculentus* - rosette leaves in a regular triangle pattern, rosettes more open than in others; tubers spherical with an acute top; spikelets rather short (5-12 mm).

In the United States, the variety *esculentus* is present in temperate climates and var. *leptostachyus* in the more moderate regions (Yip, 1978).

Other varietal differences have also been reported that are not used for taxonomic identification. The distance between the primary shoot and higher order shoots was greater among var. *leptostachyus* than var. *esculentus*. Tuber weight of var. *leptostachyus* was affected more by shading than that of var. *esculentus* (Borg et al., 1988). Variety *leptostachyus* was also found to be less frost tolerant than var. *esculentus*. Variability in herbicide susceptibility has also

been observed between these two varieties (Costa and Appleby, 1976, Yip, 1978). Variety *esculentus* was more tolerant to atrazine and metribuzin and more susceptible to 2,4-D oil-soluble amine than var. *leptostachyus* (Costa and Appleby, 1976).

Researchers in the Netherlands have identified four taxa that have recently established themselves as a result of contaminated gladiolus bulb shipments from different parts of the world (Borg et al., 1988). Both varieties *esculentus* and *leptostachyus* were identified, as well as var. *cyclolepis* and one other variety that has not been identified to date. This example underscores the importance of preventing the transport of contaminated goods. Further, varietal classification will assist in providing appropriate control practices for the different nutsedge varieties.

2.4. Yellow Nutsedge - Growth Analysis

Growth analysis has been a useful technique in developing an understanding of the dynamic growth of yellow nutsedge. Although very little of these data have been fitted to models, the descriptive nature of growth analysis has assisted in defining the proliferation of nutsedge tubers and the partitioning of biomass.

In Zimbabwe, the growth of yellow nutsedge was examined over a two year period. Tuber production was exponential in both growing seasons with 17 681 tubers being formed by a single plant in the first year, increasing to 163 004 tubers in the second year (Lapham, 1985). This is the highest reproductive potential of yellow nutsedge recorded to date. In Minnesota, 6 900 tubers were produced in one year from a single tuber (Tumbleson and Kommedahl, 1961).

Single nutsedge plants produced 332 tubers after 16 weeks of growth in Minnesota and 1 073 tubers in Zimbabwe (Tumbleson and Kommedahl, 1961; Lapham, 1985). After 17 weeks of growth in Georgia, a single nutsedge plant formed 662 tubers (Hauser, 1968).

Clonal growth is a measure of the soil area covered by a single nutsedge plant over the growing season. Lapham (1985) used clonal growth as a measure of the intrinsic growth rate of yellow nutsedge. In Zimbabwe, clonal growth of yellow nutsedge was described by the equation $y=a(1-\exp(-bt^2))$ where y is the clonal radius, t the time after first growth and a and b are constants (Lapham, 1985). After two years of growth, a single clone covered an area of 21.7 m^2.

By using growth analysis to compare yellow and purple nutsedges, differences in tuber production and biomass distribution were determined between the species (Williams, 1982). Tuber production by purple nutsedge was described by an exponential equation, whereas production by yellow nutsedge was described by a linear function. Tubers comprised up to 50% of the total biomass of purple nutsedge compared to only 28% of the total biomass of yellow nutsedge (Williams, 1982). An increase in biomass allocation to yellow nutsedge tubers towards the end of the growing season resulted in a concomitant reduction in partitioning to roots and rhizomes. In contrast, shoot biomass partitioning remained unchanged.

2.5. Freezing Tolerance

The ability of yellow nutsedge tubers to survive freezing temperatures has been implicated as the primary factor responsible for the weed's widespread

distribution, particularly in comparison to purple nutsedge which occurs over a narrower geographic range (Bendixen, 1973; Stoller, 1973; Stoller and Weber, 1975; Mulligan and Junkins, 1976). In Illinois, the lethal temperature at which 50% of yellow nutsedge tubers were killed (LT_{50}) was determined to be -6.5 to -7.0 C in experiments using a temperature gradient bar (Stoller and Wax, 1973; Stoller, 1973). The duration of exposure to these temperatures did not significantly influence tuber viability. These results were not consistent with the results from field studies where tubers survived temperatures of -15 C (Stoller and Wax, 1973).

Nutsedge tubers buried in the fall at 2.5 and 5 cm below the soil surface did not produce as many shoots as those placed deeper in the soil, presumably because many of those nearer to the soil surface winterkilled (Stoller and Wax, 1973). With increasing depth in the soil, tubers avoid extreme cold temperatures because of the insulation provided by the soil and/or snow (Stoller, 1973). However, insufficient information about winter soil temperatures precludes the precise identification of critical freezing temperatures for tubers.

Stoller and Weber (1975) investigated nutsedge tuber composition and its relationship to cold hardiness. Cold treatment of purple nutsedge and several yellow nutsedge ecotypes caused an increase in the starch, sugar, and lipid fractions of the more cold tolerant ecotypes. Additionally, cold hardiness in yellow nutsedge appears to be related to a higher proportion of unsaturated fatty acids in tuber triglycerides. Thus, differences in tuber composition may be part of the hardening process that permits tubers to survive freezing temperatures (Bendixen and Nandihalli, 1987).

2.6. Desiccation Tolerance

An important component of perennial weed control is tillage where vegetative growth is destroyed by depletion of energy reserves and desiccation (Ross and Lembi, 1985). Breaking up plant tissues will encourage those perennating organs left in the soil to regrow and deplete food reserves. Additionally, cultivation carries fleshy underground structures to the soil surface where they are exposed to drying conditions. The ability of vegetative structures to survive desiccation decreases the effectiveness of tillage as a weed control practice.

The effect of cultivation on reducing nutsedge tuber viability has been reported previously (Day and Russel, 1955; Thullen and Keeley, 1975; Keeley et al., 1983; Lanini, 1987). Day and Russel (1955) conducted field studies with purple nutsedge and showed that tillage under dry conditions effectively killed tubers. Smith and Mayton (1937) reduced the number of viable purple nutsedge tubers by 80% after plowing at four week intervals during a single growing season. Tumbleson and Kommedahl (1961) collected yellow nutsedge tubers from the soil surface immediately after cultivation and 90 percent sprouted. However, when these tubers were left on the soil surface for 2 days after cultivation only 10 percent sprouted (Tumbleson and Kommedahl, 1961). Deep tillage did not effectively kill tubers since it placed tubers deeper in the soil profile where tuber viability was prolonged (McDonald, 1986).

Several studies have been conducted on the desiccation tolerance of vegetative propagules of other difficult-to-control perennial weeds. The viability of quackgrass (*Elytrigia repens*) rhizome pieces was reduced to 40% after being exposed to 56% relative humidity for 3 days. As the time of exposure increased

there was a continual decline in viability (Grummer, 1963). Tubers of purple nutsedge (*Cyperus rotundus* L.) were found to have a critical moisture content of approximately 10% following storage in dry soil for 10 days (Horowitz, 1972). Purple nutsedge tubers were more tolerant of desiccation than single node rhizome fragments of johnsongrass (*Sorghum halepense* (L.) Pers.) and less tolerant than bermudagrass (*Cynodon dactylon* L.) rhizomes. Thomas (1969) reported that rhizomes of *Cynodon dactylon* remain viable until they reach a moisture content of approximately 10%.

Day and Russel (1955) determined that yellow nutsedge tubers were more tolerant of desiccation than purple nutsedge. As moisture content of yellow nutsedge tubers decreased there was a trend towards reduced tuber viability but no significant relationship could be established. After 25 days storage in dry soils, 36% of the yellow nutsedge tubers were viable compared with no viable purple nutsedge tubers after only 14 days storage (Day and Russel, 1955). In an experiment where relative humidity was reduced in a stepwise progression from 90 to 30% over a 12 week period, nearly half of the *Cyperus esculentus* tubers survived (Thomas, 1969). Greater reductions in viability occurred when tubers were placed directly in air dry soil (82% nonviable) as compared to those that underwent progressive desiccation in humidity chambers (60% nonviable). Nutsedge tubers stored in air dry soil desiccated more rapidly than those that underwent progressive desiccation and resulted in greater tuber mortality (Thomas, 1969).

2.7. Influence of Crop Interference on Nutsedge Growth

Yellow nutsedge lacks many of the characteristics normally associated with competitive weeds. Because of its relatively short stature and its C4 pathway of photosynthesis, crop yield losses are minimized when nutsedge is shaded (Stoller, 1981). However, in row crops where canopy closure may be slow, yellow nutsedge is often highly competitive (Patterson et al., 1980).

In both row and non-row crops, the time of crop emergence relative to weed emergence is an important determinant of competitiveness. Cotton growing for two weeks prior to nutsedge emergence reduced nutsedge shoot numbers to 29 shoots m^{-2} compared with 61 shoots m^{-2} when they emerged together (Keeley et al., 1983). Delayed seeding of corn in Quebec until early June permitted the formation of a wide variety of nutsedge tuber sizes. In contrast, corn seeded by the third week of May caused tuber production to shift towards the formation of smaller tubers (Ghafar and Watson, 1983a). This may be important since smaller tubers do not remain viable as long as large tubers (Thullen and Keeley, 1975) and are less tolerant of freezing temperatures (van Groenendael and Habekotte, 1988). In addition, smaller tubers produce shoots with less vigor than those emanating from larger tubers (Stoller et al., 1972; Stoller and Wax, 1973).

Investigations on the influence of crop interference on nutsedge growth have concentrated on determining biomass distribution and tuber production. Studies designed to determine the influence of shade on yellow nutsedge growth have shown a direct correlation between the amount of light and the number of tubers and shoots and total dry matter accumulation. In California, reducing incident radiation by 30 and 80% reduced tuber production by 32 and 80%, respectively

(Keeley and Thullen, 1978). Providing 80 and 94% shade to nutsedge plants for a 3 month period limited tuber production to 381 and 51 tubers per plot, respectively, (Keeley and Thullen, 1978) indicating that even very dense shade did not prevent tuber production entirely (Jordan-Molero and Stoller, 1978; Patterson, 1982). Under 73% shade, tuber production of yellow and purple nutsedges was 12 and 28%, respectively, of where there was no shade (Jordan-Molero and Stoller, 1978). Shading yellow nutsedge plants resulted in a shift in biomass partitioning from the tubers to the leaves. Shaded nutsedge plants, although having fewer leaves in total, produced the same area per leaf as non-shaded plants with reduced dry matter per leaf (Patterson, 1982). Shading also influenced the onset of tuberization. Under an open canopy, tuber formation began earlier than under a dense canopy (Fischer, 1987).

In interference studies, an increase in corn population density reduced light penetration and caused a reduction in above ground biomass, tuber number and tuber weight of yellow nutsedge (Ghafar and Watson, 1983b). Seeding corn at 33 300 plants ha^{-1}, one half the normal seeding rate, resulted in a 44% increase in tuber production compared to the normal seeding rate. In contrast, doubling the normal seeding rate of corn reduced tuber production by 71% compared to the normal seeding rate (Ghafar and Watson, 1983b).

2.8. Integrated Management of Yellow Nutsedge

Control of yellow nutsedge cannot be obtained through the use of a single weed control practice, but rather requires the integration of several weed control tools (Stoller, 1981; Glaze, 1987). Eradication of yellow nutsedge requires several years of continuous attention. A number of weed management practices that

should be considered in planning a yellow nutsedge control program include; cultivation, date of crop planting, crop density, competitive crops and crop cultivars, crop rotation, fertilization, and use of both selective and nonselective herbicides.

Cultivation is an integral component of nutsedge control since this can both kill sprouted plants and stimulate tubers to sprout thereby depleting tuber energy reserves (Thullen and Keeley, 1975). In addition, cultivation prior to herbicide application stimulates tubers to sprout and enhances the effectiveness of the herbicide. Shallow tillage leaves tubers near the soil surface facilitating even sprouting and promoting winterkill (McDonald, 1986). Tillage may also aggravate nutsedge problems by spreading tubers to uninfested areas. Therefore, careful cultivation is required in order to provide effective control of spotty infestations (Stoller, 1981).

In choosing a crop to plant on nutsedge infested areas, important considerations include the rapidness of crop canopy closure, the number of days to crop maturity and the choice of available herbicides. In corn there are several selective herbicides for nutsedge control and once established, the crop creates a dense canopy (Habekotte and van Groenendael, 1988). Barley is also effective because it is a short season crop that produces a dense crop canopy very rapidly. In Holland, growing hemp without any herbicides was as effective in reducing the nutsedge population as growing corn and applying a herbicide (Habekotte, 1988).

In addition to selecting a highly competitive crop to suppress nutsedge, it is important to select the best crop variety. Of three potato cultivars growing with

yellow nutsedge, 'Green Mountain' competed more effectively than 'Norchip' or 'Hudson' (Yip et al., 1974). The greater competitiveness of 'Green Mountain' was ascribed to the cultivar's early emergence, rapid growth and persistent dense leaf canopy throughout the growing season.

Long term studies into effective crop rotations have provided the most useful data on nutsedge control over more than a single growing season. In all of the crop rotation studies relatively good control of nutsedge was attained within three years (Keeley et al., 1979; Keeley et al., 1983). In Illinois, less than 20% of the tubers were able to survive in the soil for more than three years (Stoller and Wax, 1973). Fallowing with tillage after growing barley reduced tuber populations by 98% in California within three years. Similarly, double cropping potatoes with milo or double cropping EPTC-treated potatoes with alachlor-treated soybean resulted in 97 and 99% reductions in tuber populations, respectively (Keeley et al., 1983). In a different experiment, growing alfalfa treated with EPTC for two years or double cropping barley with corn treated with butylate for two years reduced nutsedge tuber populations by 96% (Keeley et al., 1979). These studies show that applying continuous pressure on nutsedge, either through cultivation, crop competition and/or herbicides, effectively reduces tuber populations.

The best nutsedge control programs are those that prevent the production of tubers. In order to prevent tuber production the onset of tuberization must be defined and the influence of different agronomic practices on tuberization must be understood. Once these factors are learned, effective control of nutsedge can follow. For example, in Illinois, this means that nutsedge plants must be controlled before the beginning of August (Jordan-Molero and Stoller, 1978).

If yellow nutsedge management strategies are to be effective they must be incorporated into a combined effort using all of the research available. Models incorporating available management tools have been developed (Lapham, 1987; Habekotte,. 1988). Since nutsedge growth is variable as a result of ecotypic variation and climatic differences, models although useful must be adapted or developed for each geographic location. For example, evaluation of the cost effectiveness of controlling yellow nutsedge in tobacco in Zimbabwe involved an understanding of nutsedge reproductive potential and the effectiveness of each of the control practices. Based on this research, the economic threshold for controlling yellow nutsedge was determined to be 1.5 tubers m^{-2} (Lapham, 1987). In a population dynamic model developed for use in Holland, growing hemp alone reduced nutsedge tuber production more than did growing corn where herbicides were used (Habekotte, 1988). As more information is gathered on the growth of yellow nutsedge and the individual control practices, the models will become more effective and predictive.

3. Yellow Nutsedge Distribution in Manitoba

3.1. Introduction

Yellow nutsedge is a problem perennial weed on all continents and is recognized as a major problem weed in nearly 40 countries in 21 crops (Bendixen, 1987). Traditionally it has been a weed in the more tropical areas of the world, but has recently moved into cooler habitats (Holm et al., 1970). The wide spread distribution of yellow nutsedge has been ascribed to the ability of the tubers to withstand environmental extremes (Stoller and Weber, 1975). In the United States, the differential spread of *C. esculentus* and *C. rotundus* has been related to cold soil temperatures in winter (Stoller, 1973).

In addition to differences in geographical distribution among Cyperus species, yellow nutsedge varieties also have different distribution patterns. *Cyperus esculentus* var. *leptostachyus* is present in the warmer states of the U.S. and var. *esculentus* in the temperate regions (Yip, 1978). Prior to its establishment in Manitoba, yellow nutsedge has persisted in Canada as a weed problem in Nova Scotia, New Brunswick, southern Quebec and southern Ontario (Mulligan and Junkins, 1976).

The tuber of yellow nutsedge is the primary reproductive structure and therefore is responsible for both maintaining old infestations and establishing new ones. Lack of genetic diversity within and among populations of yellow nutsedge confirms that it primarily reproduces asexually (Holt, 1987).

Local increases in yellow nutsedge infestations are attributable to changes in agronomic practices over the last two decades. These changes include: 1) an

increase in the use of herbicides for annual weed control, 2) a reduction in the amount of cultivation and hand hoeing, 3) an increase in mechanized farming and 4) an intensification of the use of inputs to maximize yields (Hauser, 1968). These factors have encouraged a shift towards perennial weed development by eliminating or reducing competition from annual weeds and reducing disruption of the weed's perennial cycle.

Yellow nutsedge was first identified in Manitoba near Emerson in 1979. Tubers of yellow nutsedge were probably carried northward from the United States with Red River floodwaters in 1979 and deposited in fields when the flood receded (Sturko, 1979). In consideration of the potential economic importance of this weed a survey was undertaken to identify the extent of yellow nutsedge infestation in Manitoba.

3.2. Materials and Methods

Information pamphlets (Figure 1) were sent to 640 landowners along the Red River from Winnipeg south to the United States border. The distribution of pamphlets was limited to landowners along the Red River based on the presumption that the Red River floodwaters were responsible for moving tubers into the province in 1979. Recipients were asked to report nutsedge infestations which would in turn be investigated for positive identification. Names and addresses were obtained from tax evaluation forms at the rural municipal offices. Results from the survey were tabulated (Appendix 1) and also identified on a topographical map (Figure 2). In addition to the survey, fields were inspected by the district weed supervisor and the author, and awareness of the problem was raised at local farm meetings.

YELLOW NUTSEDGE ALERT!

DO YOU HAVE THIS WEED?

Figure 1: Information pamphlet used in determining the geographic distribution of yellow nutsedge.

Identification

The leaves of yellow nutsedge are grass-like, 0.5-1.5 cm wide and 20-50 cm in length, narrowing (pinched) at the leaf tip, and have a prominent midvein. The seed head is a yellow-gold colored umbel, a more or less flat topped inflorescence (like dill), that is borne on a triangular stem 15-75 cm tall. Yellow nutsedge rhizomes are slimmer than those of quackgrass (*Agropyron repens*), however, nutsedge rhizome fragments do not give rise to shoots. Small tan to brown tubers, about the size of a large pea seed, form at the ends of yellow nutsedge rhizomes from early August until a killing frost occurs. Tubers can germinate the year after they are produced or remain dormant in soil for three or more years. Approximately 80% of the tubers are produced in the top 15 cm (6") of the soil. However, tubers can be produced deeper in the soil.

Habitat

Yellow nutsedge is capable of growing in a wide range of soil types, however it does not grow well under drought conditions. In Manitoba, nutsedge is found along the Red River particularly in low lying areas that are subject to flooding in spring. It commonly grows in dense patches that vary in size from a few square meters to 0.5 hectares or more depending upon the age of the infestation.

Control

Yellow nutsedge is a troublesome, difficult-to-control weed. It is almost impossible to eradicate due to the presence of underground tubers. Research conducted in the U.S. indicates that yellow nutsedge tubers can be killed by desiccation or freezing if brought to the soil surface by tillage operations. To avoid spreading nutsedge tubers, patches should be worked separately and all equipment in contact with soil thoroughly cleaned. Yellow nutsedge is shade-intolerant, and growing a highly competitive crop (cereals or rapeseed) will reduce tuber production. Growing less-competitive crops like lentils, peas, or flax is not recommended. Killing nutsedge shoots by tilling immediately following harvest will reduce tuber production. Most of the herbicides that offer selective control of yellow nutsedge are limited to use in corn-soybean rotations. Repeated post-emergence applications of high rates of the nonselective herbicide, Roundup, will provide partial control of nutsedge patches.

Dispersal

In Manitoba, it appears that yellow nutsedge propagates only vegetatively by tubers; viable seed is not formed. Therefore tuber movement is required for long distance dispersal. Contaminated tillage equipment and other machinery is the most common way this weed is spread. Root crops and bedding plants will contribute to the spread of yellow nutsedge tubers if the product is contaminated.

Research

The growth and competitiveness of yellow nutsedge is being investigated at the University of Manitoba Glenlea Station with the objective of developing control practices applicable to Manitoba. This research is being funded by the Governments of Canada and Manitoba through the Agri-Food Program under the Economic and Regional Development Agreement (ERDA Program). Your assistance is required in identifying sites of yellow nutsedge infestation to help document the scope of the current problem and the potential spread of this weed. This information will be part of a masters thesis and is greatly appreciated.

Finding Yellow Nutsedge

Since yellow nutsedge emerges relatively late in spring and initial growth is slow, the best time to look for nutsedge in fields is after harvest in annual crops, and after cutting hay. If this weed is on your farm please fill out the attached form and mail it. We will contact you after receiving the form to confirm the findings and answer any questions you have regarding this weed. Contact your Weed Supervisor or Ag. Rep. for any additional information.

YELLOW NUTSEDGE INFESTATION REPORT FORM

Date: _____

Name: _____

Address: _____

Telephone No.: _____

Size of Infestation: _____

Legal Land Location: _____

Additional Notes: _____

Figure 1: Information pamphlet used in determining the geographic distribution of yellow nutsedge.

3.3. Results and Discussion

Based on taxonomic properties of the inflorescence, including length and shape of the spikelets, the yellow nutsedge variety present in Manitoba was identified as var. *esculentus*. This is consistent with the results of Yip (1978) who reported that var. *esculentus* was more prominent in temperate climates than var. *leptostachyus*.

The survey was primarily sent out to raise awareness to the possible presence of the weed. Secondarily it served as a reporting mechanism facilitating feedback from the landowners to the University of Manitoba. Only respondents that suspected a nutsedge infestation were asked to reply. In total 13 replies were received helping to form the distribution map (Figure 2).

Based on the responses to the information pamphlet and field visits, it is apparent that the nutsedge problem is increasing. Currently many of the infestations consist of patches several meters in diameter (Appendix 1). However, in two fields that have been monitored closely for the past several years, nutsedge has spread to other areas of the fields and the small patches have rapidly increased in size. This is largely the result of cultivation which transports tubers from one area to another.

All infestations that have been identified to date were in fields bordering the Red River (Figure 2) reinforcing the presumption that the tubers spread in flood water (Sturko, 1983). To date 21 infestations have been identified in Manitoba (Appendix 1), the majority occurring near St. Jean Baptiste which is the location

of the most convoluted part of the Red River in Manitoba (Figure 2). The reasons for this concentration of infestations near St. Jean Baptiste are undetermined, however, it is hypothesized that the tortuousity of the river may have delayed movement of the floodwaters promoting greater soil and tuber deposition.

One exception to the location of infestations occurred during the summer of 1989 where a single nutsedge plant was found in a strawberry field near Glenboro, Manitoba. Strawberry plants imported from New Brunswick were determined to have been contaminated and the nutsedge plant was eradicated. Widespread distribution of this weed commonly occurs through the transport of agricultural produce contaminated with nutsedge tubers. American shipments of gladiolus cormlets contaminated with tubers of yellow nutsedge in the early 1970's introduced this weed into the Netherlands (van Groenendael and Habekotte, 1988). These cormlets are used in the gladiolus bulb industry which is threatened by the presence of yellow nutsedge. At least four different varieties of yellow nutsedge have been identified in the Netherlands indicating that several unique infestations occurred rather than only one (Borg et al., 1988). This clearly demonstrates the vulnerability of the agricultural industry to magnification of weed problems.

The best way to control yellow nutsedge is by preventing its movement into noncontaminated soil. Clearly a large part of a provincial weed management strategy for yellow nutsedge must focus on early identification of infestations and preventing distribution. The information pamphlet (Figure 1) was part of such an extension effort which must be continued if yellow nutsedge containment is to be achieved.

4. Yellow Nutsedge Growth and Ecotypic Variability

4.1 Introduction

4.1.1 Growth Analysis

The growth and development of yellow nutsedge has been investigated and reviewed extensively (Mulligan and Junkins, 1976; Stoller and Sweet, 1987). The widespread distribution of this weed is ascribed to intraspecies variability that has encouraged the development of many ecotypes suited to many different environments (Yip, 1978; Stoller and Sweet, 1987). Differences among the yellow nutsedge ecotypes have been quantified through growth analysis (Hauser, 1968; Boldt, et al., 1976; Yip, 1978) and variability has been reported in a range of biological parameters (Stoller and Sweet, 1987).

Research on the growth of yellow nutsedge has focused primarily on the production of tubers since they are the primary reproductive structure. Single yellow nutsedge plants growing for 16 weeks produced 332 and 1 073 tubers in Minnesota and Zimbabwe, respectively, and 662 tubers after 17 weeks of growth in Georgia (Tumbleson and Kommedahl, 1961; Hauser, 1968; Lapham, 1985). By the end of the growing season, up to 28% of the total biomass of yellow nutsedge consisted of tubers compared to 50% for purple nutsedge (Williams, 1982).

4.1.2. Cold Tolerance of Tubers

Physiological differences between ecotypes may be of greater significance than morphological differences. The increasing number of yellow nutsedge infestations in cooler habitats is indicative of greater adaptation of the weed to

survival under freezing temperatures (Holm et al., 1970; Stoller, 1973). In Illinois, the lethal temperature at which 50% of yellow nutsedge tubers were killed (LT_{50}) was determined to be -6.5 to -7 C in experiments using a temperature gradient bar (Stoller and Wax, 1973; Stoller, 1973). In the field, tubers have survived temperatures of -15 C indicating that other factors such as a longer cold hardening period must be involved (Stoller and Wax, 1973).

4.1.3. Desiccation Tolerance of Tubers

Cultivation is an important component of perennial weed control, because it destroys vegetative tissues by starvation and desiccation (Ross and Lembi, 1985). Research into the effect of desiccation on the viability of different vegetative reproductive structures has shown that differences in desiccation tolerance exist and contribute to the variability in weediness of different species (Grummer, 1963; Horowitz, 1972). Thomas (1969), exposed yellow nutsedge tubers to desiccation through a stepwise progression from 90 to 30% relative humidity (RH) over a 12 week period. Nearly half of the tubers survived this treatment. Greater mortality was observed among tubers placed directly in air dry soil (82% nonviable) than those undergoing sequential desiccation in humidity chambers (60% viable). Placing tubers directly in air dry soil invoked a more rapid rate of desiccation and resulted in greater mortalities (Thomas, 1969).

Variable results and inconsistent techniques make it difficult to draw comparisons between much of the research conducted on the desiccation tolerance of vegetative propagules. The objectives of this research were to determine if tubers of *Cyperus esculentus* found in Manitoba exhibit tolerance to desiccation

and to develop a standardized technique for determining desiccation tolerance of vegetative reproductive propagules.

4.2 Materials and Methods

4.2.1 Growth Analysis - Pots

Yellow nutsedge tubers were presprouted in petri plates. When the primary rhizomes were approximately 1 cm in length, sprouted tubers were planted to a depth of 4 cm in 6.5 liter plastic pots, four per pot. This density was equivalent to 127 tubers m^{-2}. The soil was an Altona Clay Loam with 40% clay, 27% silt and 33% sand with a pH of 8.0. At planting the nutrient levels were 172, 124 and 1 470 kg/ha of N,P, and K, respectively[1]. The plants were grown outside from the first week of June until mid September and were fertilized once per month with 20-20-20 nutrient solution and watered as required.

Two ecotypes were included in the study, one from Harrow, Ontario[2] (ONT) and the second from Aubigny, Manitoba (MB). Plants were destructively sampled 36 and 90 days after emergence (DAE) in 1988 and 18, 36, 54, and 90 DAE in 1989. At each sampling date five pots of each ecotype were sampled. The following parameters were measured: leaf area[3], leaf number, shoot number, tuber number and rhizome length. The dry mass of each of these components was also determined. In addition, shoot counts were made at frequent intervals, generally 7-12 days apart.

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1 Manitoba Provincial Soil Testing Lab, Ellis Building, University of Manitoba
2 Tubers were obtained from Allan Hamill, Agriculture Canada, Harrow, Ontario
3 Portable Leaf Area Meter, Model LI-3000, Li-Cor., Lincoln, NE.

4.2.2 Growth Analysis - Field

Tubers of yellow nutsedge were presprouted in petri plates and planted in the field at the University of Manitoba Glenlea Research Station on June 7, 1988 and May 24, 1989. The sprouted tubers were planted in a field that had been broken from an alfalfa/bromegrass stand in 1987. The soil was a Red River Clay with a pH of 7.6. Nutrient levels were 92, 108, and 1 260 kg/ha of N,P, and K, respectively, in 1989[1]. The site was typical of many of the locations infested with yellow nutsedge in Manitoba in that it was located adjacent to the Red River and susceptible to flooding in spring.

Individual tubers were space planted 4 cm deep, 2 meters apart in a grid pattern. The experimental area was hand weeded regularly. Eight plants were sampled every 16 days beginning at plant emergence. After 80 DAE the sample size was reduced to five plants because the plants were too large. At each sampling date the diameter of each plant was measured after which the entire plant was dug up. This usually entailed excavation to at least 12 inches below the soil surface. Plants were separated into leaves, inflorescences, roots, rhizomes and tubers.

In 1988, growth analysis of field grown plants was limited to MB, but in 1989, both ecotypes were grown in the field to confirm the results from the pot experiments. The following parameters were measured: leaf area, leaf number, shoot number, inflorescence number, rhizome length, tuber number, soil area (clonal spread) and the biomass of each plant component. Clonal spread was calculated from the plant diameter measured prior to excavation by the formula πr^2, based on the assumption that plants spread in a circular pattern. Rhizome length, leaf area, leaf number and tuber number were determined by

extrapolation from plant subsamples on a dry mass basis. Subsample size was dependent on total plant size and decreased from 50% initially to 10% near the end of the growing season. Dry mass was obtained by drying the plant material at 80 C for 48 hours.

Meteorological data was obtained from Environment Canada from a recording station located in the field adjacent to the Glenlea field site. This data was compared with the 30 year mean in a seasonal subseries graph (Figure 3) (Cleveland and Terpenning, 1982).

4.2.3 Statistical Analysis - Pot and Field Growth Analysis

All data collected from pot grown nutsedge at the final sampling date were analyzed using analysis of variance and the means were separated using Duncan's multiple range test (p=0.05).

The field data were analyzed using nonlinear regression techniques. In most cases a logistic model was fitted to the primary data (Hunt, 1982) using a derivative-free nonlinear regression procedure (Freund and Littell, 1986). The logistic model and its use have been reviewed previously (Hsu, et al., 1984; Nickel, 1989).

4.2.4 Critical Freezing Temperature of Tubers

Tubers were harvested from an established nutsedge infestation at the University of Manitoba Glenlea Research Station. Tuber dormancy was broken by washing tubers for 12 hours under running cold water and storing at 2 C for 4 to 6 weeks

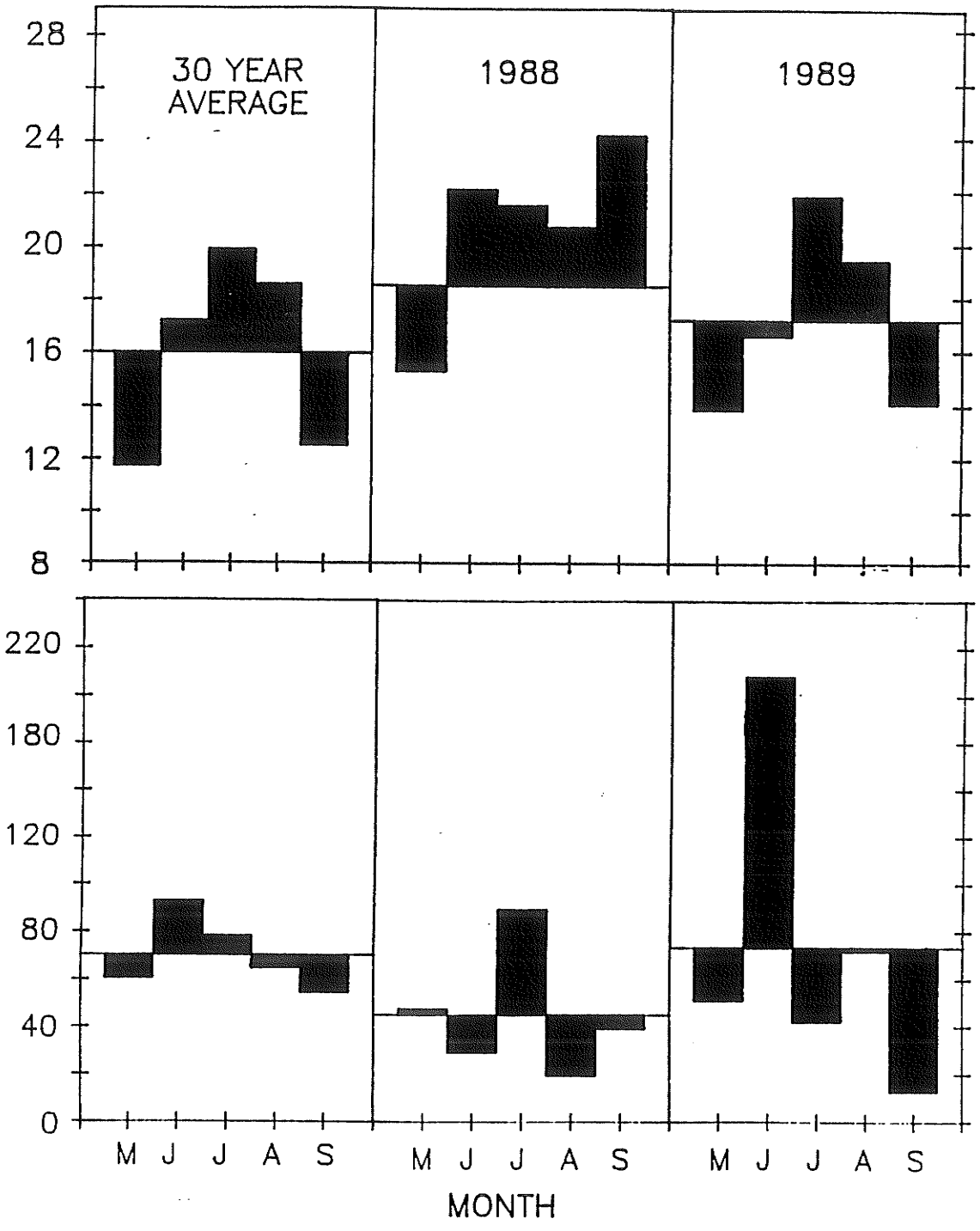

Figure 3: Seasonal subseries comparisons of monthly mean temperatures and precipitation for 1988 and 1989, and the 30 year average at Glenlea, Manitoba.

(Lapham, 1985; Stoller and Wax, 1973; Thomas, 1967; Tumbleson and Kommedahl, 1961). These tubers were sorted to reduce variability in size, color or degree of mechanical injury and treated with a fungicide solution (Appendix 2). Tubers weighed approximately 0.35 grams and were 10 to 12 mm in diameter.

The freezing apparatus was similar to that described by Wright (1984) and consisted of an 80 L plastic barrel that was filled with a solution containing 70% ethylene glycol and 30% water. The barrel was wrapped in fiberglass insulation and placed in a freezer kept at -30 C.

Ten tubers were placed in 10x10 cm plastic bags containing 30 g of sand and 1 ml of water. The bags were heat sealed and then placed in a second bag and heat sealed again to ensure that tubers would not contact the ethylene glycol solution. Similarly thermocouples were heat sealed in identical bags without tubers and temperature was recorded at 15 minute intervals using a datalogger.[4] Five tuber samples of each ecotype were attached to wire grids to facilitate removal of the tubers from the barrel. Prior to placing tubers in the ethylene glycol solution they were stored at -3 C for 24 hours. Once the temperature in the barrel fell to -3 C, the prefrozen tubers were placed into the ethylene glycol solution and removed at increments of 3 C down to -21 C.

Upon removal from the barrel, tubers were retained at 2 C for 24 hours. The tubers were then placed in plastic petri dishes with one piece of filter paper per dish and watered to keep moist. The tubers were incubated at 22 C for a period of 6 weeks. Sprouted tubers were removed from the plates as they sprouted

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[4] Omega Data Logger, Model OM205, Stamford CT.

leaving dormant and dead tubers for the duration of the incubation period. These remaining tubers were cut in half and dormant and dead tubers were separated based on the hardness and color of the tissue inside. White, hard tissue was associated with dormant, viable tubers and yellow to brown, soft tubers were deemed nonviable (Stoller, 1973; Stoller and Wax, 1973; Yip, 1978). The experiment was repeated twice, however, in the first run only MB tubers were used whereas in the second run both MB and ONT tubers were used. The results from both experiments were combined for analysis since the results were similar.

4.2.5 Tuber Viability and Winter Soil Temperatures.

Winter soil temperatures were monitored using dataloggers[5] housed in an insulated wooden box at the Glenlea Research Station. In 1988, tubers were buried 5, 10 and 15 cm below the soil surface with three repetitions per depth. A copper-constantan thermocouple was inserted next to the tubers. Three plastic mesh sacks, each containing 200 tubers, were placed at each of the three depths in a randomized design. In spring the tubers were recovered, treated with fungicide (Appendix 2), and checked for viability using criteria mentioned previously (4.2.4). In 1989, all tubers were placed 10 cm below the soil surface and attempts were made to regulate the snow depth. A structure was built to keep snow off the ground and yet permit ambient temperatures to reach the soil surface. In addition, a snow screen was placed directly south of a second treatment with the intent of blocking snow to increase the depth of snow cover over three of the buried sacks. The third treatment was left as natural snow cover.

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[5] Li-Cor Data Loggers, Model Li-1000, Li-Cor Inc., Lincoln, NE.

The influence of freezing temperatures on tuber viability was evaluated by plotting tuber viability versus cold units (CU). Cold units were calculated as the degree days of temperatures below -12.5 C during the 6 month overwintering period. The temperature of -12.5 C was chosen based on the data obtained from the critical freezing experiment where -12.5 C was the temperature at which 50% of the tubers would no longer sprout. CU were calculated from November through April of each year by: CU (degree days) = days($T_{-12.5C}$ - $T_{<-12.5C}$) (Gardner and Barnet, 1990). This type of evaluation was required since the cumulative effect of the winter temperatures was being considered rather than an individual parameter such as the minimum temperature attained during the winter.

4.2.6 Tuber Desiccation

Yellow nutsedge tubers used in the desiccation experiments were collected from the University of Manitoba Glenlea Research Station where a nutsedge infestation was established in 1988. Tubers were harvested in May and October of 1989. Of those tubers recovered in May, 6% were dormant and 20% were nonviable, probably the result of winterkill. All of the tubers recovered in October were dormant. Tuber dormancy was broken by washing tubers as described previously (4.2.4). After this treatment less than 1% of the tubers were determined to be either dormant or dead. Prior to initiation of the experiment, tubers were treated with fungicide (Appendix 2).

The desiccation chambers were designed similar to the constant humidity chambers used by Wiebe (1983) to determine the effect of water vapour uptake on wheat seed germination. The chamber consisted of a glass Mason canning

jar (1 L) with a 1 cm diameter hole cut in the lid. Ten tubers were placed in a fine grade aluminum mesh basket that was suspended by a brass wire in the desiccation chamber. The 1 cm hole in the jar lid was sealed with a rubber disk that functioned to both seal the jar and hold the aluminum basket at a constant height (Figure 4).

A constant RH was achieved by placing 75 ml of a super-saturated salt solution in each jar. Each salt solution has a characteristic vapor pressure and in a sealed container will maintain a constant RH. The solution was super-saturated in order to prevent changes in humidity as water vapor moved out of the tubers or leaked into the jar. The salt solutions used were: ammonium sulfate (81% RH); sodium nitrate (66% RH); and calcium nitrate (52% RH). These desiccation treatments were conducted under two temperature regimes, 22 and 30 C.

Ten tubers were weighed and suspended in each desiccation chamber, 5 cm from the container bottom. Daily measurements of the tuber weight were made by suspending the wire mesh assembly containing the tubers on a frame that surrounded the jar, yet permitted weighing the tubers without the jar. Measurements were made until no change in tuber weight was observed (approximately 8 days), at which point the tubers had equilibrated with the RH in the chamber.

Tubers were plated out and incubated at 22 C for 6 weeks to determine viability (Yip, 1978; Stoller and Wax, 1973; Stoller, 1973). Viability was determined as described previously in section 4.2.4.

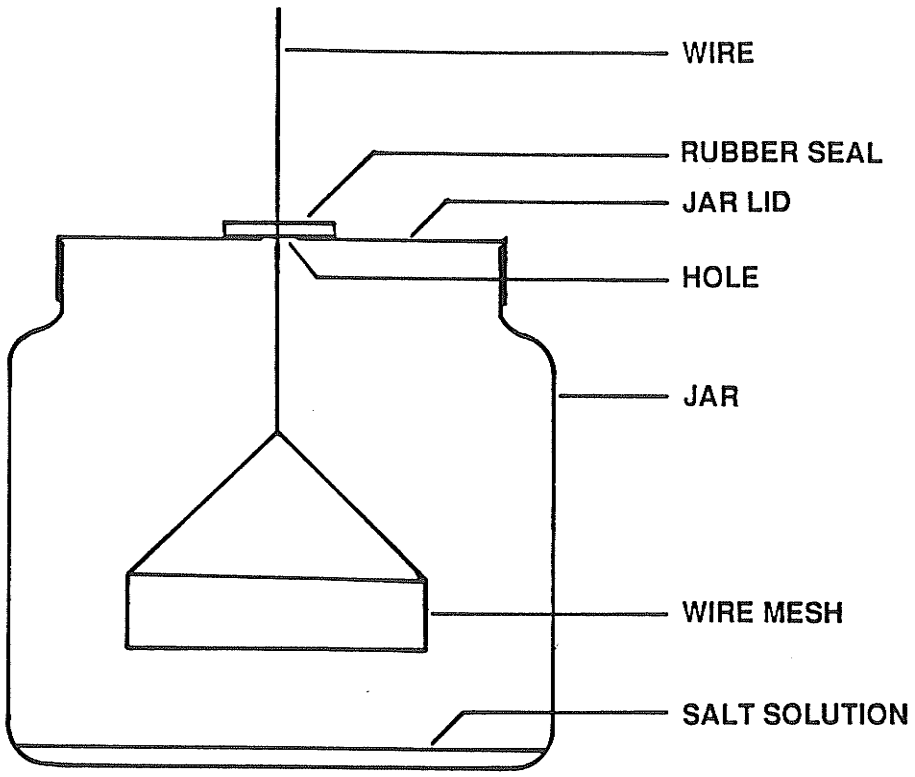


Figure 4: Desiccation chamber (adapted from Wiebe (1983)).

Analysis of the drying curves was performed by techniques adapted from Versavel and Muir (1988) where the drying behavior of wheat spikes was investigated. Tuber moisture loss was considered to be a function of the tuber's moisture content and described by

$$\frac{M - Me}{Mo - Me} = \exp(-kt)$$

where M = moisture content (%db); Me = equilibrium moisture content (%db); Mo = initial moisture content (%db); t = time (days); k = drying constant (day^{-1}) (Versavel and Muir, 1988). PROC REG was used to determine the linear drying relationship between the tuber drying rate and the tuber moisture content gradient (Freund and Littell, 1986).

4.3 Results and Discussion

4.3.1 Statistical Justification

The logistic model is described by $y=a/(1+be^{-cx})$ where *y* represents the leaf area, shoot number, tuber biomass, etc. and *x*, the DAE or independent variable. The parameters a, b and c are estimates that determine the shape of the curve. Useful values that can be obtained from this model are: ac/4, the slope at the inflection point or the maximum absolute growth rate (AGR_{max}); *ln* b/c, the value of *x* at the inflection point; a, the asymptote and a/(1+b), the *y* intercept. The curve is unique in having symmetrical upper and lower portions around the inflection point.

Yellow nutsedge displays indeterminate growth but approaches an asymptote in response to cooling temperatures in fall. Therefore, the logistic model is appropriate based on the quality of fit and the biological realism of the curve.

In some cases, where the fit of the logistic model was poor, an exponential model, $y=ae^{cx}$, was required where y is the dependent variable, x the independent variable, and c the rate of exponential increase. This model has neither an asymptote nor an inflection point (Hunt, 1982). The exponential curve is an early component of many growth curves, however, it is impossible for a growing and differentiating organ to maintain an exponential increase. Consequently, the use of an exponential model alone for growth analysis indicates that the growth process under investigation may be in its early stages. Prolonging the investigation of the growth process would result in a shift towards a more sigmoidal response.

As standard error of a parameter estimate increased confidence in that parameter estimate decreased. If the standard error was half or less of the parameter estimate it was considered acceptable (Koutsoyiannis, 1977). The coefficient of determination (R^2), was calculated for all nonlinear regressions as described by Kvalseth (1985).

4.3.2 Growth Analysis - Pots

In most cases the shoots of both the MB and ONT ecotypes emerged within one week of planting. The number of shoots produced per pot was similar in the two years (Figure 5; Table 1). In 1989 the plants of both ecotypes produced more tubers than in 1988 and the mass of tubers was more than two times greater in

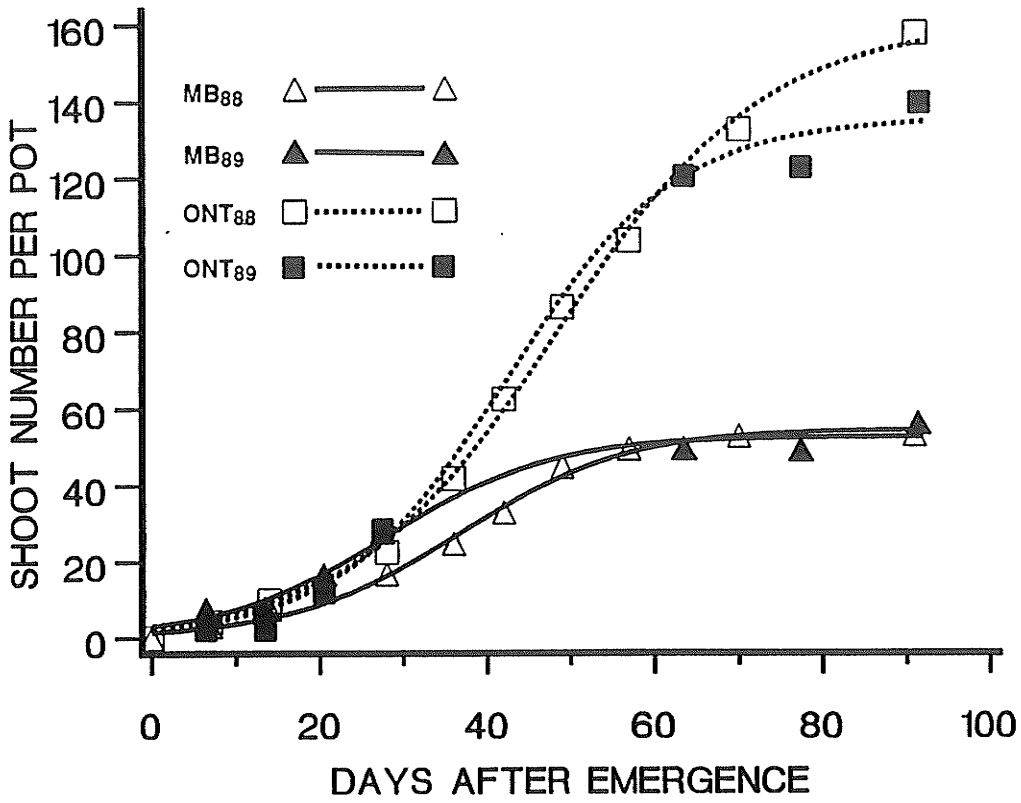


Figure 5: Increase in pot grown yellow nutsedge shoot numbers during a single growing season. Mean values from counts and the logistic function fitted to the primary data are plotted for each ecotype.

Table 1: Logistic parameter estimates (standard error in parentheses) for pot grown yellow nutsedge shoot number production.

Treatment	a	b	c	R^2	(AGR_{max}) ac/4	(DAE) ln b/c
SHOOT NUMBER (Figure 5)						
	(no.)				(no. day-1)	
MB_{88}	54.59(2.28)	38.31(18.39)	0.099(0.014)	0.83	1.36	36.57
MB_{89}	52.61(2.65)	16.92(11.63)	0.102(0.032)	0.82	1.34	27.73
ONT_{88}	160.41(4.70)	49.65(11.18)	0.080(0.010)	0.95	3.21	48.81
ONT_{89}	135.85(3.25)	64.16(18.24)	0.098(0.009)	0.98	3.33	42.46

AGR_{max} = maximum absolute growth rate, DAE = days after emergence.

1989 (Table 2). Differences in yellow nutsedge growth between years was as great as the differences between the two ecotypes. The differences in growth are probably related to differences in temperature since both soil type and watering regimes remained similar in the two years. In 1988, mean monthly temperatures were 3 C warmer than the 30 year mean and 2 C warmer than the mean monthly temperature for 1989 (Figure 3). Based on its geographical distribution, yellow nutsedge should grow better under warmer temperatures (Bendixen, 1987). The large difference in tuber production between the two years cannot be ascribed to the small difference in temperature between 1988 and 1989 and therefore remain inexplicable.

ONT produced three times more shoots than MB at the final harvest date in both 1988 and 1989 (Table 2). Conversely, MB plants consistently produced approximately twice as many tubers as ONT (Table 2). While the data are not presented, other differences included greater final leaf area and leaf number for the ONT ecotype as compared to the MB ecotype.

Because of the ecotypic differences observed in the pot experiments, both ecotypes were grown in the field in 1989 for further comparisons. Growing plants in pots provides useful comparative information between ecotypes (Williams, 1982), but field grown plants provide more realistic values. This is particularly important for yellow nutsedge since it is a perennial plant with an indeterminate growth habit.

Analysis of the drying curves was performed by techniques adapted from Versavel and Muir (1988) where the drying behavior of wheat spikes was investigated. Tuber moisture loss was considered to be a function of the tuber's moisture content and described by

$$\frac{M - Me}{Mo - Me} = \exp(-kt)$$

where M = moisture content (%db); Me = equilibrium moisture content (%db); Mo = initial moisture content (%db); t = time (days); k = drying constant (day^{-1}) (Versavel and Muir, 1988). PROC REG was used to determine the linear drying relationship between the tuber drying rate and the tuber moisture content gradient (Freund and Littell, 1986).

4.3 Results and Discussion

4.3.1 Statistical Justification

The logistic model is described by $y=a/(1+be^{-cx})$ where *y* represents the leaf area, shoot number, tuber biomass, etc. and *x*, the DAE or independent variable. The parameters a, b and c are estimates that determine the shape of the curve. Useful values that can be obtained from this model are: ac/4, the slope at the inflection point or the maximum absolute growth rate (AGR_{max}); *ln* b/c, the value of *x* at the inflection point; a, the asymptote and a/(1+b), the *y* intercept. The curve is unique in having symmetrical upper and lower portions around the inflection point.

Table 2: Comparison of shoot numbers and tuber production of two yellow nutsedge ecotypes grown in outdoor pots.

	Shoot Number	Tuber Number	Tuber Dry Weight (g)
MB_{88}	153.1a	448.2b	26.96b
MB_{89}	56.6a	734.7a	49.49a
ONT_{88}	157.9b	273.4c	11.40c
ONT_{89}	141.2b	414.8b	21.98b

1 Values presented are the means from the final sampling date (September 4, 1988, September 13, 1989). Tubers were planted on June 7, 1988 and June 8, 1989.

4.3.3 Growth Analysis - Field

4.3.3.1 Seasonal Differences

Primary rhizomes began emerging from tubers during the second week of June in both years. Differences in MB shoot dry weight, leaf area, rhizome length and tuber production were ascribed to different growing conditions in the two years. The summer of 1988 was unusually dry and hot, whereas in 1989 precipitation was above average and temperatures were normal (Figure 3).

Despite the similarity in shoot numbers in the two years for MB (Figure 6, Table 3), shoot dry matter production was significantly different, with 181 and 290 grams per plant in 1988 and 1989, respectively. A two-fold difference in nutsedge shoot dry matter was also reflected in the leaf area measurements. By the end of the growing season the leaf area of MB in 1988 was about half that in 1989. In the first year the plants produced 2.18 m^2 leaf area compared to 3.98

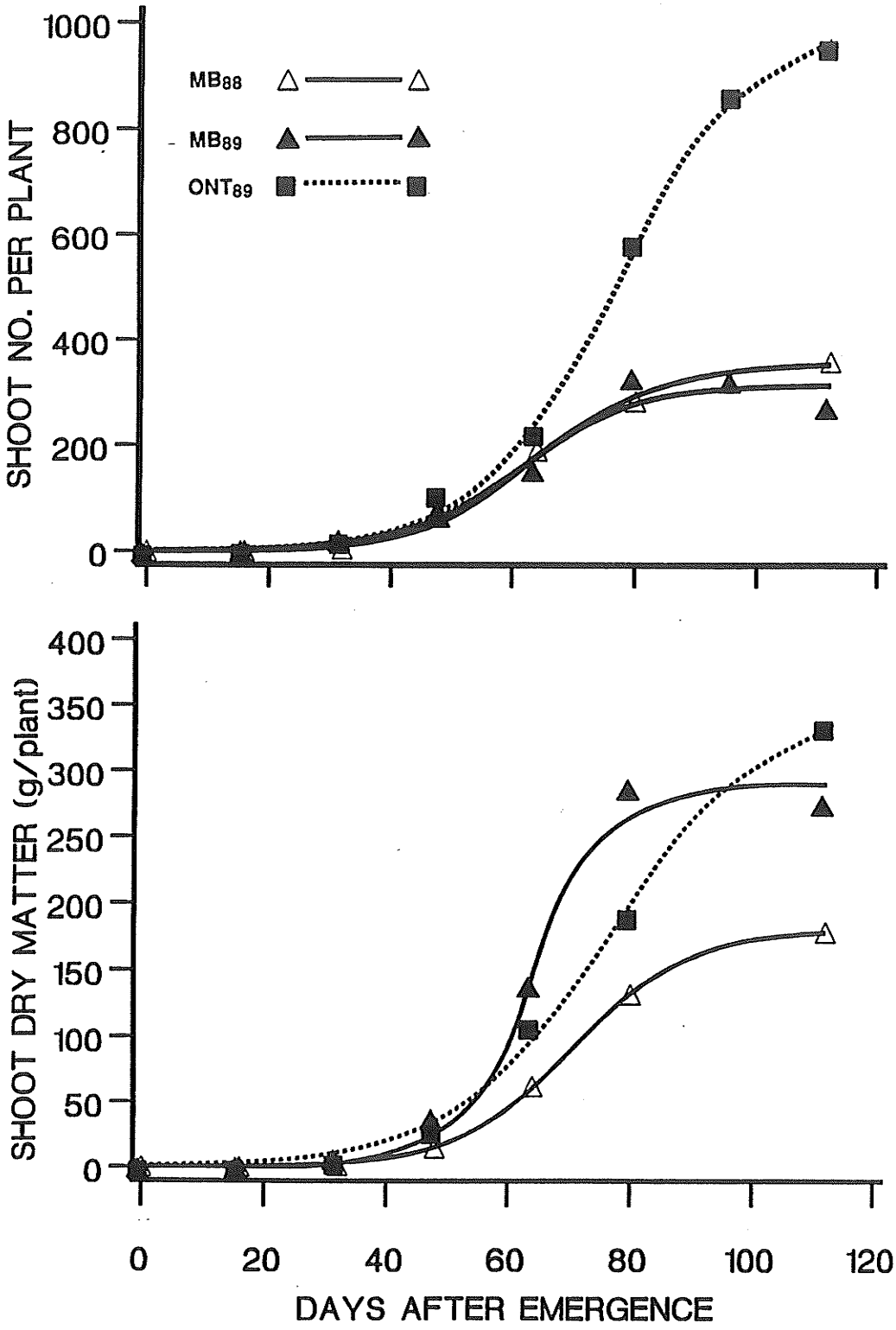


Figure 6: Increase in yellow nutsedge shoot numbers and accumulation in shoot dry matter during a single growing season. Mean values from each sampling date and the logistic function fitted to the primary data are plotted for each ecotype.

Table 3: Logistic parameter estimates (standard error in parentheses) for yellow nutsedge shoot number production and shoot dry matter accumulation.

Treatment	a	b	c	R^2	(AGR_{max}) ac/4	(DAE) ln b/c
Shoot Number (Figure 6)						
	(no.)				(no day^{-1})	
MB_{88}	357.2(26.3)	414(539)	0.095(0.022)	0.81	8.48	63.43
MB_{89}	314.9(19.5)	1070(1707)	0.113(0.027)	0.83	8.89	61.73
ONT_{89}	1013.8(69.1)	880(846)	0.088(0.014)	0.91	22.30	77.04
Shoot Dry Weight (Figure 6)						
	(g)				(g day^{-1})	
MB_{88}	180.9(12.6)	1725(2692)	0.106(0.02)	0.83	4.79	70.31
MB_{89}	290.5(10.9)	1173(12071)	0.147(0.02)	0.95	10.68	63.74
ONT_{89}	355.1(21.5)	355(239)	0.077(0.01)	0.93	6.84	76.29

AGR_{max} - maximum absolute growth rate; DAE - days after emergence

m^2 in 1989 (Figure 7; Table 4). Therefore, nutsedge plants grown in 1989 had larger leaves and more shoots than those grown in 1988, primarily the result of higher precipitation received in June of 1989.

Differences in subterranean growth also occurred in the two years. In 1988 yellow nutsedge plants produced 300 meters of rhizomes compared with 644 meters in 1989. This difference in rhizome length did not correlate with increased clonal spread since MB_{88} covered 78% of the area covered by MB_{89}. In 1989 the number of tubers produced was double the number in 1988 with 3 400 and 1 500 tubers per plant, respectively (Figure 8). Therefore, the increase in rhizome length in 1989 verses 1988 is attributable to an increase in rhizomes that differentiated to form tubers rather than rhizomes that differentiated to form shoots.

In 1989 the onset of tuberization began approximately 2 weeks earlier than in 1988 (Figure 8). A logistic model provided the best fit (R^2 = 0.87) to the 1989 data, whereas an exponential model provided the best fit (R^2=0.95) for the 1988 data (Table 5). In 1989 tuber production was sigmoidal and was twice the magnitude of tuber production in 1988. Tuber production in 1988 remained exponential.

The lifecycle of nutsedge in Manitoba (Figure 9) is compressed when compared with nutsedge lifecycle diagrams from other locations (van Groenendael and Habekotte, 1988; Stoller, 1981). In Manitoba, tubers sprout in late May and begin forming towards the end of July, as compared to Illinois where tubers sprout from the beginning of May and tuber formation begins at the beginning of August (Stoller, 1981). The entire nutsedge lifecycle in Manitoba is completed in

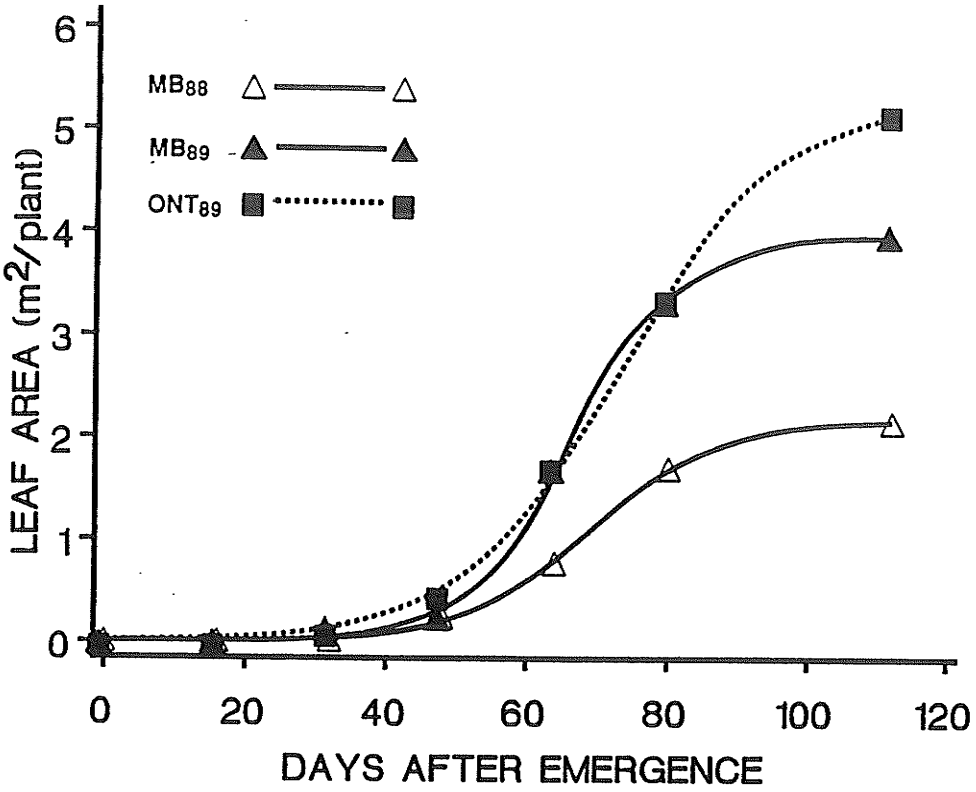


Figure 7: Accumulation of yellow nutsedge leaf area during a single growing season. Mean values from each sampling date and the logistic function fitted to the primary data are plotted for each ecotype.

Table 4: Logistic parameter estimates (standard error in parentheses) for yellow nutsedge leaf area.

Treatment	a	b	c	R^2	(AGR_{max}) ac/4	(DAE) ln b/c
Leaf Area (Figure 7)						
	(m^2)				(m^2 day^{-1})	
MB_{88}	2.176(0.160)	2019(3518)	0.110(0.027)	0.80	0.06	69.2
MB_{89}	3.979(0.075)	6135(118814)	0.131(0.058)	0.96	0.13	66.6
ONT_{89}	5.334(0.293)	621.3(501.9)	0.088(0.013)	0.92	0.12	73.1

AGR_{max} - maximum absolute growth rate; DAE - days after emergence

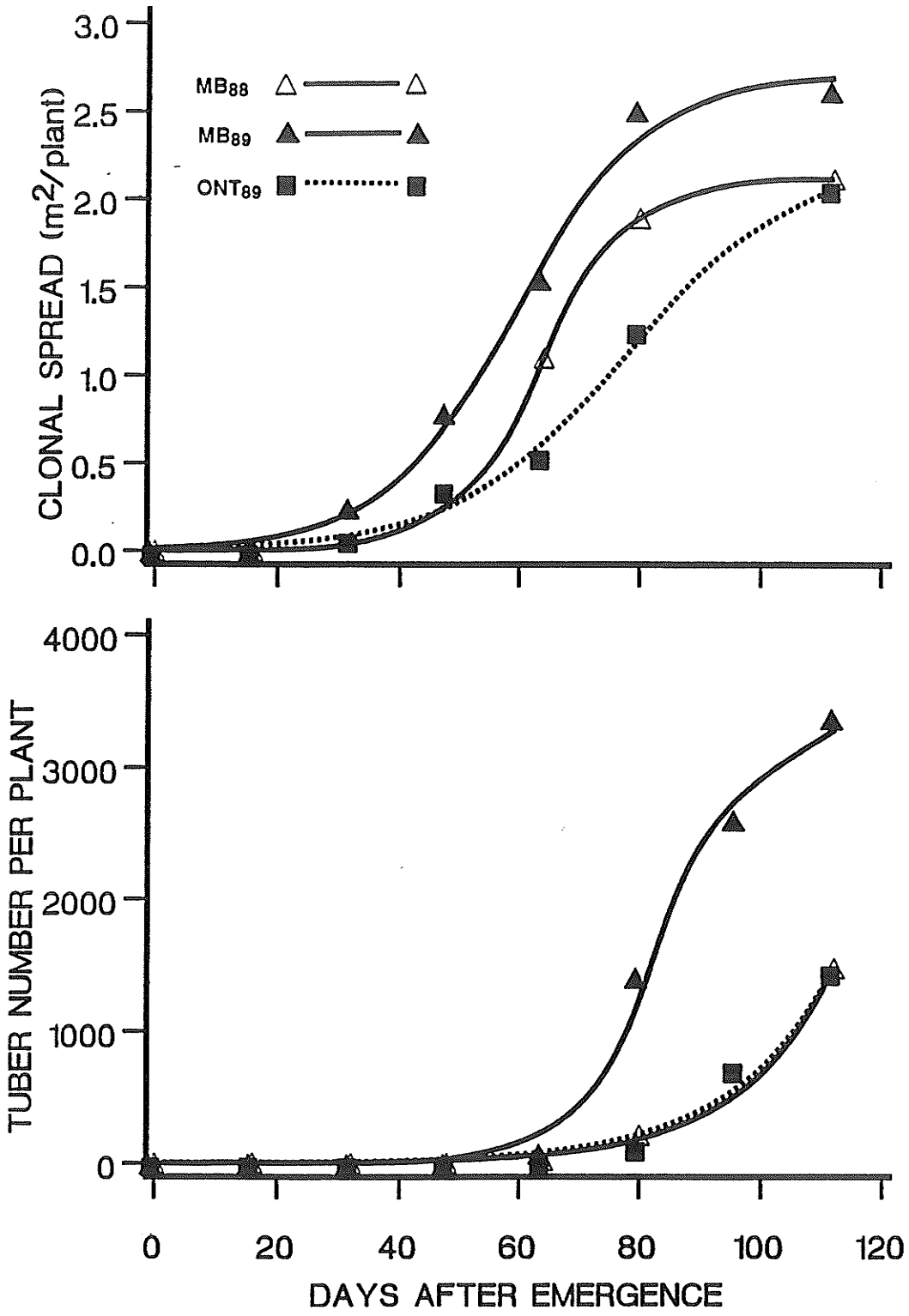


Figure 8: Clonal spread and tuber production of yellow nutsedge during a single growing season. Mean values from each sampling date and the logistic and exponential functions fitted, where appropriate, to the primary data are plotted for each ecotype.

Table 5: Logistic parameter estimates (standard error in parentheses) for yellow nutsedge clonal spread and tuber production.

Treatment	a	b	c	R^2	(AGR_{max}) ac/4	(DAE) ln b/c
Clonal Spread (Figure 8)						
	(m^2)				(m^2 day^{-1})	
MB_{88}	1.735(0.121)	1619(2492)	0.105(0.023)	0.83	0.046	70.38
MB_{89}	2.334(0.103)	4081(5030)	0.129(0.020)	0.93	0.075	64.45
ONT_{89}	3.540(0.213)	350(231)	0.076(0.010)	0.93	0.067	77.08
Tuber Number (Figure 8)						
	(no.)				(no. day^{-1})	
$^1MB_{88}$	1.842(0.775)	-	0.060(0.004)	0.95	-	-
MB_{89}	3395(280)	44195(104496)	0.127(0.030)	0.87	107.8	84.2
$^1ONT_{89}$	0.985(1.065)	-	0.065(0.01)	0.89	-	-

[1] Exponential model parameter estimates

AGR_{max} - maximum absolute growth rate; DAE - days after emergence

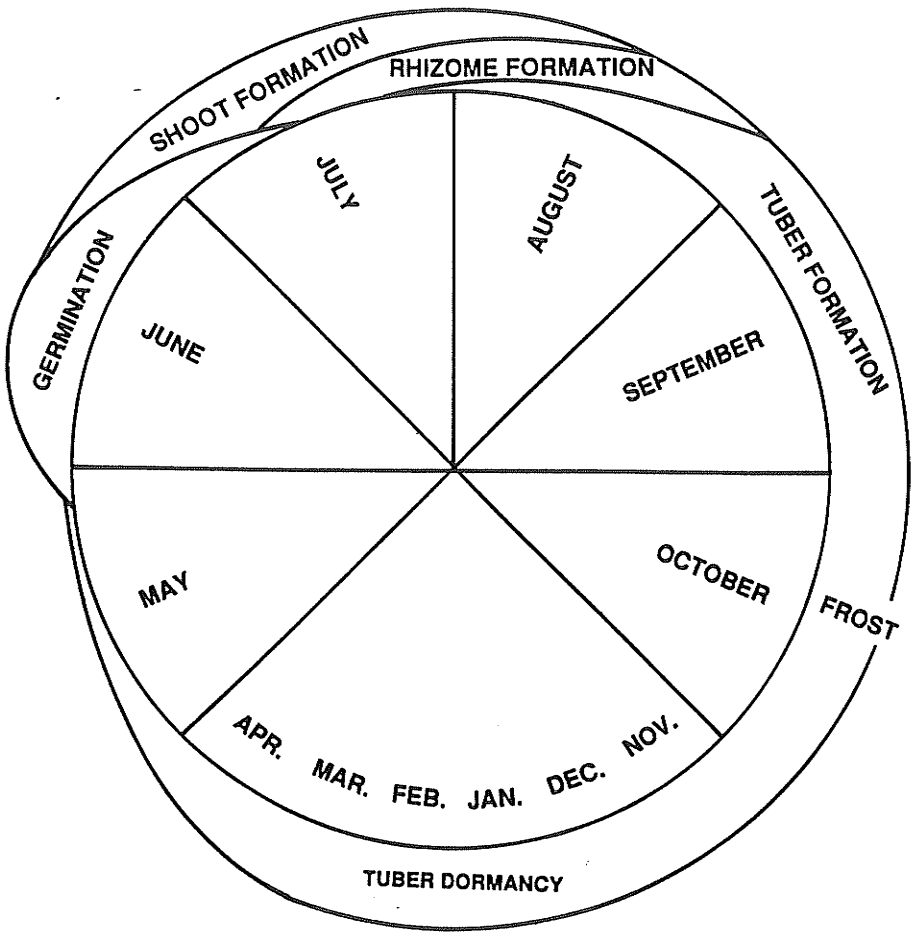


Figure 9: Life cycle of yellow nutsedge under Manitoba growing conditions. The relative amount of biomass accumulation of each growth component is reflected by the width of the outside band. (adapted from van Groenendael and Habekotte (1988) and Stoller (1981))

a four month period with September and October providing time for more tuber formation, tuber maturation and winter hardening. Adaptation to a shorter growing season is crucial to the survival of yellow nutsedge in Manitoba.

4.3.3.2 Ecotypic Differences

Based on the similarity of the spikelets and their orientation, the two ecotypes were determined to be the same variety of yellow nutsedge, *Cyperus esculentus* var. *esculentus*. Aside from these taxonomic similarities, inflorescence production was quite different between ecotypes. MB produced over 59 inflorescences per plant in a single growing season compared with only 1.6 for ONT. ONT inflorescences did not extend out of the plant in the same way as those from MB. The leaves of ONT plants were less prone to breakage and therefore sheltered the inflorescence, giving the plants a more bunched appearance (Figure 10).

In the field, the growth of ONT was very different from that of MB. This was expected based on the preliminary observations made from the yellow nutsedge growing in pots in 1988. Similar to the ecotypic differences observed in the pot experiments, ONT produced three times more shoots than MB, with predicted values of 970 and 314 shoots per plant, respectively (Figure 6; Table 3). In addition, the AGR_{max} for ONT shoot numbers occurred 2 weeks later than for MB. Despite these differences in shoot number, MB had larger shoots than ONT, accounting for the similar accumulation in shoot dry matter between the two ecotypes with asymptotes of 290 and 355 grams per plant, respectively (Figure 6; Table 3). The leaf areas for the two ecotypes were similar until approximately 80 DAE when MB approached an asymptote and ONT continued to increase such

Figure 10: Individual yellow nutsedge plants on September 14, 1989, 96 DAE; **A**) Manitoba ecotype **B**) Ontario ecotype.

that by final harvest ONT leaf area was 5.16 m^2, fully 1.2 m^2 greater than MB (Figure 7; Table 4).

Ecotypic differences were also observed in rhizome and tuber production. The ONT plants produced 920 meters of rhizomes compared to 644 meters by MB. In contrast, clonal spread of ONT was limited to 2.3 m^2 by the end of the growing season compared to 2.7 m^2 for MB. Greater rhizome length was not related to increased clonal spread but rather to increased shoot production by higher order rhizomes that did not spread far from the parent shoot. Additionally, AGR_{max} for the clonal spread of ONT occurred nearly 20 days after that of MB (Figure 8; Table 5).

Tuber production, ie., reproductive potential, differed significantly between the two ecotypes. MB produced a mean of 3 400 tubers compared to only 1 450 by ONT. ONT began forming tubers approximately two weeks after MB and followed an exponential increase. In the case of MB, tuberization was sigmoidal producing twice the magnitude of ONT tuber production near the end of the growing season. Curves describing ONT tuber production did not approach an asymptote nor did they have an inflection point.

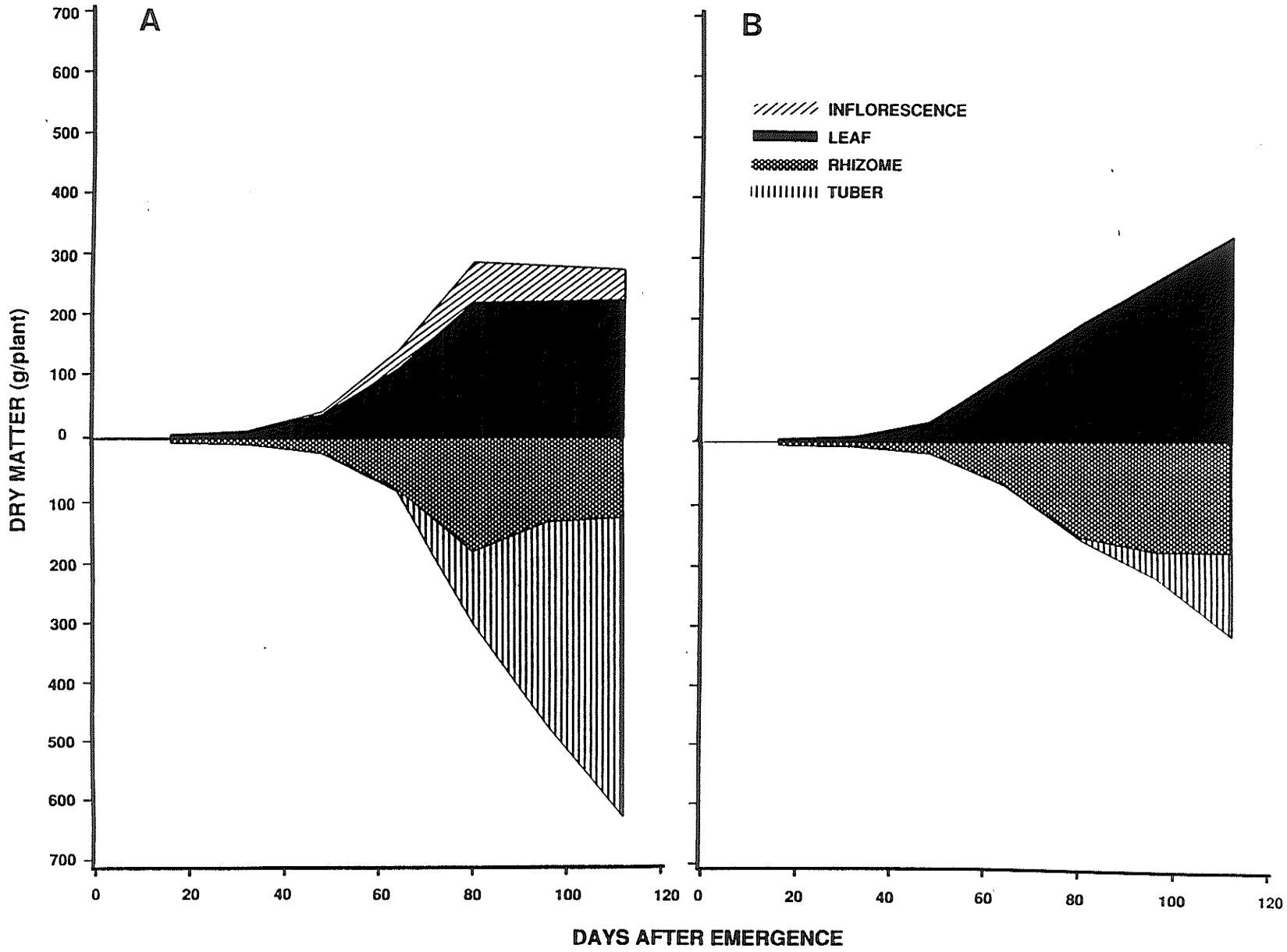
The above results show that the ONT plants require a longer growing season since the AGR_{max} for the shoot number occurred 2 weeks later than for MB as did the onset of tuber formation. The AGR_{max} for clonal spread in ONT was delayed by 3 weeks compared to that of MB. In addition, tuberization in ONT began during a photoperiod of approximately 14 hours compared with a 15.2 hour photoperiod for MB. Previous work has indicated that a 14 hour photoperiod may be critical for the onset of tuberization (Jordan-Molero and

Stoller, 1978), however, this research indicates that photoperiod may not be important since tuberization was initiated at different times over the two years. These results support those of Stoller (1983) where factors other than photoperiod such as day/night temperature variation and soil nitrogen levels were shown to be important in the initiation of tuberization.

Total biomass partitioning over the growing season was also very different for these two ecotypes. MB allocated approximately 25% of its biomass to leaf tissue compared with 50% for ONT at 112 DAE (Figure 11A and 11B). Conversely, tuber biomass constituted over 50% of the total biomass of MB compared to 21% for ONT. Williams (1982), working in Oklahoma, determined that 28% of the total yellow nutsedge biomass was allocated to tubers which was similar to the ONT results reported in this study. In total, MB produced 37% more dry matter than ONT, with most of the difference ascribed to differences in tuber formation. Clearly, under Manitoba growing conditions, MB plants are a greater weed threat than ONT plants.

4.3.4 Freezing Tolerance of Tubers

Temperature reductions were similar for both runs of the experiment with a linear decrease of approximately one degree Celsius per hour. MB was determined to be more hardy than ONT with an LT_{50} of -15.0 C compared with -7.3 C for ONT (Figure 12; Table 6). The data were fitted to a logistic model since this best described the rapid reduction of tuber viability at near-lethal temperatures. As temperature of MB was decreased from -11 to -13 C, tuber sprouting was reduced from 84.6% to 5.8%. The validity of the logistic model was further justfied by the similarity between its inflection point and the calculated LT_{50} which


A
B
INFLORESCENCE
LEAF
RHIZOME
TUBER
DRY MATTER (g/plant)
700
600
500
400
300
200
100
0
100
200
300
400
500
600
700
0
20
40
60
80
100
120
DAYS AFTER EMERGENCE

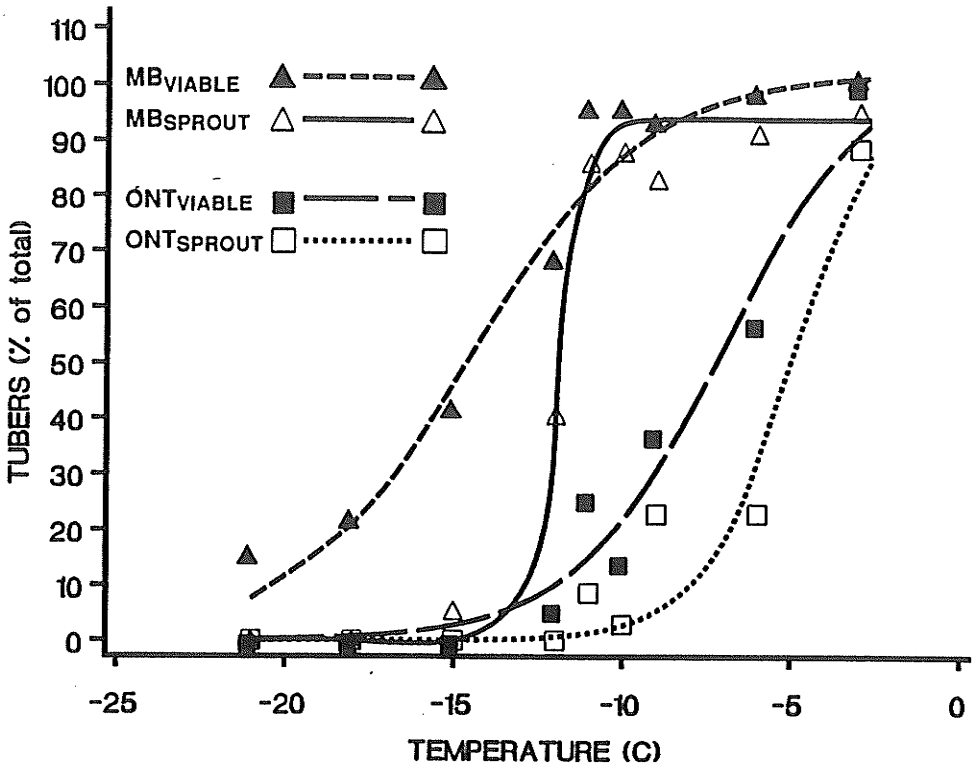


Figure 12: Effect of freezing temperatures on yellow nutsedge tuber viability. Sprouted tubers are those that sprouted during a 30 day incubation period. Viable tubers did not sprout during incubation but they did not rot suggesting that they are dormant.

Table 6: Logistic parameter estimates (standard error in parentheses) for yellow nutsedge tuber freezing tolerance.

Treatment	a	b	c	R^2	ac/4	ln b/c
Tuber Freezing Tolerance (Figure 12)						
	(%)				(% C-1)	(Temp.)
MB_{SPROUT}	93.9(2.1)	1.7E-13(N/A)	2.470(1.191)	0.87	57.96	-11.89
MB_{VIABLE}	102.3(2.3)	0.003(0.002)	0.390(0.041)	0.84	9.97	-14.57
ONT_{SPROUT}	104.8(6.4)	0.033(0.033)	0.703(0.169)	0.88	18.42	-4.85
ONT_{VIABLE}	105.9(6.2)	0.042(0.033)	0.451(0.082)	0.88	11.95	-7.05

Sprout - those tubers that sprouted during the incubation period.

Viable - those tubers that did not sprout and did not decay during the incubation period and were presumed dormant.

indicates symmetry of the curve around the 50th percentile. A sigmoidal response was also observed in similar work conducted by Stoller and Wax (1973) and Stoller (1973) where the critical freezing temperature for yellow nutsedge tubers in Urbana, Illinois was determined to be -6.5 to -7 C. ONT yellow nutsedge tubers also had an LT_{50} near -7 C and therefore exhibit similar cold hardiness to tubers from Illinois.

Figure 12 indicates that the temperatures required to reduce tuber sprouting by 50% are 2 to 3 C warmer than the temperatures required to reduce tuber viability by 50%. This difference may have been the result of a conditional dormancy imposed on the tubers under extreme cold temperatures. Although conditional dormancy was not confirmed with this research it is a logical explanation of the results. Conditional dormancy may be an important adaptive characteristic for tuber survival under cold temperatures.

The greater tolerance of the Manitoba ecotype to freezing temperatures reinforces the extent of the ecotypic differences reported and suggests that yellow nutsedge may continue to spread to areas as cold or colder than southern Manitoba.

4.3.5 Winter Soil Temperatures and Tuber Viability

During the winter of 1988/89 soil temperatures did not fall below -13 C at any of the three burial depths. Both at the 5 and 10 cm depths, approximately 46% of the tubers survived. Of those tubers buried 15 cm below the soil surface, 65% remained viable. These differences in tuber viability could not be proven to be

related to differences in soil temperature or CU in 1988/89 as had been hypothesized. Therefore the differences in tuber viability remain unexplained.

In the winter of 1989/90 colder soil temperatures occurred than in the previous year, probably the result of reduced snowfall and extremely cold temperatures early in the winter prior to any significant snowfall. Although attempts at regulating snow depth were unsuccessful because of the lack of snow in 1989/90, differences in soil temperature did occur. Nine of the ten bags of tubers were exposed, at least briefly, to temperatures below -15 C (Figure 13A). With an increase in the degree days below -12.5 C there was a decrease in tuber viability described by the linear relationship $y = -0.80x + 56.4$ ($R^2 = 0.80$) (Figure 13B). Tuber survival declined from 61% to 8% with increased exposure to cold temperatures. The tolerance of tubers to freezing in the field was similar to that observed in the laboratory. Although the same critical temperature of -15 C in the lab was not determined in the field, it appears that temperatures near -15 C were effective in reducing viable tuber populations in the field. Previous research has shown large discrepancies between field and laboratory freezing tolerance (Stoller and Wax, 1973) but these were not observed here..

In both years, under the most favourable (warmest) field conditions, at least 30% of the tubers did not survive the winter. From personal observation, it appears that very few tubers would survive more than two winters. Therefore, exposure to cold temperatures should be incorporated as an integral component in yellow nutsedge control programs where the winters are severe.

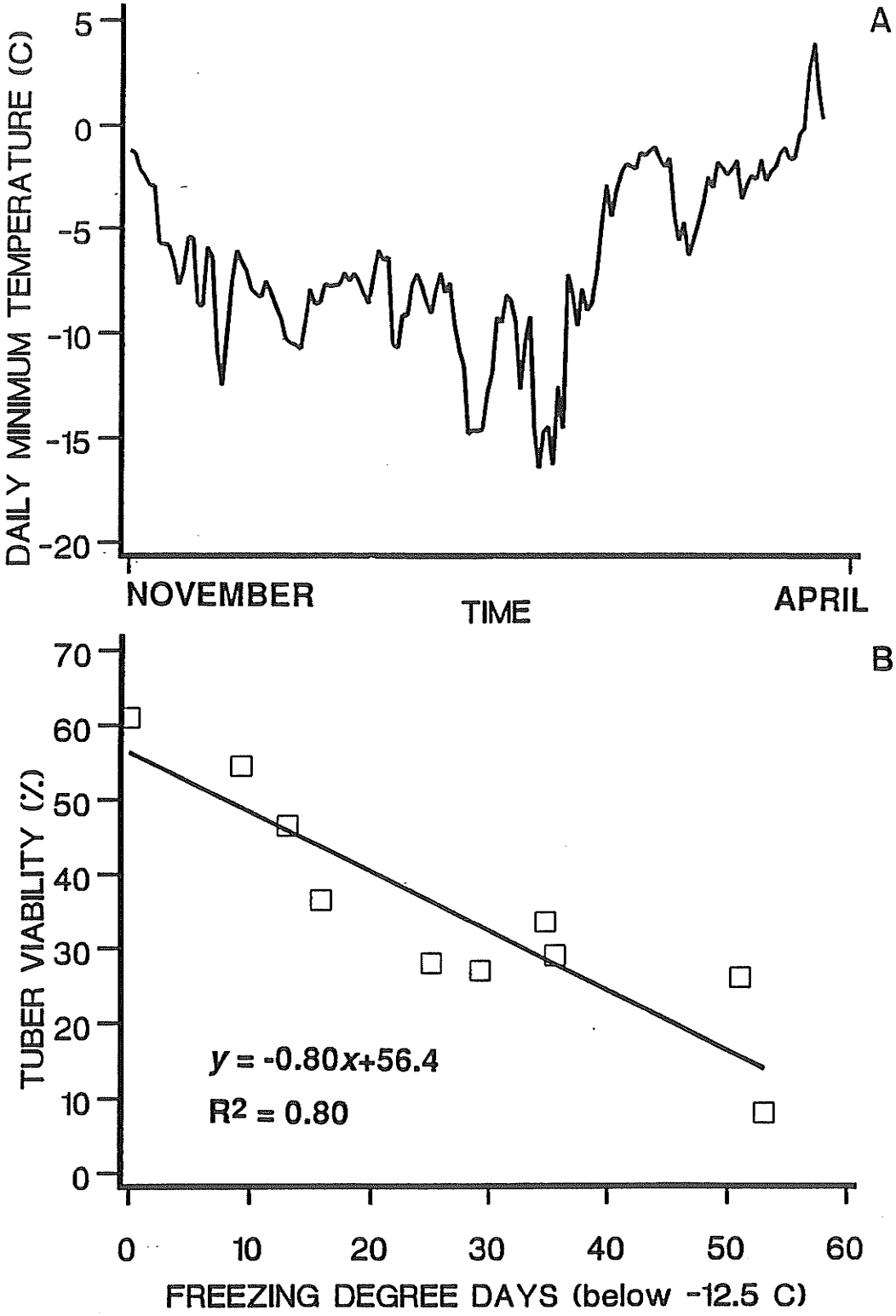


Figure 13: **A)** Daily minimum soil temperatures 10 cm below the soil surface during the 1989/90 winter. **B)** Effect of winter soil temperatures, expressed as freezing degree days below -12.5 C, on nutsedge tuber viability. Means of each datalogger channel are plotted.

4.3.6 Desiccation Tolerance of Tubers

The desiccation chambers effectively desiccated the nutsedge tubers to a range of moisture contents and at different rates (Figure 14). Although identical treatments were used in each experiment, rates of desiccation were not consistent across experiments. One possible reason for this inconsistency may be the condition of the tubers. Potato tubers are known to lose water more rapidly if the periderm is bruised (Nash, 1985). Rough handling of tubers may increase the rate of water loss from the tubers as compared to nonbruised tubers. Regardless of the basis for the inconsistencies, the greater number of desiccation rates provided a larger range of data for analysis.

Final tuber moisture content did not influence tuber viability directly since no significant relationship could be established between these parameters. Tubers desiccated to 2.5% moisture content maintained 77% viability upon rehydration (Figure 15A). Similarily, no relationship between temperature at desiccation and tuber viability could be established (data not presented) as had been previously reported by Thomas (1969).

Tuber moisture loss was curvilinear as is typical of most tissues undergoing desiccation (Figure 14). The tuber drying curves that describe tuber moisture loss as a function of the moisture gradient, for the range of moisture conditions under study were described by a linear relationship (Versavel and Muir, 1988). The slopes of these linear relationships were plotted against tuber viability to determine the influence of the rate of drying on tuber viability (Figure 15B). The rate of desiccation strongly affected tuber viability and is described by $y = -298.7x + 214.5$ ($R^2 = 0.74$). This result implies that tubers located on or

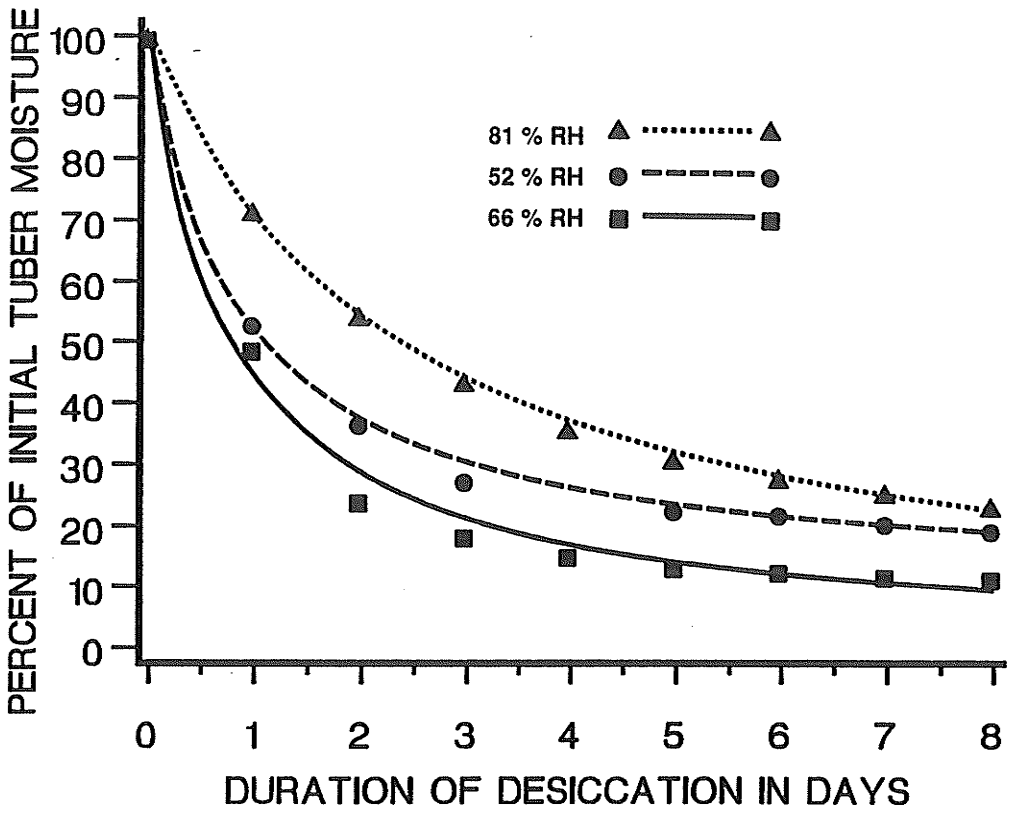


Figure 14: Rate of yellow nutsedge tuber desiccation under different relative humidities. Mean percent of original tuber moisture for each day and a curvilinear function fitted to the primary data are plotted for each treatment.

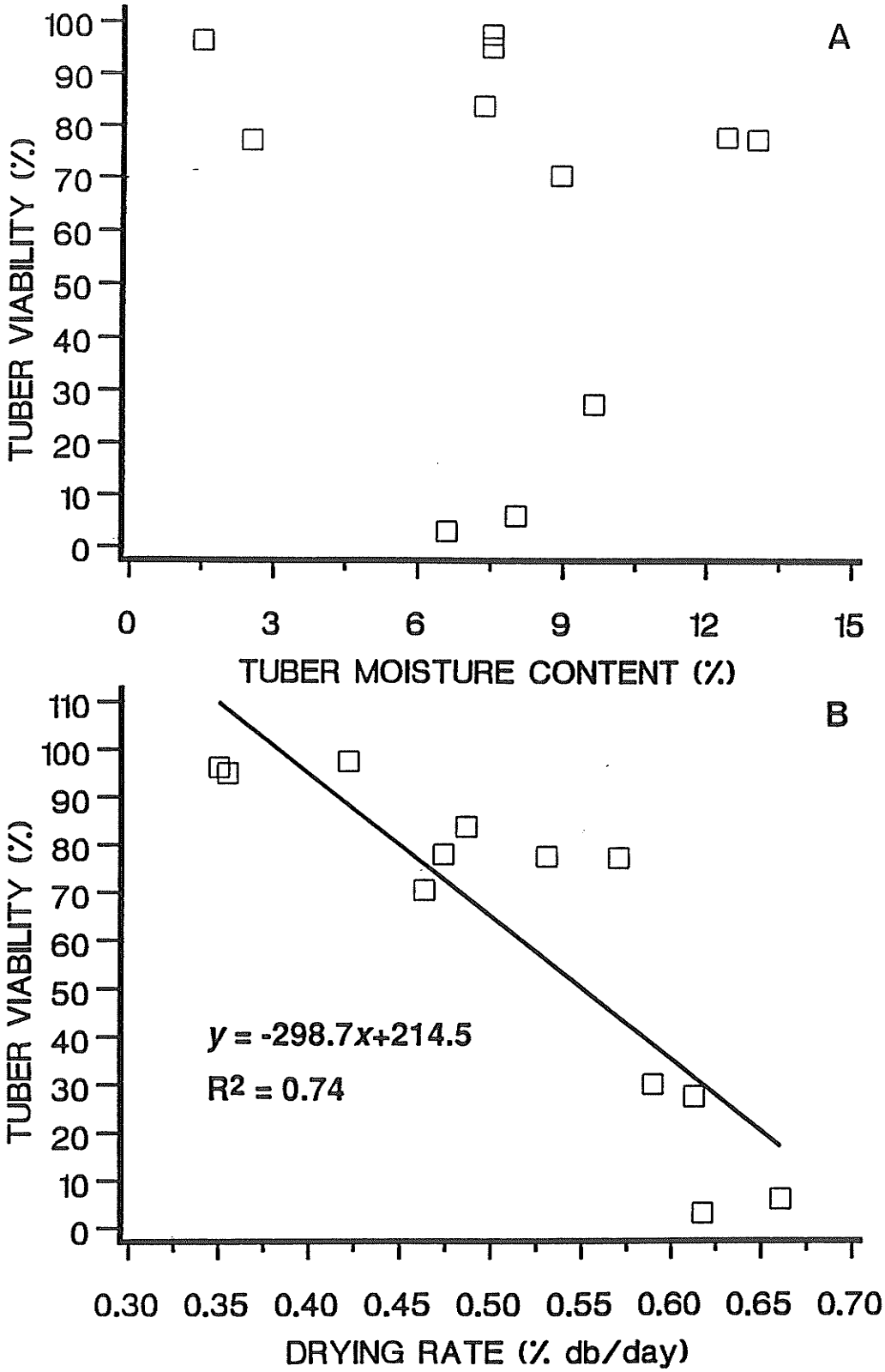


Figure 15: A) Effect of nutsedge tuber moisture content after desiccation on tuber viability. Means of each desiccation treatment are plotted. B) Effect of the nutsedge tuber drying rate on tuber viability. Means of each desiccation treatment are plotted.

near the soil surface would be susceptible to greater mortality than would tubers found deeper in the soil where soil moisture loss occurs more slowly. Since deep tillage places tubers deeper in the soil profile (McDonald, 1986) it would not be as effective as shallow tillage in facilitating tuber desiccation.

5. Crop Interference and Nutsedge Growth

5.1 Introduction

Yellow nutsedge does not possess many of the characteristics normally associated with competitive weeds. Because yellow nutsedge has a C4 photosynthetic pathway, it grows rapidly under high light intensities and temperatures (Keeley and Thullen, 1978). Since yellow nutsedge is sensitive to shading, crops that grow taller than the weed have a competitive advantage (Stoller, 1981).

Yield losses due to competition from yellow nutsedge do occur when the crop is slow to emerge or emerges later than the weed. Cotton yields in Alabama were reduced by 41% if yellow nutsedge competition lasted for the entire growing season with an average nutsedge density averaging 60 shoots m^{-2} (Patterson et al., 1980).

The major objective of all yellow nutsedge control practices, including growing of competitive crops, is to reduce or prevent the production of tubers. Tubers are the primary reproductive propagules and are responsible for the perennial growth habit of this weed (Keeley and Thullen, 1978). Shading effectively reduces both the growth and production of tubers in yellow nutsedge. Shading nutsedge plants caused a shift in biomass partitioning from the tubers to the leaves (Patterson, 1982). Additionally, Patterson (1982) determined that shaded nutsedge plants had the same area per leaf as non-shaded nutsedge but dry matter per leaf was lower and fewer leaves were formed per plant. In California, reducing incident radiation by 30 and 80% reduced tuber production by 32 and 80%, respectively (Keeley and Thullen, 1978). Providing 80 and 94% shade to

nutsedge plants for a 3 month period limited tuber production to 381 and 51 tubers per plot, respectively, (Keeley and Thullen, 1978) indicating that even very dense shade does not prevent tuber production entirely (Jordan-Molero and Stoller, 1978; Patterson, 1982). These studies emphasize the importance of good crop management in impeding nutsedge growth.

In Manitoba, no successful yellow nutsedge control measures have been established and many control practices in use elsewhere relate to cropping practices that are not common in Manitoba. There are no selective herbicides that effectively control yellow nutsedge in crops commonly grown in Manitoba and therefore a greater emphasis must be placed on agronomic practices that provide effective weed control. The objectives of this research were to determine the influence of a competitive crop, grown in Manitoba, on the growth and reproductive potential of the Manitoba ecotype of yellow nutsedge.

5.2 Materials and Methods

An infestation of yellow nutsedge was established at the University of Manitoba Glenlea Research Station in the spring of 1988. The soil was a Red River Clay that was broken from an alfalfa/bromegrass hay crop in 1987. In 1989 the soil had a pH of 7.6 and nutrient levels were 92, 108, and 1 260 kg/ha of N,P, and K, respectively. Nutsedge growth in 1988 increased the tuber density from the initial planting density of 50 m^{-2} to 1 018 m^{-2} by the spring of 1989.

The infestation was initiated in a randomized complete block design that would facilitate eight replications of three treatments. In 1989 treatments were established and included nutsedge growing alone, nutsedge growing together

with a barley crop and barley growing alone. Prior to seeding, the experimental area was fertilized with 22 kg N ha^{-1} and 23 kg P ha^{-1}. The fertilizer was broadcast and incorporated with a double discer. Barley (Bedford) was sown on May 27, 1989 at 110 kg ha^{-1} to a 4 cm depth. On those plots where nutsedge was to grow alone, barley was selectively removed with fluazifop-butyl at 200 g ai ha^{-1} on June 8. Fluazifop-butyl was re-applied at 200 g ai ha^{-1} on June 18 to remove late emerging barley from those same plots. There was no visible injury to the nutsedge plants as a result of either application of fluazifop-butyl. The experiment was hand weeded regularly and nutsedge was removed from where it had spread into the nutsedge free plots.

Nutsedge was destructively sampled from two 25x25x20 cm quadrats per plot 0, 18, 36, 54 and 72 days after emergence. All above ground plant tissue in the quadrat was removed and the area beneath the quadrats was excavated to a depth of 20 cm. Samples were separated into barley culms, nutsedge shoots, rhizomes, and tubers. The biomass of each plant component was determined after the material was dried at 80 C for 48 hours.

Photosynthetic photon flux density (PPFD) was measured with a line quantum sensor[1] at weekly intervals. Measurements were made at ground level and above the plant canopy during 1200 to 1400 hours in order to evaluate changes in incipient radiation.

Where appropriate, the primary data were fitted to the logistic model using a derivative free nonlinear regression procedure (Freund and Littell, 1986).

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[1] Line Quantum Sensor, Model LI-191SB, Li-Cor Inc., Lincoln, NE.

Otherwise PROC REG was used to define the linear relationship of the primary data as outlined by Fruend and Littell (1986). Final sampling date parameters were also compared in a table adapted from van Groenendael and Habekotte (1988) through the use of the *t* test.

5.3 Results and Discussion

Barley effectively reduced the amount of light penetrating the plant canopy at a faster rate than did nutsedge growing alone. Where nutsedge was growing with barley, 14% of available PPFD was penetrating the plant canopy at 23 DAE compared with 87% where nutsedge was growing alone. By 30 DAE only 2% of available PPFD entered the canopy in the plots containing barley and nutsedge compared with 56% for nutsedge growing alone (Figure 16). In plots containing the barley crop, there was little light available to support nutsedge growth only three weeks after emergence.

Interference from the barley crop effectively reduced growth of all measured parameters of yellow nutsedge. Initially, nutsedge shoot numbers were similar in the plots containing barley and those without, as expected based on the uniform tuber densities recovered on the first sampling date (0 DAE). Nutsedge shoot production was drastically reduced by the barley crop, culminating in final shoot densities of 302 and 1 000 shoots m^{-2} in the plots containing barley and those without, respectively (Table 7).

Differences in shoot biomass were more pronounced than differences in shoot number. In the absence of crop competition, nutsedge produced almost 12 times as much shoot dry matter as where it was competing with barley (Figure 17).

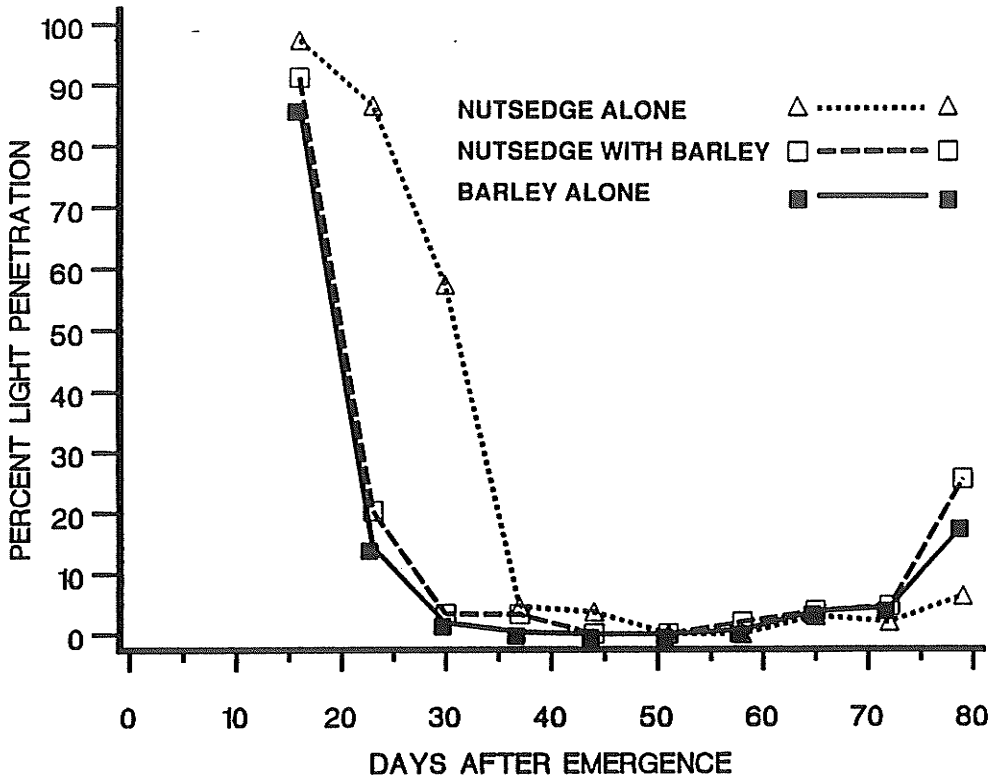


Figure 16: Percent of incipient photosynthetic photon flux density (PPFD) penetrating to the ground through the plant canopies. Mean percent penetration values are plotted and connected.

Where nutsedge was growing alone nutsedge shoot dry matter production was best described by a logistic model but in plots containing barley and nutsedge a linear model provided a better fit to the data (Figure 17, Table 8). Barley interference reduced nutsedge growth to only 0.66 grams day^{-1}. This marginal increase in nutsedge shoot biomass, compared with the large increases in nutsedge shoot biomass (17.8 g day^{-1}) observed where there was no crop, suggests that there was a strong reduction or complete lack of autotrophic growth where nutsedge was grown with barley.

The barley crop also interfered with the production of nutsedge tubers. In plots where nutsedge was growing alone, net tuber production was sigmoidal with a net increase of 3 151 tuber m^{-2}. In competition with barley, the nutsedge tuber increase was limited to 168 tubers m^{-2} (Figure 17, Table 8). This represents a 18 fold difference in reproductive potential. It is noteworthy, however, that despite the large reduction in both nutsedge shoot dry matter production and available light penetrating the plant canopy in the cropped plots, a net increase in tuber number still occurred.

Both where nutsedge was growing alone and with the barley crop, the final tuber weight was less than the initial tuber weight in the spring of 1989 (Table 7). The lower density of nutsedge in 1988 permitted the formation of larger tubers; however in 1989, barley interference resulted in the formation of smaller tubers of lower individual weight (Table 7). This reduction in tuber size is important since smaller tubers have been shown to be less cold tolerant (van Groenendael, 1988) and produce shoots with reduced vigor (Stoller et al., 1972; Stoller and Wax, 1973). Nutsedge growing alone produced 7.8 tubers per gram of shoot

Table 7: Effect of barley competition on yellow nutsedge growth and tuber production at Glenlea in 1989. Values presented are the treatment means from the final sampling date, 72 DAE. (* = significant at 5%)

Assessment	Nutsedge Shoot Density (no. m-2)	Shoot Dry Matter (g m-2)	Tuber Density (no. m-2)	Tuber Dry Matter (g m-2)	Individual Tuber Wt (g)	Net Tubers Per Gram of Shoot (no. g-1)
Initial (0 DAE)	-	-	1018	153.8	0.151	-
No crop (72 DAE)	1000	401.7	4169	417.7	0.101	7.8
Crop (72 DAE)	302	34.6	1186	100.5	0.085	4.9
	*	*	*	*	*	*

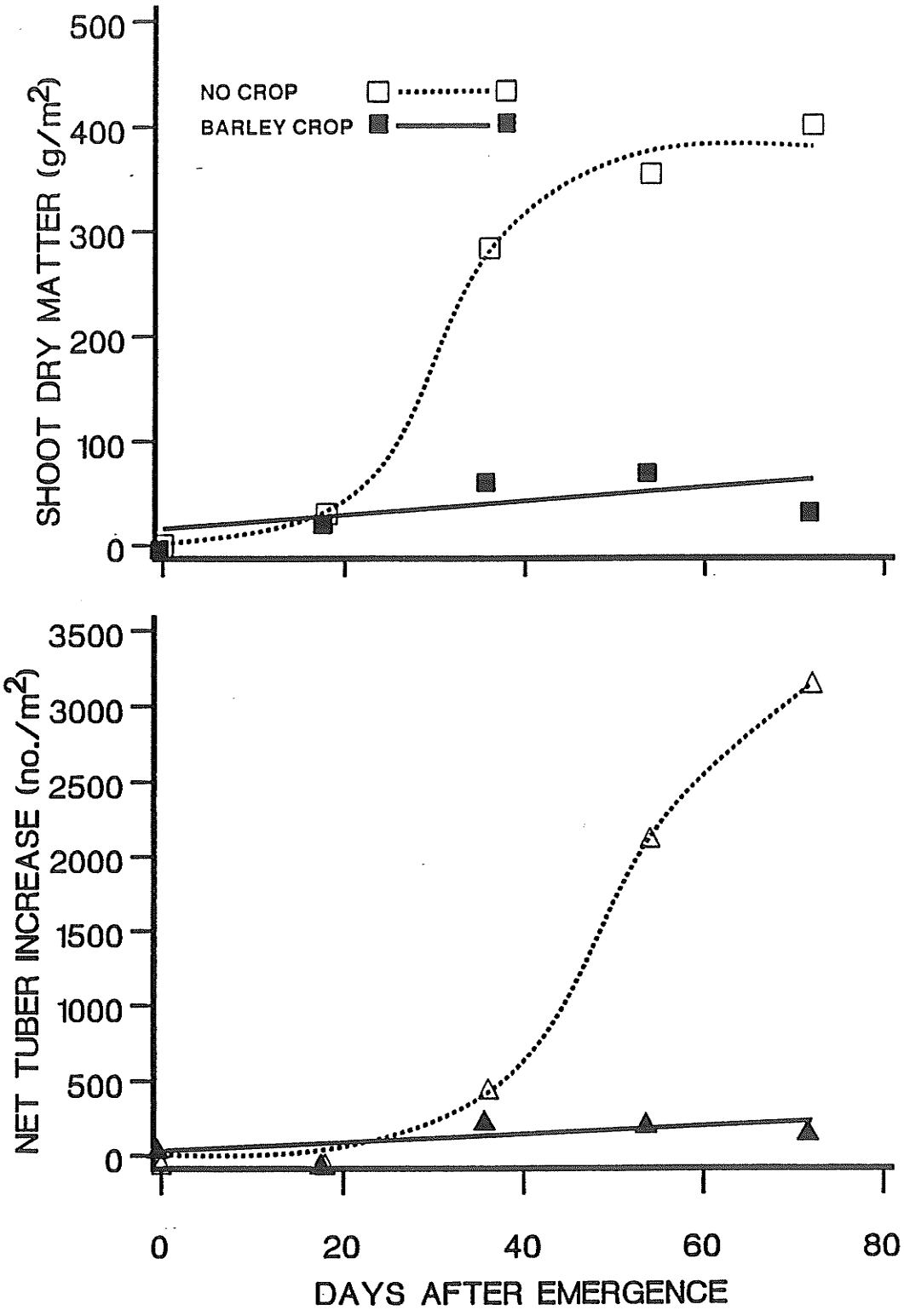


Figure 17: Effect of barley interference on yellow nutsedge shoot dry matter accumulation and tuber production.

Table 8: Logistic parameter estimates (standard error in parentheses) for yellow nutsedge shoot dry matter and tuber production with and without crop interference. (Figure 17)

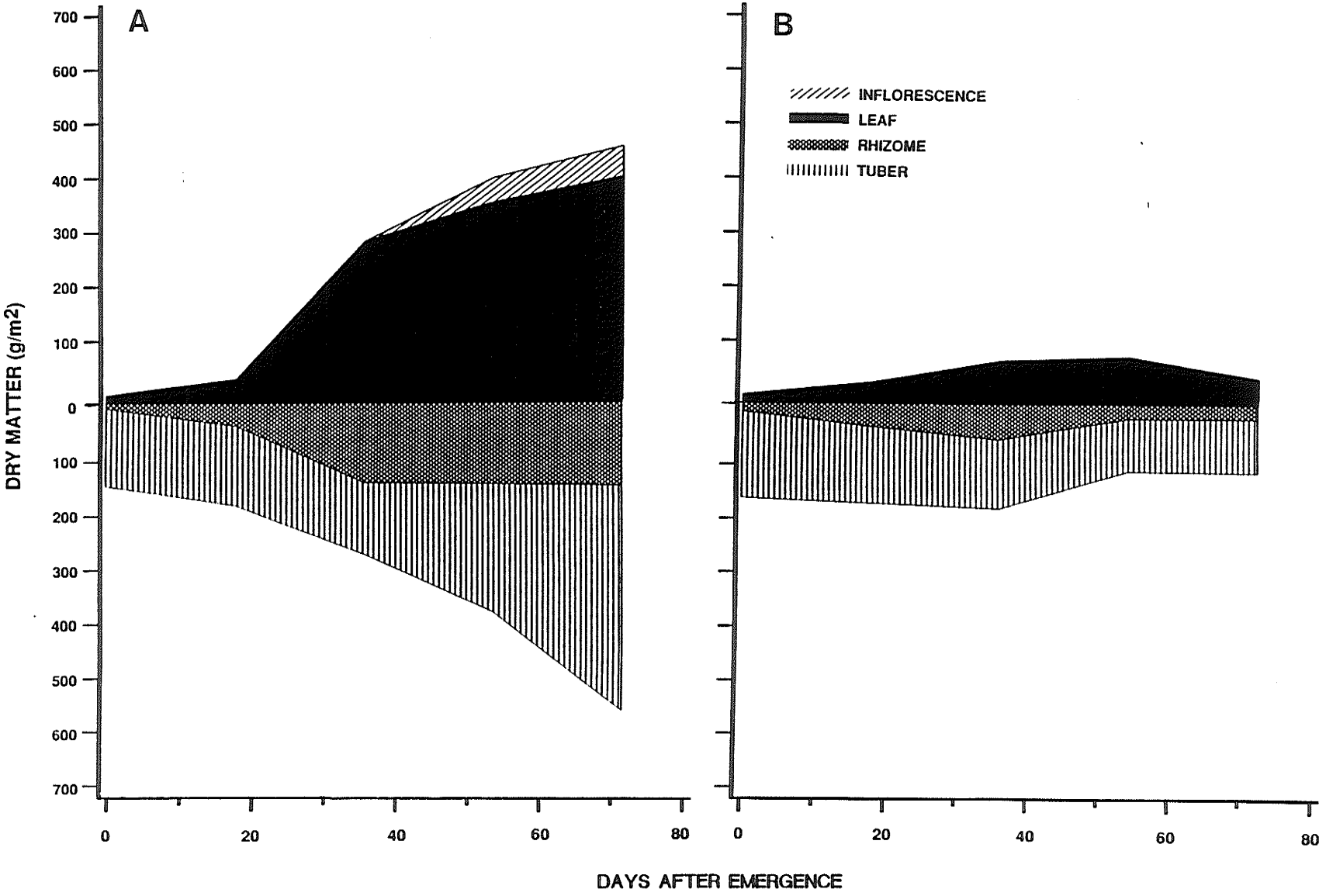
Treatment	a	b	c	R^2	(AGR_{max}) (DAE) ac/4	ln b/c
No Crop (logistic)						
Shoot	437(24.2)	201(345)	0.163(0.051)	0.86	17.81	32.54
Tuber	3285(216)	1070(1440)	0.140(0.028)	0.92	115.0	49.8

Crop (linear)	a	b	R^2
Shoot	15.5(9.57)	0.66(0.22)	0.22
Tuber	29.5(104.9)	2.72(2.38)	0.04

biomass compared with 4.9 tubers per gram of shoot biomass when grown together with barley (Table 7).

Nutsedge reduced crop dry matter production by approximatley 30% (Appendix 3). The barley affected nutsedge more than nutsedge affected barley. Where yellow nutsedge was growing with the barley, both rhizome and tuber dry matter decreased later in the season, probably the result of decay (Figure 18). In the cropped plots no nutsedge inflorescences were produced. In total, barley caused a 7-fold reduction in total nutsedge biomass production, emphasizing the importance of growing a competitive crop like barley in a yellow nutsedge control program.

The results of the interference experiments compare well with results obtained elsewhere. In California, after one year of barley followed by one year of fallow, the tuber population declined by 75% (Keeley et al., 1983). Other cropping rotations such as potato/soybean and continuous cotton included herbicide treatments and achieved similar results. Tuber numbers fell by 71 and 60% after one year and 99 and 98% after three years, respectively. Growing alfalfa continuously in California was as effective as a barley/corn rotation reducing tuber populations by 50% in the first year and by 95% of the original population after three years. On a three year basis these rotations proved to be more effective than the barley/fallow combination which had 88% fewer tubers after 3 years (Keeley et al., 1979). The common factor among all of the most effective treatments is to maintain a continuous pressure on the weed in order to effectively reduce tuber populations. Intensive crop management can strongly reduce nutsedge growth and drastically impede tuber production.


A
B
INFLORESCENCE
LEAF
RHIZOME
TUBER
DRY MATTER (g/m2)
700
600
500
400
300
200
100
0
100
200
300
400
500
600
700
0
20
40
60
80
DAYS AFTER EMERGENCE

6. Summary and Conclusions

Infestations of the pernicious weed, yellow nutsedge, have been identified in Manitoba. Tubers of yellow nutsedge moved northward with Red River floodwaters in the spring of 1979. Investigations into the geographical distribution of yellow nutsedge in Manitoba determined that at least 21 infestations now exist, all bordering the Red River. Identification and isolation of yellow nutsedge infestations is key to preventing further spread of this weed. Extension efforts have begun and must be continued to maintain awareness of this weed in high risk areas, namely fields along the Red River south of Winnipeg.

Since yellow nutsedge is an important weed in many parts of eastern Canada, tubers from a population of nutsedge in Harrow, Ontario were collected and sent to Manitoba for comparison purposes. Yellow nutsedge populations from Manitoba and Ontario were determined to be the same variety but different ecotypes. Plants from Manitoba produced twice as many tubers and one-third as many shoots as those from Ontario. From a single tuber planted in early June, 3 400 tubers were produced by the Manitoba ecotype in a single growing season.

Ontario plants appeared to require a longer growing season since they generally initiated growth processes such as tuber initiation and maximum shoot production later than Manitoba plants. As a result of these and other ecotypic differences, the Manitoba ecotype is a more prolific weed under Manitoba growing conditions. Since control is required prior to tuber formation, the Manitoba ecotype must be controlled earlier in the growing season than the Ontario ecotype. Determining

when tuber formation occurs strongly impacts the choice of weed management strategies.

An additional study was designed to compare the freezing tolerance of tubers from the two ecotypes. This comparison revealed that in addition to morphological differences, nutsedge ecotypes differed physiologically as well. Lethal temperatures for the two ecotypes were determined to be -15.0 and -7.3 C for Manitoba and Ontario, respectively. This difference in freezing tolerance reinforces the uniqueness of the two ecotypes. In the field, no Ontario tubers were seen to survive through winter confirming these differences.

Explorations into the freezing conditions in the field were critical to determine whether tubers were avoiding or tolerating the cold temperatures. Tubers from the Manitoba ecotype were stored at several soil depths through a winter and evaluated for survival. Tubers that were exposed to a greater number of degree days below -12.5 C responded with lower viability. Regardless of the soil depth, at least 30% of the tubers lost viability through the winter. Tubers of yellow nutsedge in Manitoba are capable of overwintering not by avoidance but by tolerance of the freezing temperatures. Freezing tolerance of tubers clearly indicates that yellow nutsedge can potentially survive in areas as cold or colder than southern Manitoba.

Having established the morphological characteristics and resulting potential economic importance of this weed, attention was also directed to control. Cultivation is an important component of perennial weed control programs. Cultivation exposes tissues that are normally unavailable to desiccation and starvation conditions. Tubers of yellow nutsedge were evaluated for tolerance to

desiccation in order to determine the effectiveness of tillage. The rate of desiccation was confirmed as the most important determinant of tuber viability. Rapid desiccation caused greater mortality in yellow nutsedge tubers. Tuber moisture content after desiccation was not related to tuber viability. Tubers of yellow nutsedge were very tolerant of desiccation suggesting that tubers located on or near the soil surface would be more susceptible to mortality than would tubers buried deeper in the soil.

Unfortunately, the technique used to evaluate desiccation tolerance cannot be directly related to field conditions. In order to maximize the use of this technique it must be developed further to relate to conditions in the field. This was beyond the scope of this project but should be considered in future research.

Yellow nutsedge is a very shade intolerant plant and grows poorly in a competitive environment. Barley, a crop commonly grown in Manitoba, was grown together with yellow nutsedge to evaluate crop competition on nutsedge growth. Within three weeks after emergence light penetration in the plots where nutsedge was growing with barley was limited to 14% of the incident radiation compared with 87% where nutsedge was growing alone. Nutsedge growth reflected these large differences in light penetration. Where nutsedge was growing alone, nutsedge accumulated seven times more biomass than where there was crop. Similarily, where nutsedge was grown in the absence of barley, 18 fold more tubers were produced than where there was barley. Despite the influence of the barley crop on nutsedge growth and tuber production, intensive crop competition did not prevent a marginal net increase in tuber production. Crop competition can and should be used in a yellow nutsedge control program. This control practice requires further development to maximize its potential.

Unfortunately, light penetration was the only component of crop interference that was measured. It would have been useful to measure water and nutrient availability in each plot and the influence of different seeding densities of both crop and weed (replacement series design). Determining which component of interference most effectively reduced nutsedge growth would assist in selection of other competitive crops.

These investigations have provided a good understanding of how nutsedge plants grow and are able to survive under Manitoba growing conditions. Eradication of this weed will be difficult but possible with continued attention. The harsh climate of Manitoba can contribute to the control of this weed if good management is used. Provided that the weed does not spread, it should not become a major agronomic weed in Manitoba but rather a remain a serious local concern.

Bibliography

1. **Bayer, D.E.** 1987. Tuber dormancy, germination, apical dominance, and translocation in yellow and purple nutsedge. Proceedings of the California Weed Control Conference. 39:90-92.

2. **Bendixen, L.E.** 1973. Anatomy and sprouting of yellow nutsedge tubers. Weed Science. 21:501-503.

3. **Bendixen, L.E.,** and **U.B. Nandihalli.** 1987. Worldwide distribution of purple and yellow nutsedge (*Cyperus rotundus* and *C. esculentus*). Weed Technology. 1:61-65.

4. **Boldt, P.F., C.P. Yip, R.D. Sweet.** 1976. Preliminary studies with five New York yellow nutsedge selections. Proceedings of the Northeastern Weed Science Society. 30:126-131.

5. **Borg, S.J.T., L.J.D. Nijs, H.V. Oene.** 1988. Intraspecific variation of *Cyperus esculentus* L. in the Netherlands; A preliminary report. VIIIeme Colloque International Sur La Biologie L'ecologie Et La Systematique Des Mauvaises Herbes 8:181-185.

6. **Causton, D.R.,** and **J.C. Venus.** 1981. The Biometry of Growth. Edward Arnold Publishers Ltd. London. 307pp.

7. **Cleveland, W.S.,** and **I.J. Terpenning.** 1982. Graphical methods for seasonal adjustment. J. Am. Stat. Assoc. 77:52-62.

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

9. **Cousens, R.** 1985. A simple model relating yield loss to weed density. Annals of Applied Biology. 107:239-252.

10. **Darbyshire, S.J.** 1989. Personnel Communication. Agriculture Canada. Ottawa, Ontario.

11. **Day, B.E.,** and **R.C. Russel.** 1955. The effect of drying on survival of nutgrass tubers. California Agricultural Experimental Station Bulletin 751.

12. **Elias, C.O.,** and **D.R. Causton.** 1976. Studies on data variability and the use of polynomials to describe plant growth. New Phytologist. 77:421-430.

13. **Fischer, B.B.** 1987. Yellow nutsedge control in orchards and vineyards. Proceedings of the California Weed Control Conference. 39:184-194.

14. **Freund, R.J.,** and **R.C. Littell.** 1986. SAS System for Regression. Cary, NC. pp 133-150.

15. **Friesen, G.H.,** and **A.S. Hamill.** 1977. Sprouting and development of yellow nutsedge tubers. Canadian Journal of Plant Science. 57: 509-514.

16. **Friesen, L.F.** 1986. Biology and control of weedy Cyperaceae species of the Kenyan highlands. MSc. Thesis University of Manitoba. 153pp.

17. **Gardner, F.P.,** and **R.D. Barnett.** 1990. Vernalization of wheat cultivars and a triticale. Crop Science. 30: 166-169.

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

19. **Ghafar, Z.,** and **A.K. Watson.** 1983a. Effect of corn (*Zea mays*) seeding date on the growth of yellow nutsedge (*Cyperus esculentus*). Weed Science. 31:572-575.

20. **Ghafar, Z.,** and **A.K. Watson.** 1983b. Effect of corn (*Zea mays*) population on the growth of yellow nutsedge (*Cyperus esculentus*). Weed Science. 31:588-592.

21. **Glaze, N.C.** 1987. Cultural and mechanical manipulation of Cyperus spp. Weed Technology. 1:82-83.

22. **van Groenendael,** and **J.M.B. Habekotte.** 1988. *Cyperus esculentus* L. - biology, population dynamics and possibilities to control this neophyte. Z. PflKrankh. PflSchutz Sonderh. 11:61-69.

23. **Grummer, G.** 1963. Das verhalten von rhizomen der quecke (*Agropyron repens*) gegen trockene luft. Weed Research. 3:44-51.

24. **Habekotte, B.J.M.,** and **van Groenendael.** 1988. Population dynamics of *Cyperus esculentus* L. (yellow nutsedge) under various agricultural conditions. Med. Fac. Landbouww. Rijksuniv. 53: 1251-1260.

25. **Hauser, E.W.** 1968. Yellow Nutsedge - problems, research trends and outlook. Proceedings of the Northeastern Weed Control Conference. 22:37-48.

26. **Holm, L.G., D.L. Plucknet, J.V. Pancho., J.P. Herberger.** 1977. The Worlds Worst Weeds. The University Press of Hawaii, Honolulu.

27. **Holt, J.S.** 1987. Yellow and purple nutsedge California distribution biotypes and seed production. Proceedings of the California Weed Control Conference. 39:87-89.

28. **Horak, M.J., J. Holt, N.C. Ellstrand.** 1987. Genetic variation in yellow nutsedge (*Cyperus esculentus*). Weed Science. 35:506-512.

29. **Horak, M.J.,** and **J.S. Holt.** 1986. Isozyme variability and breeding systems in populations of yellow nutsedge (*Cyperus esculentus*). Weed Science. 34:538-543.

30. **Horowitz, M.** 1972. Effects of desiccation and submergence on the viability of rhizome fragments of bermudagrass and johnsongrass and tubers of nutsedge. Israel Journal of Agriculture. 22:215-220.

31. **Hsu, F.H., C.J. Nelson, W.S. Chow.** 1984. A mathematical model to utilize the logistic function in germination and seedling growth. Journal of Experimental Botany. 35:1629-1640.

32. **Hunt, R.** 1982. Plant growth curves: The functional approach to a plant growth analysis. Edward Arnold (Publishers) Limited. London, U.K. 248 pp.

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

34. **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.

35. **Keeley, P.E.** 1987. Interference and interaction of purple and yellow nutsedges with crops. Weed Technology. 1:74-81.

36. **Keeley, P.E.,** and **R.J. Thullen.** 1978. Light requirements of yellow nutsedge (*Cyperus esculentus*) and light interception by crops. Weed Science. 26:10-16.

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

38. **Keeley, P.E., R.J. Thullen, J.H. Miller, C.H. Carter.** 1983. Comparison of six cropping systems for yellow nutsedge (*Cyperus esculentus*) control. Weed Science. 31:63-67.

39. **Koutsoyiannis, A.** 1977. Theory of econometrics. 2nd Edition. MacMillan Education Ltd. London. pp 81-91.

40. **Kvalseth, T.O.** 1985. Cautionary note about R^2. The American statistician. 39:279-285.

41. **Lanini, W.T.** 1987. Yellow nutsedge control strategies. Proceedings of the California Weed Control Conference. 39:93-95.

42. **Lapham, J.** 1985. Unrestricted growth, tuber formation and spread of *Cyperus esculentus* L. in Zimbabwe. Weed Research. 25:323-329.

43. **Lapham, J.** 1987. Population dynamics and competitive effects of *Cyperus esculentus* (yellow nutsedge) - prediction of cost-effective control stategies. Proceedings of the 1987 British Crop Protection Conference - Weeds. pp 1043-1050.

44. **Lapham, J.,** and **D.S.H. Drennan.** 1990. The fate of yellow nutsedge (*Cyperus esculentus*) seed and seedlings in soil. Weed Science. 38:125-128.

45. **Little, T.M.,** and **F.J. Hills.** 1978. Curvilinear Relations. Agricultural Experimentation: Design and Analysis. John Wiley and Sons, Toronto. pp 195-227.

46. **Lorougnon, G.** 1969. Etude morphologique et biologique de deux varietes de *Cyperus esculentus* L. (*Cyperacees*). Cahiers Office de la Recherche Scientifique et Technique Outre-Mer Serie Biologie. 10:35-63.

47. **Matthiesen, R.L.,** and **E.W. Stoller.** 1978. Tuber composition in yellow nutsedge (*Cyperus esculentus* L.) variants. Weed Research. 18:373-377.

48. **McDonald, I.D.** 1986. An evaluation of the effect of the depth of tillage on the placement and viability of yellow nutsedge (*Cyperus esculentus* L.) tubers. MSc Thesis. University of Guelph. 56pp.

49. **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.

50. **Nash, M.J.** 1985. Crop conservation and storage in cool temperate climates. 2nd Ed. Pergamon Press. Oxford. 286pp.

51. **Nicholls, A.O.,** and **D.M. Caller.** 1973. Comments on the use of regression analysis for the study of plant growth. New Phytlogist. 72:571-581.

52. **Nickel, K.P.** 1989. The growth and competitiveness of round-leaved mallow (*Malva pusilla* Sm.) in spring wheat (*Triticum aestivum* L.). MSc. Thesis. University of Manitoba. 94pp.

53. **Patterson, D.T.** 1982. Shading responses of purple and yellow nutsedges (*Cyperus rotundus* and *C. esculentus*). Weed Science. 30:25-30.

54. **Patterson, M.G., G.A. Buchanan, J.E. Street., R.H. Crowley.** 1980. Yellow nutsedge (*Cyperus esculentus*) competition with cotton (*Gossypium hirsutum*). Weed Science. 28:327-329.

55. **Ross, M.A.,** and **C.A. Lembi.** 1985. Applied Weed Science. Burgess Publishing Company. Minneapolis. pp. 20-45.

56. **Smith, E.V.,** and **E.L. Mayton.** 1937. Nut grass eradication studies: II. The eradication of nut grass, *Cyperus rotundus* L., by certain tillage treatments. Journal of the American Society of Agronomy. 29:1007-1013.

57. **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.

58. **Stoller, E.W.** 1975. Growth, development, and physiology of yellow nutsedge. Proceedings of North Central Weed Control Conference. 30:124-125.

59. **Stoller, E.W.** 1981. Yellow nutsedge: A menace in the corn belt. U.S. Dept. Agric. Tech. Bull. No. 1642.

60. **Stoller, E.W., D.P. Nema., V.M. Bhan.** 1972. Yellow nutsedge tuber germination and seedling development. Weed Science. 20:93-97.

61. **Stoller, E.W.,** and **R.D. Sweet.** 1987. Biology and lifecycle of purple and yellow nutsedges (*Cyperus rotundus* and *C. esculentus*). Weed Technology. 1:66-73.

62. **Stoller, E.W.,** and **L.M. Wax.** 1973. Yellow nutsedge shoot emergence and tuber longevity. Weed Science. 21:76-81.

63. **Stoller, E.W.,** and **E.J. Weber.** 1975. Differential cold tolerance, starch, sugar, protein, and lipid of yellow and purple nutsedge tubers. Plant Physiology. 55:859-863.

64. **Stoller, E.W.,** and **J.T. Woolley.** 1983. The effects of light and temperature on yellow nutsedge (*Cyperus esculentus*) basal-bulb formation. Weed Science. 31:148-152.

65. **Sturko, A.R.W.** 1983. Yellow nutsedge, a new intruding weed in Manitoba. Manitoba Agronomist's Proceedings. pp 52-54.

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

67. **Thomas, P.E.L.** 1969. Effects of desiccation and temperature on survival of *Cyperus esculentus* tubers and *Cynodon dactylon* rhizomes. Weed Research. 9:1-8.

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

69. **Thullen, R.J.,** and **P.E. Keeley.** 1978. The effect o*f Cyperus esculentus* tuber maturity on ^{14}C accumulation. Weed Science. 26:270-273.

70. **Thullen, R.J.,** and **P.E. Keeley.** 1979. Seed production and germination in *Cyperus esculentus* and *C. rotundus.* Weed Science. 27:502-505.

71. **Thullen, R.J.,** and **P.E. Keeley.** 1987. Influence of the date of planting on the growth of yellow nutsedge (*Cyperus esculentus*). Weed Science. 35:173-176.

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

73. **Tumbleson, M.E.,** and **T. Kommedahl.** 1962. Factors affecting dormancy in tubers of *Cyperus esculentus.* Botanical Gazette. 87:186-190.

74. **Vegis, A.** 1964. Dormancy in higher plants. Annual Review of Plant Physiology. 15:185-224.

75. **Versavel, P.A.,** and **W.E. Muir.** 1988. Drying behavior of high moisture content wheat spikes. American Society of Agricultural Engineers. 31:1260-1263.

76. **Wall, D.A.** 1989. Bladder campion (*Silene vulgaris* (Moench) Gracke): Investigations on its biology, growth and response to herbicides. PhD Thesis. University of Manitoba. 148pp.

77. **Wiebe, T.J.** 1983. Water - temperature interactions in germination and dormancy in wheat. MSc. Thesis. University of Manitoba. 195pp.

78. **Williams, R.D.** 1981. Intraspecific competition of yellow nutsedge. Proceedings of the Southern Weed Science Society. 34:231-238.

79. **Williams, R.D.** 1982. Growth and reproduction of *Cyperus esculentus* L. and *Cyperus rotundus* L. Weed Research. 22:149-154.

80. **Wills, G.D.** 1987. Description of purple and yellow nutsedge (*Cyperus rotundus* and *C. esculentus*). Weed Technology. 1:2-9.

81. **Wills, G.D.**, R.E. Hoagland., R.N. Paul. 1980. Anatomy of yellow nutsedge. Weed Science. 28:432-437.

82. **Wright, E.B.** 1984. Tolerance of winter wheat to fall-applied herbicides and dinitrolaniline herbicide residues. MSc Thesis. University of Manitoba. 119pp.

83. **Yip, C.P., R.D. Sweet, J.B. Sieczka.** 1974. Competitive ability of potato cultivars with major weed species. Proc. North East Weed Control Conf. 28:271-281.

84. **Yip, C.P.** 1978. Yellow nutsedge (*Cyperus esculentus* L.) ecotypes, their characteristics and responses to environment and herbicides. PhD Thesis. Cornell University. 309pp.

Appendix 1

	Name and Address	Location	Size
1.	Barnabe Bros. Box 259 Letellier, MB. R0G 1C0 737-2339	R.L. 95	2-3 acres
2.	Mr. D. Houle Box 313 Letellier, MB. R0G 1C0 737-2386	R.L. 108, 110	1-2 acres
3.	Mr. B. Cadieux Box 345 Letelleir, MB R0G 1C0 737-2635 or 737-2319	R.L. 121	Spotty over 20 acres.
4.	Mr. G. Fontaine Fontaine Farms Lettelier, MB R0G 1C0 737-2398	R.L. 181-179	Patches covering one half acre.
5.	Mr. R. Fillion Box 216 St. Jean Baptiste, MB. R0G 2B0 758-3484	R.L. 191, 193	3 patches 50 sq.ft.
6.	Mr. P. Sabourin Sabourin Seeds Ltd. St. Jean Baptiste R0G 2B0 758-3595	R.L. 195-197	
7.	Mr. R. Manikel St.Jean Baptiste, MB R0G 2B0 758-3543	R.L. 215	
8.	Mr. A. Sabourin Box 340 St. Jean Baptiste, MB. R0G 2B0 758-3595 or 737-2202	R.L. 217, 219	Patches covering 1 acre.

9.	St. Jean Baptiste Park St. Jean Baptiste, MB	R.L. 241	Small patches along edge of river.
10.	Mr. A. DeCruyenaere Box 48 St. Adolphe, MB. R0A 1S0 882-2388	R.L. 248	20 sq foot patch.
11.	Lorandon Farms Box 612 Morris, MB. R0G 1K0 758-3531	R.L. 259, 261 325-331	Small patches from St. Jean to Morris.
12.	Mr. G. Marion Box 297 St. Jean Baptiste, MB. R0G 2B0 758-3885	R.L. 264	Patches covering 5 acres along the river.
13.	Mr. R. Lafond Box 273 St. Jean Baptiste, MB. R0G 2B0 758-3937	R.L. 278, 276.5	One half acre patch and smaller patches covering 50 acres.
14.	Mr. R. Marion Mr. G. Marion St. Jean Baptiste, MB R0G 2B0 758-3491 or 758-3463	R.L. 280-282 284	Patches covering 5 acres.
15.	Mr. G. Vermette Box 164 St. Jean Baptiste, MB. 746-8227	R.L. 285	3 - 4 spots 5-6 sq.ft.
16.	Mr. C. Vermette Box 102 Morris, MB. R0G 1K0 746-8372	R.L. 299, 301	1500 sq.ft. on each lot
17.	Mr. A. Snarr Box 58 Morris, MB. R0G 1K0 746-8598	R.L. 383, 381, 379	15-20 small patches.

18.	Mr. M. St. Onge Aubigny, MB. R0G 0C0 882-2380	R.L. 461	Various sized patches covering 20 acres.
19.	Mr. J. Blatta Ste. Agathe, MB R0G 1Y0 882-2110	R.L. 490	Small patches at rivers edge.
20.	Glenlea Research Station University of Manitoba		0.5 acre
21.	Mr. E. Zylema 4176 St. Mary's Rd. Winnipeg, MB.	R.L. 189	1 acre

APPENDIX 2

Fungicide Treatment of Tubers

Introduction

Preliminary research on the influence of various physiological stresses on tuber viability were unsuccessful since tuber viability determination was rapidly confounded by fungal invasions. A soaking treatment similar to that described by Shurtleff, (1966) was used on tubers to remove the fungal pathogens. These treatments were required for all tuber desiccation and freezing trials conducted. The objectives for this experiment were to first, determine the effectiveness of the fungicides on controlling fungal growth and secondly, to determine the influence of the fungicides on tuber viability.

Materials and Methods

Tubers were placed in 1500 ml flasks with a 1100 ml of treatment solution. The treatment solution contained 2.25 g captan and 0.98 g Benomyl (50% wp). Tubers were soaked for 1, 3 and 24 hours in this treatment solution that was stirred continuously. Untreated controls were placed in water for equivalent times. After the fungicide treatment, tubers were allowed to dry to 65% of their original weight to simulate a desiccation treatment. Viability was assessed using the technique prescribed by Yip (1978) and Stoller (1973). Tubers were incubated for a period of 6 weeks at 22 C during which time sprouted tubers were removed and the remaining tubers were cut in half and dormant and dead tubers were seperated based on the hardness and color of the tissue inside. White hard tissue was associated with dormant viable tubers and yellow to brown, soft tubers were nonviable.

Each treatment had five replications with 10 tubers per treatment. The data were analyzed using Duncan's multiple range test at the 5% level of significance.

Results and Discussion

The fungicide was equally effective among all treatments for controlling fungal growth on the tubers. Tuber viability was not affected by the desiccation treatment and no significant difference in tuber viability was observed between the fungicide treatments.

Treatment	Percent Viable
Untreated - 1 hour	95 a
Untreated - 3 hour	96 a
Untreated - 24 hour	91 a
Treated - 1 hour	92 a
Treated - 3 hour	99 a
Treated - 24 hour	96 a

Although no significant differences were observed the greatest percent viable was observed in the 3 hour treatment with fungicide. This treatment was chosen for use in all subsequent experiments.

Appendix 3

Influence of nutsedge interference on barley dry matter production. Values are the means of eight replications.

DAE	Barley Dry Matter Production	
	- Nutsedge grams m^{-1}	+ Nutsedge grams m^{-1}
0	0	0
18	104.4	80.8
36	703.7	449.4
54	1101.0	785.4
72	848.9	589.1