Identifying the optimum storage and handling conditions for sweetpotato slips

by

Joseph Rundquist

B.S., Kansas State University, 2015

A THESIS

submitted in partial fulfillment of the requirements for the degree

MASTER OF SCIENCE

Department of Horticulture and Natural Resources
College of Agriculture

KANSAS STATE UNIVERSITY
Olathe, Kansas

2020

Approved by:

Co-Major Professor
Dr. Eleni Pliakoni

Approved by:

Co-Major Professor
Dr. Jason Griffin

Copyright

Abstract

Sweetpotato [*Ipomoea batatas* (L.) Lam.] is a tropical perennial native to the Americas. Sweetpotato is propagated through vine cuttings known as slips. In the United States, Production is concentrated in the Southeast region and sweetpotato growers in the Northern and Central regions are left reliant on plant material that is shipped from outside their region. Transportation and storage conditions can result in low quality slips that may perform poorly after being planted in the field. The objective of this study was to identify the optimal storage and shipping conditions related to temperature and packaging for sweetpotato. Sweetpotato slips, cultivar 'Orleans', were harvested at the John C. Pair Horticultural Center (Haysville, Kansas, USA) and transported to the Postharvest Physiology Laboratory at Kansas State University Olathe (Olathe, Kansas, USA). 50 slips were placed into small waxed cardboard boxes (12" x 4" x 4") with or without a nylon film liner. Boxes were stored at three different temperatures: 16°C, 22°C, 30° at 65% relative humidity. An overall quality rating scale was developed to evaluate the visual quality of the slips with ratings from 1 to 9 (1- completely senesced to 9- field fresh slip). Changes in slip quality were evaluated throughout storage by measuring overall visual quality, water loss, chlorophyll fluorescence, respiration, color, and chlorophyll content. Slips stored at 16°C with a liner had the longest shelf life maintaining marketable quality for just over 11.3 days, followed by 10.6 days for 16°C without a liner, 8.3 days for 22°C with a liner, 7.5 days for 22°C without a liner, and 6.5 and 6.4 days 30°C with and without a liner respectively. After 4 days of storage, slips stored with a nylon film liner exhibited significantly lower rates of water loss at all temperatures ($P \leq .05$). We also conducted a second experiment with the objective of investigating the influence of slip storage duration on slip establishment, growth, and storage root yield. Slips beds were established at the Olathe Horticulture Research and Extension

Center. Slips were stagger-harvested from the same bed, every 3 days in order to achieve storage durations of 0, 3, 6, 9, and 12 days. All slips were held at 22°C and 65% relative humidity. The slips were then planted in the open field at John C. Pair Horticultural Center as well as replicate trial at the Olathe Horticulture Research and Extension Center. After planting the slips, a series of measurements were taken to evaluate establishment and growth. These measurements included: survivability, stem diameter, vine length, leaf area, root biomass, and shoot biomass. In addition to these measurements storage root yields were weighed after 93 days of growth. Slips planted the same day of harvest (0 days in storage) established the quickest after transplant according to various parameters. However, slips that were stored for 6 days often out performed slips stored for 3 days in establishment and growth measurements. Plants produced from slips planted the same day of harvest (0 days in storage) and slips stored for 6 days had significantly higher bulk yields than those stored for 12 days prior to planting ($P<.01$). The results from this study indicate that storing slips for more than 6 days prior to planting could have a detrimental effect on early vine growth as well as, storage root yield. Our experiments are the first, that we know of, to investigate, in-depth, the postharvest quality changes of sweetpotato slips and how storage root yield is affected by slip storage duration. Our study confirms a consensus view within postharvest research that temperature management is the most important tool for maintaining the quality of fresh horticultural commodities. Information from this study increases the postharvest handling knowledge of sweetpotato slips with hopes of contributing to the development of standardized practices for shipping and storing sweetpotato slips.

Table of Contents

List of Figures

List of Tables

Chapter 1 - Introduction

Sweetpotato [*Ipomoea batatas* (L.) Lam.] is a tropical American vine from the morning glory family Convolvulaceae (Purseglove, 1968), which consists of approximately 1,650 species (Xu and Chang, 2017). Within Convolvulaceae, sweetpotato is the most important food crop, with widespread consumption and a diversity of culinary applications (Woolfe, 1992). While the entire aerial vine is edible, the large, sweet and starchy storage roots of the sweetpotato are the most widely-consumed and commercially important portion of the plant (Padmaja, 2009). Fresh sweetpotato storage roots can be prepared and consumed in a variety of ways, including raw, fried, baked, boiled, or canned for long-term storage (Padmaja, 2009). While not widely consumed in the United States, the leaves and shoots of the sweetpotato vine are consumed as greens, incorporated into dishes, or used as animal fodder throughout the world, especially in some African countries and East Asia (Scott et al., 2000 a, b; Thottappilly, 2009; Woolfe, 1992). Sweetpotato storage roots are typically produced to be sold fresh to market. However, sweetpotato use has diversified considerably over the last four decades (CIP, n.d.a). Many countries have started to process sweetpotato into value-added products such as juice, dried/fried chips, candy, soda, noodles, flour, and liquor to name a few (Thottappilly, 2009). Advances in technology have made it possible for sweetpotato to be used in various industrial processes such as the production of biofuels and starch additives (Carpena, 2009; Lareo et al., 2013). Additionally, there is a growing interest in the use of anthocyanin pigment from purple sweetpotato cultivars for food colorings and cosmetics (CIP, n.d.a). Certain *I. batatas* cultivars have also been bred for their attractive foliage and are used specifically for ornamental horticulture applications but are not recommended for human consumption (Smith et al., 2009).

Originally domesticated in the American tropics at least 5000 years ago, sweetpotato is thought to be one of the earliest agricultural food crops (Austin, 1988; Yen,1982). Archeological remains discovered in Peru indicate that humans have been consuming sweetpotato since at for 8,000-10,000 years (Bovell-Benjamin, 2007; Austin, 1988). The exact origin of the sweetpotato is unclear and has been the subject of extensive research (Zhang et al., 2000). However, thanks to Archaeological, linguistic, genetic and ethnobotanical data, researchers now consider the area between the Yucatan Peninsula of Mexico and the Orinoco River in Venezuela to be the primary center of diversity and most likely center of origin (Huang and Sun, 2000; Loebenstein, 2009; Roullier et al., 2013; Woolfe, 1992; Zhang et al. 2000). The sweetpotato diffused to Oceania (Hawaii, Polynesia, and New Zealand) in pre-Columbian times either by natural processes or by early seafarers (Mwanga et al., 2017; Rollier et al., 2013). In the 16th century Spanish voyagers introduced the crop to the Philippines, thereafter, the crop spread westward to mainland Asia; reports indicate the sweetpotato had arrived in China by 1594 (Grüneberg et al., 2017). During the same period, Spanish and Portuguese seafarers spread the sweetpotato eastward out of the Central and South America to the Mediterranean, Africa, India, and into South-east Asia (Mwanga et al., 2017; O'Brien, 1972; Rollier et al., 2013)

Sweetpotato is a hexaploid crop (2n=6x=90) of which there are thousands of known varieties (CIP, n.d.b). The International Potato Center (CIP) in Lima, Peru has an extensive collection of over 8,000 cultivars from all over the world; roughly 1,000 of which are wild varieties (CIP, n.d.b). While many wild relatives and varieties have been identified, any direct ancestors have yet to be discovered (Woolfe, 1992). Unlike the potato (*Solanum tuberosum*), which is a true tuber or modified stem (Spooner, 2013), the sweetpotato a tuberous root. The tuberous root of the sweetpotato, also known as a storage root, functions as a sink, storing large amounts of starch during the growth of the plant (Hattori et al., 1990). Sweetpotato storage roots grow beneath the soil and produce, on average, 4-10 storage roots per plant (Thottapilly 2009). However, yield is variable and highly dependent on several factors, including cultivar, soil type, propagule quality, and production practices (Lowe and Wilson, 1974; Togari, 1950). Sweetpotato storage roots come in a wide range of skin and flesh colors, ranging from white, yellow-orange, and red, to blue and deep purple (CIP, n.d.b). The above-ground portion of the plant is a vine that forms a thick canopy within a few weeks of planting. The leaves of the sweetpotato vine are variable in size, shape, and color across cultivars. The single flowers are funnel shaped and are white or reddish-violet in color; typical of the morning glory family (Thottappilly, 2009). Sweetpotato can grow at altitudes ranging from sea level to 2,500 meters and is well adapted to large or small-scale production systems, in several climates, and can be grown successfully using a variety production practices (CIP, 2010; Ewell, 1990).

Globally, sweetpotato is the sixth most important food crop behind wheat, rice, potatoes, maize, and cassava (Drapal et al., 2019). Over 105 million tons of sweetpotato are produced each year around the world with approximately 95% of the production taking place in developing countries, where it is the fifth most important food crop in terms of fresh weight (Drapal et al., 2019; CIP, 2010). Sweetpotato is increasing in importance as a food crop in some regions of the world. More than 40% of children under age five in Sub-Saharan Africa are affected by Vitamin A deficiency, contributing to high rates of disease, vision impairment, and premature death in children and pregnant women (Low et al., 2017). Orange–fleshed sweetpotato varieties contain high levels of the precursor to Vitamin A, beta-carotene, just 100g of a fresh sweetpotato storage root from most orange-fleshed varieties contains enough beta-carotene to provide the daily pro-vitamin A needs of a young child (Low et al., 2017). Sweetpotato storage roots are also a good source of Vitamins B, C, and E and contain moderate levels of zinc and iron (Bovell-Benjamin, 2007). Asia is currently the largest sweetpotato producing region of the world with China accounting for over 80% of the world's production (CIP, 2010). Roughly half of all sweetpotato produced in Asia is for animal fodder, with the remainder being used primarily for human consumption (Crop Trust, 2019). Nutritionists at Kansas State University have investigated the cancer fighting potential of purple-fleshed, high-anthocyanin varieties of sweetpotato. Lim et al., demonstrated in their 2013 study the anticancer activity of P40, a purple-fleshed sweetpotato variety, in both *in vitro* cell culture and *in vivo* animal model. Their findings suggest that the purple, anthocyanin-enriched sweetpotato variety 'P40' has positive colorectal cancer preventive benefits without the toxicity, suggesting a potential for dietary use in at-risk populations (Lim et al., 2013).

Production and Consumption of Sweetpotato in the U.S.

Sweetpotato is in the top ten most commonly consumed fresh vegetables in the country although the United States produces and consumes considerably less sweetpotato than many countries in the world (FAO, 2017). While research has indicated that the sweetpotato is native to the Americas, no evidence suggests that the sweetpotato was cultivated by the indigenous populations of North America (Edmond, 1971; Loebenstein, 2009; Roullier et al., 2013; Woolfe, 1992). The sweetpotato is not thought to have been widely cultivated in the U.S. until the 18th century; although, the first reports of sweetpotato in the modern-day U.S are from Virginia in 1648 (O'Brien, 1972; Smith et al., 2009). During the colonial era in the early United States the sweetpotato was primarily produced in the southeastern region of the country (Gray et al., 1933). During this period, sweetpotato was an important crop; used for making bread, molasses, vinegar, beer, and was often used as animal fodder (Gray et al., 1933).

The sweetpotato became important in the United States during the great depression era of the 1930's (Smith et al., 2009). Sweetpotato production peaked to more than 900,000 planted acres during the great depression; with per capita consumption of more than 25lb annually (Smith et al., 2009; USDA ERS, n.d.). During the 1930's an emphasis was placed on self-sufficiency and homesteading, and the sweetpotato was grown in many regions of the country as a result (Edmond, 1971; Smith et al., 2009). However, during this period, sweetpotato yields decreased as acreage increased indicating limited income was available for inputs such as fertilizer (Smith et al., 2009). Second to the Irish potato, sweetpotato was one of the most highly-produced vegetable crops in the state of Kansas in 1938 (Elmer, 1938). After World War II, sweetpotato production and consumption in the United States experienced a downward trend that lasted well into that latter parts of the 20th century (USDA ERS, 1994). Subsequently there were several decades of waning consumption and production with, just 100,000 planted acres and a per capita consumption of 4.5lb annually from 1970-2007 (Smith et al., 2009; USDA ERS, n.d.).

In the last 20 years, both consumption and production of sweetpotato has increased substantially in the United States (Bond, 2017). In 2000, total 95,000 acres of sweetpotato were harvested which equaled 13.8 million hundredweight (cwt), close to the 10-year (1990-99) average of 12.42 million cwt (USDA ERS, 2015). Since 2000 sweetpotato production has increased by 6% every year, leading to record high production with 163,000 acres harvested (31.54 million cwt) in 2016 (AgMRC, 2018). While sweetpotato was produced in many regions of the country in the early parts of the 20th century, current production is largely concentrated in four states: North Carolina, California, Mississippi, and Louisiana (Bond, 2017; Estes,2009). North Carolina is the leading sweetpotato production state, producing approximately 60% of all sweetpotato grown in the country. According to the USDA's Economic Research Services North

Carolina produced 5.6 million cwt of sweetpotato in 2000. By 2014, production in North Carolina had expanded immensely to 15.8 million cwt. With a 185% increase in North Carolina's production the state has kept the sweetpotato industry in the United States afloat. However, other core production states have also seen large increases in production. Over the same period, California sweetpotato production saw an increase of nearly 100%; Mississippi's production has increased by 155% (USDA ERS, 2015). The USDA's National Agriculture Statistics Service estimated that in 2016 there were 3.1 billion lb. of sweetpotato in production, with a gross revenue of more than $705million. By 2017 national sweetpotato production was 35.6 million cwt; a 13% increase from the previous year (USDA NASS, 2018).

The rise in consumer demand for sweetpotato in the United States has largely been encouraged by promotion of the crop's health benefits (Bond, 2017). Consumers have, in recent years, become more concerned with the importance of health, wellness, and maintaining a balanced diet (Bond, 2017). Because the sweetpotato provides a well-balanced provision of important macronutrients, vitamins, and minerals, the sweetpotato has become hailed as a "superfood" or "powerhouse" vegetable. (Di Noia, 2014; Smith et al., 2009). As a result, domestic availability, an indicator of consumption trends, has increased from 4.2 lb. in 2000 to 7.5 lb. in 2014 (Bond, 2017; USDA ERS, n.d).

Local Market Demand

In a survey conducted in 2014, the Food Market Institute found that among U.S. shoppers, the top three motivations for purchasing local foods were: freshness, support of the regional economy, and taste (Brain, 2012). The increase in per capita consumption of locally-produced fruits and vegetables among U.S. consumers has led to notable increases in sales for the local produce industry (USDA-NASS, 2012). Package Facts, a leading market research publisher anticipates that local foods will grow faster than the annual pace of total food and beverage sales reaching nearly $20 billion in 2019. David Sprinkle, Research Director at market research publisher Packaged Facts, points out that “Over the past 10 years, there has been a surge in consumer demand for locally-produced foods, along with widening availability” Furthermore, it’s not just natural food retailers and farmers markets that are contributing to this trend. An increasing number of large, retail grocery chains are offering and promoting locally sourced food products. “Even Walmart has been promoting local farmers in its bid to tailor its store selections more toward local communities” (Sprinkle, Packaged Facts, 2015). In November of 2014, Packaged Facts conducted its National Consumer Survey. The survey found that among U.S. adults, 53% of respondents specially seek out locally-grown or locally-produced foods, with 19% “strongly” agreeing and 34% “somewhat” agreeing. Furthermore, nearly half the respondents agreed that they would be willing to pay up to 10% more for locally grown or produced foods, and almost a third said they are willing to pay up to 25% more. A third of consumers also claim to consciously purchase locally-grown or locally-produced foods a minimum of once per week (Sprinkle, Packaged Facts, 2014).

Two separate food hub feasibility studies conducted in the Kansas City metro as well as Northeast Kansas concluded that the demand for locally-grown fresh produce exceeds the supply provided by local producers (Flaccavento et al., 2014; Greater Kansas City Food Hub Working Group, 2015). Specifically, within the Kansas City area, the feasibility study determined that the value of unmet demand for local fruits and vegetables was more than $150 million (Greater Kansas City Food Hub Working Group, 2015) As these figures suggests, the greatest challenge of producing local food is meeting consumer demand in terms of both availability and providing a consistent, high-quality product. This can be especially challenging for smaller scale producers which make of most direct-to-consumer operations. The Food Hub Feasibility Study: Northeast Kansas made specific mention of sweetpotato as “core item” in the regions local food system because of its “appearance, flavor, freshness, uniqueness of varieties, and good production conditions in the region” (Flaccavento et al., 2014).

Propagation and Commercial Sweetpotato Slip Production

The sweetpotato is a hexaploid species (2n=6x=90). Having six copies of each of its 15 chromosomes, for a total of 90 somatic chromosomes (Jones, 1967). Sweetpotato also has difficulty selfing due to sterility, incompatibility, and uneven or complete failure to bloom in temperate climates (Jones, 1967). Given the genetic complexity of the crop, seeds are typically only used for breeding purposes, making the methods through which sweetpotato is propagated unique among vegetable crops (Gurmu et al., 2012; Woolfe, 1992). There are three primary ways through which sweetpotato reproduction occurs (Woolfe, 1992). The plant can reproduce asexually and populate an area by allocating energy from photosynthesis into storage roots. In tropical regions, storage roots (if not harvested) will ultimately sprout to produce new plants, all of which are genetic clones of the original mother plant. Sweetpotato can also reproduce by allocating significant energy into its vines. When the vine comes in contact the soil, roots will form at the nodes, producing new plants. Lastly, and of least importance numerically is sexual reproduction through seed. The plant allocates very little energy to this form of reproduction (Woolfe, 1992). Additionally, embryonic seeds will rarely produce true-to-type offspring and is therefore not suitable for commercial agricultural applications but are used in breeding programs (Loebenstein et al., 2009).

Commercially, sweetpotato is propagated by transplanting stem cuttings, which are adventitious sprouts produced by planting storage roots that were held over from the previous year. These cuttings are commonly referred to as “slips”. Seed roots (storage roots saved from the previous year) are planted in propagation beds from which slips will be grown and harvested. When the slips reach approximately 1’ in length, they are harvested and directly transplanted on-farm, or sold and distributed to other producers. Like the potato new sweetpotato storage roots can be grown from whole or pieces of saved storage roots. However, it has been reported that slip yield and quality are poor when grown from pieces of storage root (George et al., 2011) and commercial practices typically include bedding whole seed roots.

Most commercial sweetpotato growers in U.S. purchase slips annually from slip producers. The majority of slips available throughout the U.S. are commonly shipped in from major production states like North Carolina. Slip prices vary depending on several factors including if the slips are organic, or virus-tested, and how many plantings generations the stock is removed from micro-propagation (i.e. G1, G2, G3). As of 2018, organic wholesale prices for three orange flesh varieties range from $60/1000 (Jones Farm, Bailey, NC) to $120/1000 plants (John C. Pair Horticultural Center) to $462/1000 (Johnny’s Selected Seeds, Fairfield, ME). Exotic, rare, or heirloom cultivars can sell for more than $1.00/slip (Sandhill Preservation Center, Calamus, IA). Currently no standardized value for sweetpotato slips exist at the retail or wholesale level. Additionally, there is little information available regarding the market value of slip production in the U.S.

Most slips sold in the U.S. are stem cuttings from bedded seed roots, although some nurseries and clean plant centers will sell multiplied slip cuttings taken from recently planted slips (Smith et al., 2009). Each year, a portion of the storage roots that are harvested in late summer and fall will be stored separately, held through the winter, and used to produce the next years slip stock. Major production states typically reserve small to medium sized storage roots, referred to as canners (diameter 1-1.75 in), for propagation bed planting, although any size of storage roots can be sprouted, (Smith et al., 2009; Stoddard, 2013). Smaller roots are preferred because sprouts largely form at the proximal end of the root and small roots provide greater sprouting points/ft² (Coolong et al., 2012). However, reports indicate that utilizing only small storage roots for slip propagation could lead to inferior traits in the progeny (Coolong et al., 2012). In the U.S. sweetpotato storage root production predominantly takes place in the open field, in raised bed or hilled rows on 48" row centers. Slips are typically planted in 12" row spacing, requiring approximately 10,890 slips per acre. Commercial slip producers recommend bedding 1 bushel (40-50 lbs.) of seeds roots to produce 500 slips. On average, one acre of bedded seed root should produce approximately 600 acres of slips (Jones Farm, Baily, NC; Stoddard, 2006). Nursery producers in North Carolina reportedly employed anywhere from 24 to 73 bushel/1000ft² (50 lbs./bushel) to plant their seedbeds (Barkley et al., 2017a). Commercial production manuals and extension publications vary in their recommendations for seed root planting density in propagation beds. Some make suggestions based on seed root weight, and/or seed root count. Coolong et al. (2012) recommend seven seed roots averaging 8oz in weight/ft². Large commercial nurseries suggest laying seed as close together without overlap, which they claim amounts to 1.0 bushel of seed/20-30ft² (Jones Farm, Bailey, NC).

Large sweetpotato slip operations require the implementation of mechanized agricultural equipment. In these larger operations, seed roots are placed into a large hopper, conveyed on a belt into the propagation beds. Seed roots are typically laid at soil grade or onto shaped ridges and covered with 2 in of soil (Wilson et al., 1977). Beds are commonly covered with either clear or black polyethylene mulches, usually 1.5-2ml 15 thick (Smith et al., 2009). Along with hand tools and manual labor, large mechanized implements can be pulled behind a tractor to lay plastic and cover edges with soil, helping to secure the mulch. Plastic mulches help maintain adequate soil moisture while simultaneously raising soil temperature for sprouting in early spring (Barkley, 2015; Saglam et al., 2017). Openings are cut in the mulch layer to promote ventilation and temperature regulation which results in controlling decay brought about by high temperatures and the buildup of CO_2 levels (Boudreaux et al., 2005, Schultheis et al., 2008). Ultimately, the plastic mulch will be removed upon sprouting of the seed roots or when temperatures increase.

Although variable, once the average distance from the soil to canopy reaches 10-14in, the vines are cut with hand shears, grass sickle, or mechanized tools (Barkley, 2015). Ideally slips should be cut at least 1 inch above soil line, harvesting implements should be sanitized before use to reduce the spread of disease during slip harvest (Clark et al., 2009). Because the height of the canopy varies from point to point within the propagation bed, slip size is often variable (Barkley et al., 2017b). Sweetpotato slips can be harvested from the same bed multiple times over the course of one grow season. Requiring approximately four weeks of growth between harvests. However, large commercial slip producers typically implement a “once-over harvest” practice. Taking only one harvest from each bed of sprouted seed roots (Barkley et al., 2017a). Transplanting undersized slips can results in improper planting depth or inadequate plant tissue above the soil and is usually avoided by large producers as they are not suitable for interfacing with mechanized planters (Thompson et al., 2017a). Sweetpotato slips are typically planted a depth of 8 - 15 cm and slips less than 12 cm long will likely fail if planted below the soil and are therefore not viable (Barkley, 2015). In a 2014 study in North Carolina, significantly greater total storage root production at a level of $P \leq 0.10$ was found for slips transplanted at depth of 15.2 cm compared to shallower planting depths ($P=.088$) for cv. ‘Covington’ (Thompson, 2014). A separate study by Thompson et al. (2017) reported that slips measuring between 20 cm and 30 cm had greater survival rates and higher storage root yields compared to that of shorter slips. The authors also reported significantly greater US #1 and total storage roots/plant for slips $\geq$ 15.9cm (Thompson, 2014). However, research indicates that slips with an apical meristem will perform better after transplant (Hossain and Mondal, 1994; Low et al., 2009). Another important factor regarding slip quality is the number of nodes present on the slip. Stem nodes are the site of root primordia where adventitious roots are produced and develop into the desired fleshy storage roots (Firon et al., 2009). A minimum of three nodes under the soil surface has been recommended. Slips that are considered too short are often discarded as they lack enough nodes (Thompson, 2014).

Slip Production in the United States

In terms of acres planted, sweetpotato production in the United States was approximately 150,000 acres in 2018. Down from 162,000 acres in 2017, and 169,000 acres in 2016 (USDA-NASS, 2018). Based on the 150,000 acres planted in 2018 we can estimate the market at approximately 1.6 billion slips that are currently being produced in the U.S. Slip producers, primarily in leading production states specialize in production for both wholesale and retail markets. Each of these leading sweetpotato producing states has their own active grower organizations and advertising commissions, which promote, advertise and support the sweetpotato industry in the U.S. (Smith et al., 2009). Sweetpotato grower organizations and commissions have become models for crop-specific economies. In the last 50 years, the leading production states have become increasingly vertically integrated to manage the germplasm, breeding, inputs, tools, storage infrastructure, processing and distribution for most of the U.S. sweetpotato industry (Smith et al., 2009).

Given the consolidation of the sweetpotato industry, the available supply of sweetpotato is increasingly vulnerable to shortages due to losses caused by extreme weather. North Carolina, Mississippi, and Louisiana, all key production states, happen to be situated in areas of the country affected by hurricanes. In 2016, Hurricane Matthew caused severe flooding to the primary production areas of North Carolina at harvest (Bond, 2017). Similarly, the excessive rains from Hurricane Harvey negatively impacted production in Louisiana and Mississippi in 2017 (USDA NASS, 2018). Natural disasters and their accompanying difficulties pose a major risk to national supply of sweetpotato storage root and slip stock and therefore could affect food security. Because the production of sweetpotato slips is concentrated in Southeast US an increasing concern that regional food systems within the U.S. are overly reliant on consolidated supply chains and outside inputs, are in response to potential food insecurity and apparent lost revenues in the local economy (Woods et al., 2013). This concern has been reflected even outside the United States. In Canada, nationally funded research is developing production methods for sweetpotato slips. An apparent over reliance on slip stock from the U.S. has become deemed a “bottleneck” to their regional crop production (Vineland et al.,2017). This example echoes a similar, interregional situation occurring in the U.S. where, for example, sweetpotato producers in the Midwest are reliant on slip producers in the Southeast. According to many researchers and international agriculture organizations, access to seed and all other of genetic resources is one the “crucial elements” for the sustainability and prosperity of farming communities (FAO, 2017; Reuter, 2017). Seeds and propagative plant material (like sweetpotato slips) are the basis of all production and the genetic improvement of crops. A lack of readily available reproductive materials for agriculture leads to a weakened regional food security (Godfray et al., 2010). Efforts to address regional access to seeds and propagative material often involve promoting increased seed sovereignty, which has become an important issue for farmers both within the U.S., and at the international level. Seed sovereignty as defined by Indian activist and scholar Vandana Shiva is “the farmer’s rights to save, breed and exchange seeds, to have access to diverse open source seeds which can be saved, and which are not patented, genetically modified, owned or controlled by emerging seed giants” (Shiva, 2016). The availability of seed, propagates, and resources at the local and regional level, supports access to profitable cultivars that are well adapted to local biophysical factors and provide increased local revenues with markets for planting material (Coomes et al., 2015).

Postharvest Handling and Transport of Slips

Fresh fruit and vegetable are living commodities that undergo continuous changes after harvest. While postharvest handling, by definition, begins at harvest, there are several preharvest factors that can affect the shelf life and postharvest quality of fresh commodities. These factors include the climatic conditions where the commodity is produced, the cultural practices employed in producing the commodity, and genetics and cultivar selection (Weston and Barth, 1997).

Climatic factors which are often unmanageable in field production, have been proven to have important influence on the quality and nutritional value of freshly harvested fruits and vegetables. Light intensity is an important climatic factor because concentration of ascorbic acid in plant tissues generally increases as exposure to light increases (Kader, 1987); especially in leafy greens (Nagy and Wardowski, 1988) Light also plays a crucial role in the formation of β-carotene in tomatoes (Raymundo et al., 1967). Additionally, tomatoes grown in full sunlight have been shown to contain higher levels of sugar and dry matter than those grown under shade (Winsor, 1979). Temperature is another important climactic factor that influences postharvest quality. For example, Lettuce is often produced in areas with cool days and nights which results in optimum lettuce quality; having firm heads and mild flavor. However, if lettuce is exposed to high temperatures during production, bitter flavor and loss of tenderness can result (Pierce et al., 1987). Perhaps the most important effect of temperature on growth and development is that it dictates the initiation of the reproduction cycle (Beverly et al., 1993). Flower initiation often depends on temperature; differences in day and nighttime temperatures have been shown to regulate flower stalk initiation as well as stem elongation (Agrawal et al., 1993). The initiation of flowering or bolting in cole crops is highly undesirable and can be triggered by prolonged exposure to cool weather as days increase in length (Peirce et al., 1987).

During the production season, fruit and vegetable producers employ a variety of selected cultural practices with the goal of maximizing yields and optimizing crop quality. This encompasses a broad range of practices given the diversity fruits and vegetables that are commercially produced (Weston and Barth, 1997). Pre-harvest cultural practices involving irrigation and nutrient management have been shown to greatly influence the postharvest quality and shelf life of many vegetable commodities. Nitrogen availability is linked to carotenoid and protein biosynthesis and influences protein/carbohydrate ratios (Mengel, 1979). Sufficient soil nitrogen can result in improved quality by encouraging the development of photosynthetic surface area. The application of nitrogen fertilizer has been shown to improve the head quality of broccoli (Dufalt, 1988). Conversely excess soil nitrogen has been shown to cause the accumulation of potentially hazardous levels of nitrate adversely affecting the nutrient quality of spinach (Maynard 1984). Water availability is another important cultural practice that affects postharvest quality. Tomatoes, peppers and other vegetables have shown increased vitamin C content with the application of a drip irrigation regiment (Cevik, 1981). Growing spinach under high rainfall conditions reduced its storage potential by 40% compared to spinach grown under normal rainfall conditions (Johnson et al., 1989). The maturity of a commodity at the time of harvest can also have an influence on its quality and relative postharvest storage potential (Weston and Barth, 1997). Optimal maturity varies is determined by the type of commodity and it use. For example, the stage of maturity considered optimal for fresh or frozen produce may be different for canning the same commodity (Salunkhe et al., 1991). Maturity also influences that nutritive value of a commodity. For example, small cabbage heads have higher levels of ascorbic acid than large ones, and small turnip leaves contain more thiamin and riboflavin but less carotene than large leaves (Salunkhe et al., 1991).

In addition to climactic factors and cultural practices, cultivar selection can also influence postharvest quality and storage potential of a commodity (Kader, 2002). Cultivars vary in genetic makeup and as a result, will vary in attributes such as size, color, texture, flavor, disease and pest resistance, storage potential, and yield (Beverley et al., 1993). Therefore, choosing the appropriate genotype for a given environment and production system can reduce the incidence and severity of physiological disorder, decay, insect damage, and unmarketable attributes (Kader, 2002). Cultivars may also vary greatly in nutritional quality (Kader, 2002; Weston and Barth, 1997). For example, Takahata et al. (1993) reported variation in the β-Carotene content of several cultivars of sweetpotato: ‘Georgia Jet’, ‘Murasakibaru’, and ‘Tokai 5’. All three cultivars had similar amino acid composition and relatively high fructose and glucose content. However, ‘Georgia Jet’ contained low concentrations of the β-Carotene ($6.9 mg/100g^{-1}$), the precursor to vitamin A.

Sweetpotato slip production in the U.S. is relatively homogenized in practice at the commercial level. Nearly all large-scale producers employ the same recommended bedding practices for slip production described by (Smith et al., 2009 and Wilson et al., 1977). Some sweetpotato cultivars such as ‘Beauregard’, ‘Evangeline', and ‘Orleans’ exhibit a growth habit that results in uniform slip sizes as the growing points in these varieties are located near the canopy at the time the slips are harvested (Thompson, et al., 2014). However, the variety ‘Covington’ unlike most commercial cultivars, has a growth habit in which the top canopy leaves are mostly at the same height, but location of growing points can vary from being close to the top of the leaf canopy to several cm below (Thompson et al., 2014). Uniform slip size could be advantageous especially for interfacing with mechanical planters. The lack of information regarding preharvest factors affecting the postharvest quality of sweetpotato slips and the relative homogeneity of production practices leaves cultivar selection as the primary controllable preharvest factor worth consideration.

Harvesting of fresh fruits and vegetables is accomplished by hand, by mechanically assisted devices or by mechanical harvesters depending on the commodity (Shewfelt et al., 2014). Factors during harvest that can influence postharvest quality include the degree of mechanical damage caused by human or machine, the accuracy of selecting acceptable fruit or vegetables without defects, the time of day of harvest, and the pulp temperature at harvest. Hand harvesting has several advantages over mechanical harvesting methods (Thompson, 2002). Human harvesters can quickly and accurately determine quality and maturity of a product in the field. Determining maturity in the field is of importance for commodities that are harvested multiple times and at different stages of maturity (Thompson, 2002). One example of a commonly hand harvested commodity is sweet cherries. Sweet cherries are harvested by hand because they are highly susceptible to bruising and are harvested at peak ripeness. The disadvantage to manual, hand-harvesting is that it requires skilled, seasonal labor and can be very time consuming and expensive (Michailides and Manganaris, 2009; Prusky, 2011). Mechanical harvest is time-efficient and is often less costly than manual labor but lacks the ability to identify desired maturity. The primary disadvantage to mechanized harvesting is mechanical damage incurred during harvest process. Damage occurring at any time during harvest, handling, or transport can majorly contribute to the postharvest deterioration of a commodity (Thompson, 2002). Browning of damaged tissue results from membrane disruption, which exposes phenolic compounds to the polyphenol oxidase enzyme. Injury damaged if incurred accelerates the rate of water loss, provides sites of easy access for pathogens, and stimulates the production of CO_2 and C_2H_4 (Thompson, 2002).

Changes that occur after harvest can't be stopped but can be decelerated to an extent with proper postharvest handling practices. An "aberrant change in physiological processes brought about by one or a combination of environmental or biological factors" is known as the stress response (Hale and Orcutt, 1987). Temperature extremes, water loss, invasion by spoilage pathogens, gaseous atmosphere, light and physical damage can all induce stress in harvested fruits or vegetables (Shewfelt et al., 2014). The sweetpotato slips produced commercially in the U.S. are propagates and are not typically consumed as greens. As a result, postharvest practices are minimal. For that reason, the two most important postharvest factors affecting sweetpotato slips: temperature management and water loss will be outlined in the following sections.

Quality and Temperature Management

Temperature management is the most important tool for maintaining postharvest freshness and quality of horticultural commodities (Kader, 2013). Refrigerated storage is highly beneficial because it serves to slow the rate of deterioration due to ripening, softening, and spoilage pathogens, as well as textural/color changes and metabolic processes (Hardenburg et al, 1986). Sweetpotato slips are tropical in origin and similar to fresh leafy herbs like basil (*Ocimum basilicum*) and can't be stored at less than 10°C without deterioration due to chilling injury (Mitcham et al. 2001). Lang and Cameron (1994) determined that fresh sweet basil maintained visual quality for an average of 12 days at 5°C. They also reported that symptoms of chilling injury were severe at 0 and 5°C and below, decreasing shelf life to 1-3 days. Moderate chilling injury was also observed at 7.5 and 10°C (Lang and Cameron 1994). Additionally, it has been reported that *I. aquatica* is chilling sensitive with a recommended storage temperature of around 12 °C (Gross et al., 2004)

The prolonged exposure of chilling-sensitive plants to temperatures above freezing but less than 10°C results the disturbance of all physiological processes; water re-gime, mineral nutrition, photosynthesis, respiration and metabolism (Lukatkin et al., 2012). Inactivation of metabolism observed at chilling of chilling-sensitive plants is a complex function of both temperature and duration of exposure. Response of plants to low temperature exposure is associated with a change in the rate of gene transcription of low molecular weight protein molecules (Lukatkin et al., 2012). Leaves that been affected by chilling injury can appear reddish brown and wilt quickly after removal from low temperature.

Quality and Water Loss

Sweetpotato slips are uprooted propagates. One of the challenges of producing plants through vegetative propagation is wilting and death before or shortly after transplant (Alem, 2010). One of the possible causes of propagate wilting, is failure to take up enough water through the stem or leaves after being severed from the stock plant (Loach and Whalley, 1978). Harvested sweetpotato slips are like leafy green vegetables in that they consist of leaves, shoots, and stems. Leafy green vegetables are composed primarily of water (>90%) (Acedo and Weinberger, 2007).

Due to high water content, high surface to volume ratios and the presence of stomata leaf tissue, leafy green vegetables are extremely vulnerable to high rates of water loss (O'Hare et al., 2001). Consequently, water loss leads to wilting, shriveling, and loss of crispness, and firmness (Ben-Yehoshua and Rodov, 2003).

Water loss is the primary cause of poor quality and postharvest loss in leafy green vegetables such as lettuce, chard, cabbage, spinach, and green onion (Ben-Yehoshua and Rodov, 2003). Leafy greens can appear wilted after just 3-5 percent water loss (Holcroft, 2015). For example, leaf lettuce can lose a maximum of 3-5 percent of its water weight before its determined to be unmarketable (Thompson et al., 2008), whereas spinach can lose only 3% of its weight before it is considered unmarketable (Kays and Paull, 2004). Watercress can lose up to 7 percent water weight and cabbage 6-11 percent before becoming unmarketable (Kays and Paull, 2004; Thompson et al., 2008).

Water loss is caused by transpiration which involves the transport of water, primarily through the stomates of the leaves, and the subsequent evaporation of this moisture from the surface of the commodity to the surrounding environment (Holcroft, 2015). Stomates consist of two guard cells that form a small pore on the surfaces of leaves. The guard cells control the opening and closing of the stomates in response to various environmental stimuli. Water loss through transpiration affects the physiological, and metabolic processes of the harvested commodity causing reduced photosynthetic capacity resulting in stomatal closure, metabolic limitations and oxidative damage to chloroplasts (Muhammad et al, 2009).

To prevent water loss and maintain freshness after harvest, it is essential that in addition to temperature and humidity management, proper packaging is used for leafy green vegetables (Bautista and Acedo, 1987). Appropriate packaging for leafy greens and herbs should reduce physical injury during transit and handling, provide adequate ventilation to hasten cooling and allow heat from respiration to escape, but also act as a barrier to prevent water loss (Gast, 1991). High-density plastic films have been shown to greatly inhibit water loss in fresh produce, especially leafy vegetables (Ben-Yehoshua, 1978). O'Hare et al. reported in a 2001 study that moisture loss was effectively reduced by packaging pak choi in plastic film and proved to be more effective than manual misting or anti-transpirant treatments. Prevention of water loss is one of the primary purposes of packaging fresh leafy greens and herbs in plastic for retail sales (Hruschka and Wang, 1979). Aharoni et al.1989, demonstrated that the quality of a variety of salad herbs (chives, chervil, coriander, dill, sorrel, and watercress) stored for 5 days at 6°C and then 2 days at 12°C was improved by the addition of polyethylene liners in the storage cartons. *Ipomoea aquatica*, also known as water spinach, or swamp cabbage is an herbaceous aquatic or semi-aquatic perennial plant that like sweetpotato, is part of the morning glory family, Convolvulaceae (Worldcrops.org, n.d.). Due to the high content of water in leaves, *I. aquatica* undergoes rapid rates of water loss after harvest resulting in a short shelf life of only a few hours at ambient temperature (Hu et al., 2015).

Polyamide also known as nylon, is a clear and printable thermoplastic that has a relatively high melting point, exceptional strength and toughness, and good oxygen barrier properties. It is also scratch, puncture, and flex-crack resistant and does not dissolve/absorb grease, oil, or acidic food. These properties make Polyamide ideal for use in conventional and microwave cooking applications. Polyamide is mainly used as a flexible packaging film for food sensitive to oxygen and is chosen when high mechanical strength, high melting point, transparency, and good oxygen barrier is required. Important food packaging applications include processed meat, smoked fish, cheese and other dairy products, and par-cooked microwavable foods (Polymerdatabase.com, n.d.). In our trials we used microperforated nylon (polyamide) film as a liner to test its influence on water loss during storage. Micro-perforation allows for in-package control of CO_2 and O_2 concentration. Microperforated films have been used have been have used to preserve the quality of a range of fresh vegetables such as broccoli, green, onion, cucumber, strawberries, and sweetcorn (Aharoni et al., 1997). In preliminary test runs of our nylon liner we observed that sealing sweetpotato slips liner increased the rate of senescence, therefore, our liner was wrapped around the slips but left open at the ends prior to being places in a waxed box for storage.

Yellowing is a physiological phenomenon related to chlorophyll degradation, which results in dramatic declines of leaf greenness (Hue et al., 2011). Postharvest yellowing occurs when leafy organs are stressed due to high light, water loss, high temperature, or when they cease to produce carbohydrates (Cantwell and Reid, 1993). The cells of the leaf initiate a catabolic process which is designed to recover important nutrients for reutilization in other plant parts. Hydrolysis of proteins and disassembly of the chloroplast apparatus are accompanied by decreased photosynthetic activity and increases in respiration and ethylene production (Cantwell and Reid, 1993). Losses in membrane integrity and the related capacity to maintain ionic balance lead to increased yellowing and ultimately senescence (Cantwell and Reid, 1993).

Sweetpotato Slip Transportation

. Most slip producers in the U.S. rely on the mail (USPS), or other package delivery services (UPS, FedEx) (personal communication) which are not temperature controlled. Slip producers exhibit a variety practices in prepping slips for transport, some of which are proprietary. For example, Victory Seed Company in Molalla, Oregon ships out small, bound bundles of slips with the root end wrapped in strips of newspaper. The bundle is wrapped again in parchment paper and placed in a cardboard box for shipping (Victory Seed Co., 2018, personal contact). Similarly, the Sandhill Preservation in Calamus, Iowa ships out slips in small bundles with the root end wrapped in newspaper, then placed in a small plastic bag. The whole bundle is subsequently wrapped in newspaper and placed in a box for shipping (Sandhill Preservation, received order). One unique practice is the use of sphagnum moss as a packing material. Steel Plant Company in Gleason, Tennessee ships out bound bundles of 25 slips with the roots packed in rehydrated sphagnum moss. The Bundle is subsequently wrapped in parchment paper and shipped out (Steele Plant Co., 2018, personal contact). These shipping practices are more common amongst retailers who are selling to small scale growers and home gardeners, or who specialize in rare and heirloom cultivars. Wrapping the slips in various types of material before packing could help prevent water loss throughout the shipping process, preserving quality. Conversely, Jones Family Farm in Baily, North Carolina ships out slips that are placed unbound into waxed produce boxes; approximately 1000 slips/bushel box (Jones Family Farm, 2018, personal contact). This is the same practice that the John C. Pair Horticultural Center in Hayesville, KS implements for shipping. The primary differences in these various shipping practices is reflective of the volume of slips that are being sold and shipped. Operations who are supplying small amounts of slips to home gardeners take more steps in the packing process to ensure a quality product. Wrapping up bundles of 25 slips may not be feasible for larger scale producers who are shipping 1000's of slips at a time.

Research Objectives

Despite slips commonly being shipped from region to region within the U.S., there is an evident lack of standardized practices and recommendations for shipping and storage of sweetpotato slips. Furthermore, several days in shipping combined with current shipping practices could result in poor quality slips. Producers and distributors employ a variety of practices in preparing slips for shipping that seem to be reflective of the size of the operation, and number of slips being purchased. Some producers who sell rare and heirloom cultivars have adapted more specialized preparation practices for shipping small amounts of slips. Moreover, even less is known about the postharvest physiology and handling of sweetpotato slips. In their 2017 research, Thompson et al., evaluated the main effect of holding slips in storage for various days before planting. the 1 and 3 DBP (days before planting) storage treatments produced the greatest yields (Thompson et al., 2017). 1 DBP produced significantly greater yields (45.7 MT ha-1) than the slips planted on day of harvest (42.4 MT ha-1), 5 DBP (41.5 MT ha-1), and 7 DBP (40.4 MT ha-1) treatments. The 3 DBP treatment resulted in intermediate marketable yields (42.6 MT ha-1) that were comparable to all holding treatments but was numerically higher than the DOP, 5, and 7 DBP holding treatments. To our knowledge, no formal postharvest research has investigated slip quality changes after harvest and before planting especially regarding optimal storage temperature and shipping. Understanding the postharvest behavior of slips could help slip producers deliver a more consistent, high quality product. This information could apply to both organic and conventionally grown slips, as well as slips grown either in the open field or high tunnels. Our overall objective was to investigate the postharvest behavior and handling of sweetpotato slips during transportation and storage. More specifically we wanted to:

- Identify the optimum storage and transport conditions related to temperature and packaging for sweetpotato slips, that would maintain the slip quality.
- Investigate the influence of slip storage duration on slip establishment, growth and storage root yield

Bibliography

Acedo, A.L., Weinberger, K. Best Practices in Postharvest Management of Leafy Vegetables in Greater Mekong Subregion Countries. Proceedings of a GMS workshop 25-27 October 2007 Hanoi, Vietnam.

Agrawal, Madhoolika & Krizek, Donald & Agrawal, Shashi & Kramer, George & Lee, Edward & Mirecki, Roman & Rowland, Randy. 1993. Influence of Inverse Day/Night Temperature on Ozone Sensitivity and Selected Morphological and Physiological Responses of Cucumber. Journal of The American Society for Horticultural Science. 118. pp.649-654. doi:10.21273/JASHS.118.5.649

Agricultural Marketing Resource Center (AgMRC), 2017. Sweet Potatoes [WWW Document]. URL https://www.agmrc.org/commodities-products/vegetables/sweet-potatoes/ (accessed 2.13.19).

Aharoni N., Dvir, O., Reuveni, A., 1989. Modified Atmospheres in Film Packages Delay Senecsence and Decay of Fresh Herbs. Acta Hort 258: pp. 255-262.

Alem, P.O., 2010. Unrooted stem cutting physiology; water use and leaf gas exchange of severed stem cuttings, Clemson University.

Austin, D. F. 1988. The taxonomy, evolution and genetic diversity of sweetpotatoes and related wild species. In P. Gregory (ed.) Exploration, Maintenance and Utilization of Sweetpotato Genetic Resources. CIP, Lima, Peru, pp. 27–60.

Bautista, O.K., Acedo, A.L. 1987. Postharvest Handling of Fruits and Vegetables. Manila: National Book Store Inc. Tech guide Series No. 4. p. 24.

Barkley, S.L., 2015. Optimizing Cultural Management Practices and Consumer Acceptance of Sweetpotato in North Carolina. North Carolina State.

Barkley, S.L., Chaudhari, S., Schultheis, J.R., Jennings, K.M., Bullen, S.G., Monks, D.W., 2017a. Optimizing Sweetpotato Seed Root Density and Size for Slip Production. Horttechnology 27, pp. 7–15. https://doi.org/10.21273/HORTTECH03435-16

Barkley, S.L., Schultheis, J.R., Chaudhari, S., Johanningsmeier, S.D., Jennings, K.M., Truong, V.-D., Monks, D.W., 2017b. Yield and Consumer Acceptability of Evangeline Sweetpotato for Production in North Carolina. Horttechnology 27, pp. 281–290. https://doi.org/10.21273/HORTTECH03533-16

Ben-Yehoshua, S. 1978. Delaying deterioration of individual citrus fruit by seal-packaging in film of high-density polyethylene. I. General effects, In: Proceedings of the International Society of Citriculture. pp. 110-115.

Ben-Yehoshua, S. and V. Rodov. 2003.Transpiration and water stress. In: J.A. Bartz and J.K. Brecht. (Eds.). Postharvest Physiology and Pathology of Vegetables. 2nd ed., Marcel Dekker, Inc., NY. pp. 111-159.

Beverly, R.B., Latimer, J.G., Smittle, D.A.1993. Preharvest Physiology and Cultural Effects on Postharvest Quality.In: R.L. Shewfelt and S.E. Prussia (eds) Postharvest Handlig: A Systems Approach. Academic, New York pp. 73-98.

Bond, J., 2017. U.S. Sweetpotato Production Swells | USDA [WWW Document]. URL https://www.usda.gov/media/blog/2017/01/5/us-sweet-potato-production-swells (accessed 10.18.19).

Boudreaux, J. E., Cannon, J. M., and Villordon, A. 2005. Sweetpotato, pp. 54–56. In J. E. Boudreaux (ed.) Commercial Vegetable Production Recommendations. LSU Ag Center Publication, No. 2433, www.lsuagcenter.com.

Bovell-Benjamin, A.C., 2007. Sweetpotato: A Review of its Past, Present, and Future Role in Human Nutrition. Advances in food and nutrition research. 52. pp. 1-59. doi:10.1016/S1043-4526(06)52001-7

Brain, R., 2012. The Local Food Movement: Definitions, Benefits & Resources. Utah State Univ. Ext.

Cantwell, M., and Reid M., 1993. Postharvest Physiology and Handling of Fresh Culinary Herbs, Journal of Herbs, Spices & Medicinal Plants, 1:3, pp. 93-127, doi:10.1300/J044v01n03_09

Carpena, A.L., 2009. Important Cultivars, Varieties, and Hybrids, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 27–40. https://doi.org/10.1007/978-1-4020-9475-0_4

Cevik, B., Kirda, C., Ding, G., 1981. Some effect of irrigation systems on yield and quality of tomato grown in plastic covered greenhouse in the south of Turkey. Acta Hort. 119: pp. 333-342.

Clark, C.A., Holmes, G.J., Ferrin, D.M., 2009. Major Fungal and Bacterial Diseases, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 81–103. https://doi.org/10.1007/978-1-4020-9475-0_7

Coolong, T., Seebold, K., Bessin, R., Woods, T., Fannin, S., 2012. Sweetpotato Production for Kentucky. University of Kentucky College of Agriculture Cooperative Extension Service, Lexington, KY.

Coomes, O.T., McGuire, S.J., Garine, E., Caillon, S., McKey, D., Demeulenaere, E., Jarvis, D., Aistara, G., Barnaud, A., Clouvel, P., Emperaire, L., Louafi, S., Martin, P., Massol, F., Pautasso, M., Violon, C., Wencélius, J., 2015. Farmer seed networks make a limited contribution to agriculture? Four common misconceptions. Food Policy 56, pp. 41–50. https://doi.org/10.1016/J.FOODPOL.2015.07.008

Crop Trust, n.d. Sweetpotato – Crop Trust [WWW Document]. URL https://www.croptrust.org/crop/sweet-potato/ (accessed 10.9.18).

Di Noia, J., 2014. Defining Powerhouse Fruits and Vegetables: A Nutrient Density Approach. Prev. Chronic Dis. 11, E95. https://doi.org/10.5888/pcd11.130390

Drapal, M & Fraser, P. 2019, 'Metabolic diversity in sweetpotato (Ipomoea Batatas, Lam.) leaves and storage roots', Horticulture Research, vol. 6, 2, pp. 1-9. https://doi.org/10.1038/s41438-018-0075-5

Dufalt, R. 1988. Nitrogen and phosphorus requirements for greenhouse broccoli production. HortScience 23: pp. 576-578.

Edmond, J.B., 1971. Taxonomy, History and Industry, in: Edmond, J.B., Ammerman, G.R. (Eds.), Sweetpotatoes: Production, Processing and Marketing, Major Feed and Food Crops in Agriculture and Food Series. Avi Pub. Co., Westport, Conn., p. 334.

Elmer, O.H., 1938. SWEETPOTATOES IN KANSAS. Manhattan.

Estes, E.A. 2009 Marketing Sweetpotatoes in the United States: a serious challenge for small-to-moderate volume growers. In: Loebenstein, G. and Thottappilly, G. (eds) The Sweetpotato Springer Science + Business Media BV, Houten, The Netherlands, pp. 269–283.

Ewell, P. T., 1990. Sweetpotato in Eastern and Southern Africa. Paper presented at 'Sweetpotato in the Food Systems of Eastern and Southern Africa' workshop. Nairobi, Kenya.

Firon, N., Labonte, D., Villordon, A., McGregor, C., Kfir, Y., Pressman, E., 2009. Botany and physiology: Storage root formation and development, in: The Sweetpotato. pp. 13–26. https://doi.org/10.1007/978-1-4020-9475-0_3

Flaccavento, A., Williams, M., Shuman, M., 2014. Food Hub Feasibility Study: Northeast Kansas.

Food and Agriculture Organization of the United Nations (FAO), 2017. Value of Agricultural Production [WWW Document]. Food Agric. Organ. United Nations. URL http://www.fao.org/faostat/en/#data/QV (accessed 10.17.18).

Gast, K.L.B. 1991. Containers and Packaging Fruits & Vegetables, Kansas State University, March 1991. http://www.oznet.ksu.edu.

George, N.A., Pecota, K. V, Bowen, B.D., Schultheis, J.R., Yencho, G.C., 2011. Root Piece Planting in Sweetpotato—A Synthesis of Previous Research and Directions for the Future. HortTechnology 21, 703–711.

Godfray, H.C.J., Beddington, J.R., Crute, I.R., Haddad, L., Lawrence, D., Muir, J.F., Pretty, J., Robinson, S., Thomas, S.M., Toulmin, C., 2010. Food security: the challenge of feeding 9 billion people. Science 327, 812–8. https://doi.org/10.1126/science.1185383

Gray, L.C., Thompson, E.K., 1933. History of Agriculture in the Southern United States to 1860, No. 439. ed. The Carnegie Institution of Washington.

Greater Kansas City Food Hub Working Group, 2015. Kansas City Food Hub Feasibility Study 1–36.

Gross, K.C., Wang, C.Y., Saltviet, M.E. 2004. The commercial storage of fruits, vegetables and florist and nursery stocks. USDA Agricultural Handbook 66, U.S. Dept. of Agriculture, Washington.

Gurmu, F., Shimels, H., Laing, M., 2013. Self and cross-incompatibilities in sweetpotato and their implications on breeding. Australian Journal of Crop Science. 7. 1854-1860.

Hale, M.G., Orcutt, D.M. 1897. The Physiology of Plants under Stress, John Wiley & Sons, New York.

Hattori, T., Nakagawa, S. & Nakamura, K., 1990. High-level expression of tuberous root storage protein genes of sweet potato in stems of plantlets grown in vitro on sucrose medium. Plant Mol Biol 14: pp. 595-604. https://doi.org/10.1007/BF00027505

Holcroft, D. 2015. Water Relations in Harvest Fresh Produce. The Postharvest Foundation White Paper No. 15-01. ISBN978-1-62027-005-9.

Hossain, M.M. and Mondal, M.A.A., 1994. Effect of vine parts on the growth and yield of three sweetpotato varieties. Bangladesh J. Sci. Ind. Res. 29, pp. 181–184.

Hruschka HW, Wang CY. Storage and Shelf Life of Packaged Watercress, Parsley, and Mint. 1979. U.S. Dept. of Agriculture, Mktg. Res. Rept. 1102.

Huang, J. C. and Sun, M., 2000. Genetic diversity and relationships of sweetpotato and its wild relatives in Ipomoea series batatas (Convolvulaceae) as revealed by inter-simple sequence repeat (ISSR) and restriction analysis of chloroplast DNA. Theor. Appl. Genet. 100:1050–1060.

Hue, S.M., Boyce, A.N., Somasundram, C. (2011) Influence of growth stage and variety on the pigment levels in Ipomoea batatas (sweetpotato) leaves. African Journal of Agricultural Research, 6 (10). pp. 2379-2385. ISSN 1991637X.

International Potato Center (CIP), 2010. Facts and Figures About Sweetpotato. [WWW Document]. URL https://nkxms1019hx1xmtstxk3k9sko-wpengine.netdna-ssl.com/wp-content/uploads/PDF/005448.pdf (accessed 10.10.18).

International Potato Center (CIP), n.d.a. Sweetpotato Processing and Uses. [WWW Document]. URL https://cipotato.org/crops/sweetpotato/sweet-potato-processing-and-uses/ (accessed 6.17.19).

International Potato Center (CIP), n.d.b. Sweetpotato. [WWW Document]. URL https://cipotato.org/crops/sweetpotato/sweetpotato-cont/ (accessed 6.17.19).

Johnson, J.R., McGuinn, J.R., Rushing, J.W. 1989. Influence of preharvest factors on postharvest quality of prepackaged fresh market spinach. Appl. Agr. Res. 4: pp. 141-143.

Jones, A. 1967. Theoretical segregation ratios of qualitatively inherited characters for hexaploidy sweetpotato (*Ipomoea batatas*). USDA Tech. Bul. No. 1368.

Kader A.A., 1987. Influence of preharvest and postharvest environment on nutritional composition of fruits and vegetables. Proc. 1st Int. Symp. Hort. & Human Health. ASHS Symp. Ser. 1: pp. 18-32.

Kader, A, A., 2002. Chapter 4: Postharvest Biology and Technology: An Overview. In A.A. Kader A.A., Postharvest Technology of Horticulture Crops. University of California Agriculture and Natural Resources, Publication # 3311.

Kays, S.J. & Paull, R.E. 2004. Postharvest biology. Exon Press, Athens, GA

Lareo, C., Ferrari, M.D., Guigou, M., Fajardo, L., Larnaudie, V., Ramírez, M.B., Martínez-Garreiro, J., 2013. Evaluation of sweetpotato for fuel bioethanol production: hydrolysis and fermentation. Springerplus 2, 493. https://doi.org/10.1186/2193-1801-2-493

Lim, S., Xu, J., Kim, J., Chen, T., Su, X., Standard, J., Carey, E., Griffin, J., Herndon, B., Katz, B., Tomich, J. and Wang, W., 2013. Role of anthocyanin-enriched purple-fleshed sweetpotato p40 in colorectal cancer prevention. Mol. Nutr. Food Res., 57: 1908-1917. doi:10.1002/mnfr.201300040

Loach, K. and D.N. Whalley. 1978. Water and carbohydrate relationships during the rooting of cuttings. Acta Hort., 79: pp.161-168

Loebenstein, G., Thottappilly, G., Fuentes, S., Cohen, J., 2009. Virus and Phytoplasma Diseases, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 105–134. https://doi.org/10.1007/978-1-4020-9475-0_8

Low, J., Lynam, J., Lemaga, B., Crissman, C., Barker, I., Thiele, G., Namanda, S., Wheatley, C., Andrade, M., 2009. Sweetpotato in Sub-Saharan Africa, in: The Sweetpotato. pp. 359–390. https://doi.org/10.1007/978-1-4020-9475-0_16

Low, J.W., Mwanga, R.O.M., Andrade, M.I., Carey E.E., Ball A.M. Tackling vitamin A deficiency with biofortified sweetpotato in sub-Saharan Africa, Global Food Security, Volume 14, 2017, pp. 23-30, ISSN 2211-9124, https://doi.org/10.1016/j.gfs.2017.01.004

Lowe, S.B., Wilson, L.A., 1974. Comparative Analysis of Tuber Development in Six Sweetpotato (Ipomoea batatas (L.) Lam) Cultivars: 1. Tuber initiation, tuber growth and partition of assimilate. Ann. Bot. 38, 307–317.

Maynard, D.N. 1984. Fruit and vegetable quality as affected by nitrogen nutrition. Proc. Amer. Soc. Agron. 617-625

Mengel, K. 1979. Influence of exogenous factors on the quality and chemical composition of vegetables. Acta Hort. 93:133-151.

Muhammad, F., A. Wahid, D.J. Lee, O. Ito, and H. Kadambot. 2009. Advances in drought resistance of rice; critical reviews in Plant Sci., 28:199-217.

Mwanga, R.O.M., Andrade, M.I., Carey, E.E., Low J.W., Yencho G.C., Grüneberg W.J., 2017. Sweetpotato (*Ipomoea batatas* L.). In: Genetic Improvement of Tropical Crops. Springer, pp. 182-218. https://doi.org/10.1007/978-3-319-59819-2_6

Nagy, S. Wardowski, W.F. 1988. Effects of agriculture practices, handling and storage on fruits. In: E. Karmas and R.S. Harris (eds.). Nutritional evaluation of food processing. Van Nostrand Reinhold, New York.

O'Brien, P.J., 1972. The Sweetpotato: Its Origin and Dispersal. Am. Anthropol. 74, pp. 342–365. https://doi.org/10.2307/671520

O'Hare, T.J., Able, A.J., Wong, L.S., Prasad, A. and McLauchlan, R. 2001. Fresh-cut Asian Vegetables-Pak Choi as a Model Leafy Vegetable. Postharvest handling of fresh vegetables. Proceedings of a workshop in Beijing, P.R.C., 9-11 May 2001.

Padmaja, G., 2009. Uses and Nutritional Data of Sweetpotato, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 189–234. https://doi.org/10.1007/978-1-4020-9475-0_11

Peirce, L.C. 1987. Vegetables: Characteristics, production and marketing. John Wiley and Sons, New York.

Purseglove, J. W. 1968. Tropical crops. Dicotyledons. I. John Wiley and Sons, Inc., New York.

Raymundo, L. C., Griffiths, A. E., Simpson, K. L., 1967. Dimethyl sulfoxide and biosynthesis of carotenoids in detached tomatoes. Phytochemistry. 6, 1527 (1967).

Reuter, T.A., 2017. Seeds of Life, Seeds of Hunger. http://journals.openedition.org/aof

Roullier, C., Kambouo, R., Paofa, J., McKey, D., Lebot, V., 2013. On the origin of sweetpotato (Ipomoea batatas (L.) Lam.) genetic diversity in New Guinea, a secondary centre of diversity. Heredity (Edinb). 110, 594–604. https://doi.org/10.1038/hdy.2013.14

Salunkhe, D.K., Bolin, H.R., Reddy, N.R. 1991. Preharvest factors on postharvest yield and quality. Vol. 1, pp. 7-44 In: Storage, processing, and nutritional quality of fruits and vegetables. CRC Press, Boca Raton.

Spooner, D.M. 2013. Solanum tuberosum (potato). Encyclopedia of Genetics, 2nd Edition, vol. 6, pp. 481-483, eds. S. Maloy & K. Hughes, Elsevier.

Sprinkle, D.,Packaged Facts, 2015. Sales of Local Foods Reaches $12 Billion. [WWW Document]. URL https://www.packagedfacts.com/about/release.asp?id=3717 (accessed 10.7.18).

Saglam, M., Sintim, H.Y., Bary, A.I., Miles, C.A., Ghimire, S., Inglis, D.A., Flury, M., 2017. Modeling the effect of biodegradable paper and plastic mulch on soil moisture dynamics. Agric. Water Manag. 193, 240–250. https://doi.org/https://doi.org/10.1016/j.agwat.2017.08.011

Schultheis, J. R., Boudreaux, J. E., Shankle, M.W., Smith, Tara P., et al. 2008. In G. J. Holmes and J. M. Kemble (eds.) Vegetable Crop Handbook for the Southeastern US (Sweetpotato Section). North Carolina Vegetable Growers Association, North Carolina.

Scott, G.J., Best, R., Rosegrant, M. and Bokanga, M. 2000a. Roots and tubers in the global food system: A vision statement for the year 2020. International Potato Center, Lima, Peru, p. 111.

Scott, G.J., Rosegrant, M.W. and Ringler, C. 2000b. Roots and tubers for the 21st century. Trends, projections, and policy options. Food, agriculture, and the environment, Discussion paper 31. International Food Policy Research Institute and Centro Internacional de la Papa, p. 64.

Shewfelt, R.L., Prussia, S.E., Sparks, S.A. 2014. Chapter 2Challenged in Handling Fresh Fruits and Vegetables. Postharvest Handling: A Systems Approach 3rd Edition. ISBN:978-0-12-408137-6, 2014 Elsevier Inc., https://doi.org/10.1016/C2012-0-03264-4

Shiva, V., 2016. Seed sovereignty, food security: women in the vanguard of the fight against GMOs and corporate agriculture.

Smith, T.P., Stoddard, S., Shankle, M., Schultheis, J., 2009. Sweetpotato Production in the United States, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 287–323. https://doi.org/10.1007/978-1-4020-9475-0_14

Spooner, D.M. 2013. Solanum tuberosum (potato). Encyclopedia of Genetics, 2nd Edition, vol. 6, pp. 481-483, eds. S. Maloy & K. Hughes, Elsevier.

Sprinkle, D.,Packaged Facts, 2015. Sales of Local Foods Reaches $12 Billion. [WWW Document]. URL https://www.packagedfacts.com/about/release.asp?id=3717 (accessed 10.7.18).

Stoddard, C.S., 2013. SWEETPOTATO PRODUCTION IN CALIFORNIA PRODUCTION AREAS AND SEASONS.

Takahata, Y., Noda T., Nagata T. 1993. Varietal differences in chemical composition of the sweetpotato storage root. Acta Hort. 343: pp. 77-80

Thompson, J.F., Mitchell, F.G., Rumsey, T.R., Kasmire, R.F., Crisosto, C.H. 2008. Commercial Cooling of Fruits, Vegetables, and Flowers. University of California, Agriculture and Natural Resources, Pub. 21567.

Thompson, J.F., Mitchell, F.G., Kasmire, R.F. 2002. Cooling horticultural commodities. Postharvest Technology of Horticultural Crops. Pp. 97-112.

Thompson, W.B., 2014. Sweetpotato Transplant Establishment Considerations for Sweetpotato Production in North Carolina. North Carolina State.

Thompson, W.B., Schultheis, J.R., Chaudhari, S., Monks, D.W., Jennings, K.M., Grabow, G.L., 2017a. “Covington” Sweetpotato Plant Survival and Yield Response to Preplant Irrigation, Planting Depth, and Transplant Size. HortTechnology 27, pp.824–830. https://doi.org/10.21273/HORTTECH03815-17.

Thompson, W.B., Schultheis, J.R., Chaudhari, S., Monks, D.W., Jennings, K.M., Grabow, G.L., 2017b. Sweetpotato Transplant Holding Duration Effects on Plant Survival and Yield. HortTechnology 27, pp. 818–823. https://doi.org/10.21273/HORTTECH03808-17

Thottappilly, G., 2009. Introductory Remarks, in: Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 3–7. https://doi.org/10.1007/978-1-4020-9475-0_1.

Togari, Y., 1950. A study in the tuberous-root formation of sweetpotato. Bull. Natl. Agric. Exp. Stn. 68, pp. 1–96.

USDA Economic Research Service (ERS), 1994. Sweetpotatoes: An Economic Assessment of the Feasibility of Providing Multiple-Peril Crop Insurance

USDA Economic Research Service (ERS), n.d. U.S. Sweetpotato Statistics [WWW Document]. URLhttp://usda.mannlib.cornell.edu/MannUsda/viewDocumentInfo.do?documentID=1492 (accessed 11.10.18).

USDA Economic Research Service (ERS), 2015. Vegetables and Pulses Outlook: Special Article Commodity Highlight: Sweet Potatoes.

USDA National Agricultural Statistics Service (NASS), 2012. 2012 Census of Agriculture - UNITED STATES DATA, Table 38. Vegetables, Potatoes, and Melons Harvested for Sale: 2012 and 2007.

USDA National Agricultural Statistics Service (NASS), 2018. United States Department of Agriculture National Agricultural Statistics Service Crop Production 2017 Summary

Vineland Research and Innovation Centre, 2017. 2017 Sweet Potato Slip Production Research Update.

Weston, L.A., Barth, M.M., 1997. Preharvest factors affecting postharvest quality of vegetables. HortScience 32, pp. 812 –816.

Wilson, L.G., Covington, H.M., Averre, C.W., 1977. Sweetpotato production, handling, curing, storage, and marketing in North Carolina. Proc. Symp. Int. Soc. Trop. Root Crop.

Winsor, G.W. 1979. Some factors affecting the quality and compostion of tomatoes. Acta Hort. 93. pp. 335-341.

Woods, T., Velandia, M., Holcomb, R., Dunning, R., Bendfelt, E., 2013. Local Food Systems Markets and Supply Chains. CHOICES 4.

Woolfe, J.A., 1992. Sweetpotato: An Untapped Food Resource. Cambridge University Press.

WorldCrops for Nothern United States. n.d. Sweetpotato *Ipomoea batatas*. [WWW Document]. URL https://worldcrops.org/crops/sweet-potato (accessed 11.18.18).

Xu Z., Chang L, 2017. Convolvulaceae. In: Identification and Control of Common Weeds: Volume 3. Springer, Singapore.

Yen, D. E. 1982. Sweetpotato in historical perspective. In R. L. Villareal and T. D. Griggs (eds.) Sweetpotato: Proceedings of the First International Symposium. AVRDC, Tainan, Taiwan, pp. 17–30.

Zhang, D., Cervantes, J., Huaman, E., Carey, E. and Ghislain, M. 2000. Assessing genetic diversity of sweetpotato (Ipomoea batatas (L.) Lam.) cultivars from tropical America using AFLP.Genet. Resour. Crop Evol. 47: pp. 659–665.

Chapter 2 - Identifying the Optimum Postharvest Storage and Handling Conditions for Maintaining Sweetpotato Slip Quality

Abstract

Sweetpotato [*Ipomoea batatas* (L.) Lam.] is a tropical perennial native to Central and South America and is suited for both large and small-scale, organic and conventional production. Sweetpotato is propagated vegetatively through stem cuttings known as slips. In the United States, slip and storage root production is concentrated in the Southeast and sweetpotato growers in the Northern and Central regions are left reliant on plant material that is shipped from outside the region. Transportation and storage conditions can result in low quality slips that may perform poorly after being planted in the field. The objective to identify the optimal storage and shipping conditions related to temperature and packaging for sweetpotato slips. Slips of "Orleans" were placed into small waxed cardboard boxes (12" x 4" x 4") with or without perforated nylon-film liner and stored at 16°C, 22°C, and 30°C at 65% relative humidity. An overall quality was evaluated with a rating scale that was developed, which rated slips from 1 to 9 (1- completely senesced to 9- field fresh slip). In addition to visual quality, we also evaluate quality by monitoring water loss, chlorophyll fluorescence, respiration rate, color, and chlorophyll content through the storage. Slips stored at 16°C with a liner had the longest shelf life maintaining marketable quality for just over 11.3 days, followed by 10.6 days for 16°C without a liner, 8.3 days for 22°C with a liner, 7.5 days for 22°C without a liner, and 6.5 and 6.4 days 30°C with and without a liner. After 4 days of storage, slips stored with the addition of a liner had significantly lower water loss. Slips stored at 16°C with a liner lost 3.6% water weight compared to 13.3 %

for slips stored without a liner ($P<.01$). Slips stored at 22°C with a liner lost 1.7 % water weight compared to 14 % for slips stored without a liner ($P<.01$). Slips stored at 30°C with a liner lost 6.4 % water weight compared to 19 % for slips stored without a liner ($P< 0.05$). The results of this work show that temperature and the addition of a liner play critical roles in maintaining quality and freshness of sweetpotato slips. Information from this study increases the postharvest handling knowledge of sweetpotato slips with hopes of contributing to the development of standardized shipping practices for shipping and storing sweetpotato slips.

Introduction

The sweetpotato [*Ipomoea batatas* (L.) Lam.] is a drought tolerant crop that can be grown in both tropical and temperate climates and is well adapted to low and high input systems that utilize varying degrees of technology (Bouwkamp, 1985). Sweetpotato is a tropical perennial but is grown as an annual in temperate climates as it does not tolerate frost (Bouwkamp, 1985; Thottappilly, 2009). Sweetpotato is highly heterozygous meaning it has more than one allele for the same gene. The crop is also generally self-incompatible and does not inbreed. In temperate climates most sweetpotato cultivars produce uneven blooming patterns or may fail to bloom completely (Jones, 1967). Moreover, these factors combined contribute to the unlikelihood that embryonic seed (sexual reproduction) will produce true-to-type offspring (Loebenstein et al., 2009). Currently, sweetpotato seeds are only used for breeding purposes and are not suitable for commercial agricultural applications (Gaba and Singer, 2009). In the United States, sweetpotato is vegetatively propagated from stem cuttings known as “slips”, produced from bedded seed roots (Edmond, 1971; Thottappilly, 2009).

Sweetpotato production is an important agricultural business in the United States. The value of domestic production in 2018 was estimated at over $650 million (USDA NASS, 2018).

However, core production areas for both slip and storage roots are concentrated among four states: North Carolina, Louisiana, Mississippi, and California (Estes, 2009). North Carolina has been the leading sweetpotato producer for the past 50 years accounting for more than 50% of domestic sweetpotato production (Bond, 2017; USDA NASS, 2018). In 2016, North Carolina Harvested 95,000 acres of sweetpotato, nearly 30,000 more than Louisiana, Mississippi, and California combined (USDA NASS, 2018).

Growers outside of the core production states collectively contribute less than 4% to domestic production (Estes, 2009). However, sweetpotato can be grown successfully in the cooler climates of the Central and Northern regions of the United States (Nair, 2018). In a 2014 Northeast Kansas food hub feasibility study, Flaccavento et al. 2014, reported that sweetpotato was a “core item” in the region. Which, because it’s of suitability to the region’s soils and climate, freshness, taste, appearance, and uniqueness of varieties, had a “competitive advantage” when produced locally. The feasibility study also reported that within the region, sweetpotato has the potential to be sold for processing in addition to being marketed for direct-to-consumer sales. While local demand and climate are suitable for sweetpotato production in the North Central region of the United States, sweetpotato growers in this region face a major obstacle: access to, and control of quality propagation material. Sweetpotato slips shipped to the North Central region from core production zones in the Southeast may arrive in poor condition after several days in shipping. Furthermore, unpredictable spring weather patterns or other unforeseen circumstances could delay field planting of the slips which results in a need for an extended shelf life. Currently, no standardized information exists for the proper storage and postharvest handling of sweetpotato slips, especially regarding temperature and packaging during the shipping process.

Most slip producers rely on U.S. Postal Service, or other private courier delivery services (UPS, FedEx, etc.) to deliver slips, which do not offer temperature and humidity control. Furthermore, there are many variations in preparing slips for shipping amongst producers. For example. Victory Seed Company in Molalla, Oregon ships out small, bound bundles of slips with the basal end wrapped in strips of newspaper. The bundle is wrapped again in parchment and placed in a cardboard box for shipping (Victory Seed Co., 2018, personal contact). Similarly, the Sandhill Preservation in Calamus, Iowa ships out slips in small bundles with the basal end wrapped in newspaper, the basal ends are then placed in a small plastic bag. The whole bundle is subsequently wrapped in newspaper and placed in a box for shipping (Sandhill Preservation, personal contact). Steel Plant Company in Gleason, Tennessee ships out bound bundles of 25 slips with the roots packed in rehydrated sphagnum moss. The Bundle is subsequently wrapped in parchment paper and shipped out (Steele Plant Co., 2018, personal contact). These shipping practices are more common amongst retailers who are selling to small scale growers and hobby gardeners, or who specialize in rare and heirloom cultivars and who are typically charging more money per slip. Conversely, large scale producers like Jones Family Farm in Baily, North Carolina ship out slips that are placed unbound into waxed produce boxes; approximately 1000 slips/bushel box (Jones Family Farm, 2018, personal contact). These variations in packaging practices are primarily reflective of the size of the operation and the volume of slips purchased by the grower.

Temperature management is the most effective tool for preserving quality and extending the shelf life of fresh horticultural commodities and is central to modern postharvest handling systems (Kader, 2013, Sommer, 1982). In addition to slowing the growth rate of spoilage pathogens, storage at the lowest temperature tolerated by the commodity is ideal as it maximizes its physiological postharvest life (Sommer, 1982). The ideal storage temperature of a commodity is largely dependent on its geographic origin. For example, commodities of tropical origin evolved in warmer climates and do not tolerate temperatures lower than 12°C during storage. Whereas, commodities that evolved in cooler, temperate climates can be stored at temperatures as low as 0°C (Jobling, 2000). Temperature management often involves two steps: rapid cooling to remove field heat and maintaining the ideal storage temperature (Sommer 1982). Cooling is typically accomplished by forced-air cooling or hydrocooling, followed by storage in a facility with adequate refrigeration capacity (Sommer, 1982).

Today, sweetpotato slips are shipped immediately after harvest, through the U.S. Postal Service, or private courier delivery sources which do not employ temperature management technology for standard shipping. This means that slips could be exposed to a wide range of temperatures during transport. Like basil (*Ocimum basilicum*), sweetpotato slips are tropical in origin. Basil can't be stored at less than 10°C without deterioration due to chilling injury (Mitcham et al. 2001). Lang and Cameron (1994) reported that the symptoms of chilling injury were severe for basil stored at 0 and 5°C. Moderate chilling injury was also observed at 7.5 and 10°C (Lang and Cameron 1994). *Ipomoea aquatica* also known as water spinach or swamp cabbage is a relative of the sweetpotato and is also tropical in origin. It has been reported that *I. aquatica* is chilling sensitive with a recommended storage temperature of around 12°C (Gross et al., 2004). Conversely, exposure to high temperatures can result in heat injury. For example, enzymes which keep the plant functioning slow down at temperatures above 30°C and cease operation at 40°C (Sommer, 1982). More generally, as temperatures increases, so does the rate of metabolism leading to a shorter shelf life (Sommer, 1982).

Sweetpotato slips are similar to leafy green vegetables and herbs in that they consist primarily of leaves, shoots, and stems and therefore have a high-water content and a high surface to volume ratio, making them similarly vulnerable to water loss. Leafy green vegetables are extremely vulnerable to water loss in comparison to other fresh commodities like soft fruit, bulbs, and tubers (Kays and Paull, 2004). Water loss is the primary cause of poor quality and postharvest loss in leafy green vegetables such as lettuce, chard, cabbage, and spinach (Ben-Yehoshua and Rodov, 2002). Leafy greens can appear wilted after just 3-5 percent water loss (Holcroft, 2015). For example, leaf lettuce can lose a maximum of 3-5 percent of its water weight before its determined to be unmarketable (Thompson et al., 2008), whereas spinach can lose only 3 percent of its weight before it is considered unmarketable (Kays and Paull, 2004). Watercress can lose up to 7 percent water weight and cabbage 6-11 percent before becoming unmarketable (Kays and Paull, 2004; Thompson et al., 2008).

To prevent water loss and maintain freshness after harvest, it is essential that in addition to temperature management that appropriate packaging is used for leafy green vegetables (Bautista and Acedo, 1987). Prevention of water loss is one of the primary purposes of packaging fresh leafy greens and herbs in plastic films and containers for retail sales (Hruschka and Wang, 1979). The goal of packaging technologies for leafy greens and herbs is to act as a barrier to prevent water loss, reduce physical injury during transit and handling, and to allow for adequate ventilation to hasten cooling and allow heat from respiration to escape (Gast, 1991). High-density plastic films have been shown to greatly inhibit water loss in fresh produce, especially leafy vegetables (Ben and Yehoshua, 1978). O’Hare et al. (2001) reported that moisture loss was effectively reduced by packaging pak choi in plastic film and proved to be more effective than manual misting or anti-transpirant treatments. Aharoni et al. (1993), demonstrated that the quality of a variety of salad herbs (chives, chervil, coriander, dill, sorrel, and watercress) stored for 5 days at 6°C and then 2 days at 12°C was improved by the addition of polyethylene liners in the storage cartons. While the advantages of temperature management and appropriate packing for leafy greens and herbs has been reviewed by many researchers (Acedo and Weinberger, 2007; Aharoni et al. 1993; Cantwell et al., 1998; Cantwell and Reid, 1993; Lange and Cameron, 1994; Lee and Kader, 2000; Mitcham et al., 2001; no previous postharvest research has been conducted on sweetpotato slips specifically.

A better understanding of proper postharvest handling and transportation practices will benefit sweetpotato growers in the Central and Northern regions of the United States in several ways. Slips shipped from the core production states in the southeast could arrive in a better-quality condition and perform well after planting. Additionally, sweetpotato vines are being sold as greens at farmers markets (Kansas City, City Market, 2018) and through CSA's (KC Farm School at Gibbs Road, CSA, 2019) within the northcentral region of the United States. Sweetpotato greens may have a market in the region as they grow better than other greens during the summer season (Ishiguro et al., 2004). The nutritional value of sweetpotato greens are comparable to other commercial vegetables in protein, vitamins, and mineral content (Woolfe, 1992). As interest in the consumption of fresh sweetpotato greens increases, information provided by postharvest research will give local growers recommendations on best storage and handling practices to ensure they can provide a fresh, high-quality product.

Sweetpotato slips shipping typically required several days in transit depending on the destination. During this time, slips can deteriorate to various degrees due to water loss, senescence, and temperature extremes (both high and low). Today, slip producers employ various practices to prepare slips for shipping none of which are standardized. As a result, several days in shipping could lead to low quality slips that perform poorly or even fail after being planted in the field. Given the lack of information regarding postharvest practices for sweetpotato slips, the overall objective of this study was to investigate the postharvest behavior of sweetpotato slips throughout storage. The specific research objectives included: iii) identify the optimum storage and transport conditions related to temperature and packaging for maintaining the postharvest quality of sweet potato slips, and (ii) to investigate if the addition of a perforated nylon liner prevents water loss maintains freshness throughout storage.

Materials and Methods

Experimental Design and Plant Material

The experiment was conducted during the 2017 and 2018 growing seasons at Kansas State University Olathe in the Postharvest Physiology Laboratory (Olathe, Kansas, USA). Sweetpotato slips of ‘Orleans’ were grown and harvested at the Kansas State University, John C. Pair Horticultural Center (JCPHC) in Haysville, KS, USA [Sedgwick County (latitude 37.518928°N, longitude 97.313328°W; USDA Plant Hardiness Zone 6B)]. Soil type at JCPHC is Canadian-Waldeck fine sandy loam (pH = 6.7).

The experimental design was a randomized complete block design with a split-plot arrangement of treatment. Storage temperature was the whole plot, while the liner, no-liner treatments made up the sub-plots. Slips were harvested at JCPHC from commercial slip production beds (approx. 3000 per harvest) on two dates in 2017 and 4 dates in 2018 for a total of six harvests. Slips were harvest in the morning and immediately transported in an air-conditioned vehicle to the Postharvest Physiology Laboratory at Kansas State University Olathe.

Slips were harvested on the following dates: 1 August, 1 September, in 2017 and 4 June, 26 June, 16 July, and 20 August, in 2018.

Packaging and Storage Conditions

Upon arrival to the lab, the slips were cut to 8-10" and placed (50 each) into custom built waxed cardboard boxes, dimensions 12"x 4"x 4". Any slips which were obviously diseased, damaged, and/or not meeting size requirements were discarded and were not included. The boxes were constructed from lids of boxes designed for shipping seafood which were purchased from SCHC Inc., Rosenberg, Texas, USA.

Half of the boxes were packed with slips wrapped inside a liner. The liner was a perforated, translucent, nylon film. The liners were cut from a 20" roll into 20" x 17" segments and wrapped around the slips but left open at the ends to allow for airflow. The liner-wrapped sample was subsequently placed into a box and closed. The sample boxes (both liner and no-liner) were stored at three different temperatures: 16°C, 22°C, or 30°C, all at 65% relative humidity in Forma Environmental Chambers (ThermoFisher Scientific Inc., Asheville, NC, USA). The temperatures were in-part decided by preliminary trials (data not shown) that indicated that slips exhibited signs of mild chilling injury at 12°C. Therefore 16°C was the lowest temperature tested. 22°C was used because it is consistent with an ambient "indoor" temperature. 30°C was used because it is a higher temperature that slips could be exposed to in the back of a standard courier delivery vehicle on a warms spring day. Slips were stored for 12 days at 16°C, 10 days at 22°C, and 8 days at 30°C. The purpose of these temperatures was to investigate how slip quality responds to range of temperatures. The relative humidity level in our experiment was determined by averaging the daytime and nighttime humidity for Kansas, Nebraska, Iowa, and Missouri during the entire months of May, June, and July (noaa.gov, 2018).

Sweetpotato Slip Quality

Changes in slip quality were evaluated on the day of harvest (day 0) and again every 48 hours during storage by monitoring the physical quality of the slips by measuring overall visual quality, water loss, color, and chlorophyll content. Additional physiological measurements were taken which included chlorophyll fluorescence, respiration, and ethylene content. In order to measure the chlorophyll content at various points during storage, slips were destructively analyzed. Because of this, all samples were terminated each day of analysis and new sample boxes were selected for analysis at the each 48-hour interval.

Phyiscal Quality - To evaluate the overall visual quality of the sweetpotato slips we developed a sweetpotato slip rating scale (Figure 2.1), The scale, which was adapted from a scale for spinach (Medina et al., 2012) rated sweetpotato slips from 9 to 1 (9 = field fresh slip, full green, full turgor; 7 = mostly green, minor turgor loss and minor yellowing in the most mature leaves, very few if any defects; 5 = Moderate turgor loss and yellowing in the leaves, complete senescence and rot in most mature leaves, moderate defects in newer leaves; 3 = sever turgor loss in leaves, severe yellowing and defects in newest leaves, mature leaves completely senesced or rotten; 1 = leaves are completely senesced, only the shoot remains, deterioration beginning at nodes) (Figure 2.1). Slips were considered unmarketable at or below a quality rating of five. However, in order to evaluate a visual unmarketable quality rating with an actual impact on yield, we conducted an experiment which investigated the influence of slip quality resulting from storage duration on storage root yield, which is described in Chapter 3. To analyze overall visual quality, one new sample box was selected from each of the six treatments (3 temp x 2 Liner), at each point of analysis. 15 slips were separated from each sample box and given an in individual overall visual quality rating. When approximately 30% of the sample had reached a quality rating of 5 or lower, all samples from that treatment were terminated were terminated. The shelf life of sweetpotato slips in this experiment was assessed using data from the overall visual scale that was developed. Linear trendlines were applied to each temperature and liner combination separately to identify the exact day during storage that slips reached a quality rating of 5.

9	Field fresh slip Fully green No yellowing in leaves No defects in leaves	
7	Slight turgor loss in leaves Minor yellowing in leaves Minor Defects in leaves	
5	Moderate turgor loss Moderate yellowing in leaves Moderate defects in leaves	
3	Severe turgor loss in leaves Severe yellowing in leaves Sever Defects/Deterioration/ Rotting in leaves	
1	Leaves fully senesced Only shoot remains Deterioration beginning at nodes	

Figure 2.1 Overall visual quality rating scale (9 - field fresh slip to 1 - completely senesced)

Water loss was measured by taking an initial weight of each individual sample (plant material only) on day 0, followed by a weight measurement of each sample on the day it was selected for analysis (Day 2,4,6, etc.) prior to removing the 15 slips for visual analysis. After subtracting the analysis day weight from the initial weight, that number was divided by the initial weight, and multiplied by 100. the weight loss was expressed as percent water loss over the storage period as described by Bourne (1976).

Sweetpotato leaf surface color was evaluated on leaf from different slips from every treatment at each point of analysis. One undamaged leaf was selected from each of the four slips. Two measurements were taken from each leaf, one on both sides of the midrib, near the base end, in the broadest part of the leaf blade totaling 8 readings per sample. Slip color was measured using an A5 Chroma-Meter Minolta CR-400 (Konica Minolta Co. Inc., Tokyo, Japan). Results for color analysis were expressed using CIELAB color space system, Lightness (L*) (0-full black to 100- full white) and hue (h*) which is calculated using a* and b* from CIELAB color space and the formula tg-1(a*b*). Calculating hue (h*) provides a reading which corresponds to a specific place on the visible light color spectrum.

The chlorophyll content of 3 slip leaves per treatment was measured at each analysis point, following the method described by Wellburn (1994). For each treatment 0.3 g of photosynthetic tissue was weighed and mixed with 10 ml of pure methanol using a benchtop homogenizer POLYTRON PT 1600 E (Kinematica AG, Luzern, Switzerland). The samples were incubated in darkness at 4 °C for 24 hours. After the incubation period, a spectrophotometer equipped with a 96-well microplate reader (Synergy H1, BioTek Instruments, Inc. Winooski, VT, USA) was used to measure 653 nm (chlorophyll b) and 666 nm (chlorophyll a). Chlorophyll content was calculated with the equations: Chlorophyll a (Chl a): [15.65 x (Abs666) – (7.34 x

(Abs653)]; Chlorophyll b (Chl b): [27.05 x (Abs653) – (11.21 x (Abs666)] and total chlorophyll content = Chl a + Chl b. Total chlorophyll content was expressed as mg/100g fresh weight (FW).

Physiological Measurements - Chlorophyll fluorescence measurements were taken on one leaf from three different slips from every treatment at each sampling point. One undamaged leaf was selected from each of the three slips and one measurement was taken per slip. Chlorophyll fluorescence was measured using an OS-30p+ Fluorometer (Opti-Sciences Inc., Hudson, New Hampshire, USA). Dark-adaption clips were placed on the leaves with the slide shut for 30 minutes. The slides were then opened, and the measurements were taken. Chlorophyll fluorescence was analyzed using the Fv/x protocol and expressed as (F maximum fluorescence - O minimum fluorescence) / F maximum fluorescence or Fv/Fm.

Respiration rate was determined using a closed system method (Biale and Young, 1981). Two sample boxes from each treatment were placed unopened into an air-tight, plastic, 5-gallon bucket and sealed one hour. The amount of CO_2 produced and accumulated in the overhead space of the bucket was measured with a portable gas analyzer (model 900141; Bridge Analyzers, Alameda, CA) Respiration rate was expressed as mg CO_2/kg-h.

Ethylene was measured by extracting a 1 ml sample with a syringe from the headspace of each 5-gallon bucket during the time that they were sealed closed for measuring respiration rate. This was done six times over the course of the experiment (once per harvest). The sample was analyzed by injecting it into a gas chromatograph (SRI Instruments, Torrance, CA, USA) fitted with an FID with 10 ppb limit of detection and equipped with a 6’ HAYESEP-D stainless steel column (100/120 mesh). The injector, column and detector temperatures were set at 125 °C, respectively and helium was used as the carrier gas at a flow rate of 20 ml min^{-1}.

Slip harvest dates for this experiment are listed below in Table 2.1. All statistical calculation and analysis were facilitated by using Statistical Analysis Software (SAS), Cary, North Carolina, USA. SAS procedure *Generalized Liner Model* (GLM) was employed to conduct regression and ANOVA to determine the affect(s) of: temperature, day, and liner on: overall quality, water loss, chlorophyll fluorescence, respiration, color, and chlorophyll content over the course of the storage period. Due to the mixed effects that time (day), temperature and use of liner had on all response parameters, all models were conducted by collectively analyzing day, temp and liner; to obtain a more accurate depiction of the test parameter's affect. Each test–response parameter pair was independently analyzed, and the subsequent model generated was normal, independent and homoscedastic with P value < 0.05 and appropriate F value. Therefore, any claims and/or statements for or against the efficacy of the test parameters are made with statistical significance.

Harvest Dates	
Harvest #	Date
1	August 1, 2017
2	September 1, 2017
3	June 4, 2018
4	June 26, 2018
5	July 16, 2018
6	August 20, 2018

Table 2.-1-Harvests dates for 'Orleans' sweetpotato slips harvested at John C Pair Horticulture Center in Haysville, Kansas during the 2017 and 2018 growing seasons

Results

Because the interaction of temperature, liner treatment, and storage day were significant for all parameters measured (Table 2.2) the results were analyzed and presented separately by temperature (16°C, 22°C, and 30°C)

Slips stored at 16°C had the longest shelf life, followed slips stored at 22°C and slips stored at 30°C. Slips stored at 16°C with a liner reached a quality rating of 5 after 11.3 days, while slips stored at 16°C without a liner reached a quality rating of five after 10.6 days. Similarly, slips stored at 22°C with a liner reached a quality rating of 5 after 8.3 days while slips stored without a liner at 22° reached a quality rating of 5 after 7.5 days. Slips stored at 30°C with and without a liner reached a quality rating of 5 at approximately the same time; 6.4 and 6.5 days respectively (Figure 2-2).

Sweetpotato Slip Quality During Storage at 16°C

Physical Quality - The overall visual quality of slips stored at 16°C, with or without a liner was similar until the 10th days of storage. After 10 days of storage, slips stored with a liner at had significantly higher overall visual quality than slips stored without a liner at 16°C (5.5 vs. 5.0) (P<0.01); (Table 2-2). However, by the 12 day of storage slips stored with or without a liner at 16°C had similar overall visual quality ratings.

The rate of water loss of slips stored at 16°C, with and without a liner were similar after two days storage. After 4 days of storage, slips stored with a liner exhibited significantly lower rates of water loss compared to slips stored without a liner (3.6 vs 13.0%) (P<0.01); (Table 2-2, Fig. 2-3). For the remainder of storage (12 days), slips stored at 16°C with a liner had significantly lower rates of water loss than slips stored without (Table 2-2). On day 12 of storage, slips stored with a liner at 16°C had lower rates of water loss than slips stored without a liner (12.0% vs 31.0%) at the *P*<0.0001 level of significance (Table 2-2, Fig. 2-3).

Leaf color changed only marginally and was similar between slips stored with and without a liner at 16°C for the first 10 days of storage (Table 2). On day 12 of storage, measurements for color L* of sweetpotato slips stored at 16°C, with a liner were significantly lower than those stored without a liner (43.1 vs. 45.3) (*P* <0.01); (Table 2-2). Conversely, slips stored with a liner at 16°C recorded significantly higher measurements for color h compared to slips stored without a liner (*P*<0.001) on day 12 of storage (125.53 vs.122.03); (Table 2-2). During 12 days of storage at 16°, the chlorophyll content of sweetpotato slips stored with and without a liner was similar (Table 2-2).

Physiological Measurements - During storage at 16°C, chlorophyll fluorescence (Fv/Fm) was similar between sweetpotato slips stored with a liner and slips that were stored without a liner (Table 2-2). Respiration rates were similar between sweetpotato slips stored with a liner and slips that were stored without a liner at 16°C (Table 2-2). Ethylene remained lower than detectable levels (<0.05 µl/l) throughout the trial within slip samples stored at 16°C.

Sweetpotato Slip Quality During Storage at 22°C

Physical Quality - The overall visual quality of sweetpotato slips stored at 22°C, with and without a liner was similar until the 6th day of storage. After 6 days of storage, slips stored with a liner had significantly higher overall visual quality ratings than slips stored without a liner (6.2 vs 5.8) (*P*<0.05) (Fig. 2-4). For the remainder of storage (10 days), slips stored at 22°C with a liner had significantly higher overall visual quality ratings than slips stored without a liner (Fig. 2-4, Table 2-4). After 10 days of storage, slips stored with a liner at 22°C had significantly higher overall visual quality ratings than slips stored without a liner (4.1 vs 3.6) (P<0.01); (Table 2-4).

The rate of water loss of slips stored at 22°C, with and without a liner was similar after 2 days storage. After 4 days of storage, slips stored with a liner exhibited significantly lower rates of water loss compared to slips stored without a liner (3.4% vs 14.5%) (P<0.01); (Table 2-4, Fig. 2-5). For the remainder of storage (10 days), slips stored at 22°C with a liner had significantly lower rates of water loss than slips stored without (Table 2-4). On day 10 of storage, slips stored with a liner at 22°C had significantly lower rates of water loss than slips stored without a liner (9.5% vs 33.7%) at the P<0.0001 level of significance (Table 2-4, Fig. 2-5).

Leaf color changed only marginally and was similar between slips stored with and without a liner at 22°C for the first 6 days of storage (Table 3). After 8 days of storage, measurements for color L* of slips stored at 22°C with a liner were significantly higher than those stored without a liner (44.5 vs. 41.9) (P<0.001); (Table 2-4). Conversely, slips stored with a liner at 22°C recorded significantly lower measurements for color h compared to slips stored without a liner on day 10 of storage (125.1 vs 128.0) (P<0.0001); (Table 2-4).

Physiological Measurements - After 8 days of storage at 22°C, slips stored with a liner had a significantly lower Fv/Fm response than slips stored without a liner (0.749 vs 0.791) (P=0.0114); (Table 2-4). By day 10, slips stored with and without a liner at 22°C had similar Fv/Fm responses for chlorophyll fluorescence (Table 2-4). Respiration rates were similar between sweetpotato slips stored with a liner and slips that were stored without a liner at 22°C for the duration of the 10-day storage period (Table 2-4). During 10 days of storage at 22°C, the chlorophyll content of sweetpotato slips stored with and without a liner was similar (Table 2-4).

Ethylene remained lower than detectable levels (<0.05µl/l) throughout the trial within slip samples stored at 22°C

Sweetpotato Slip Quality During Storage at 30°C

Physical Quality - The overall visual quality of sweetpotato slips stored with and without a liner were similar were similar during 8 days of storage at 30°C (Table 2-5). The rate of water loss of slips stored at 30°C, with and without a liner was similar after 2 days of storage (Table 2-5). After 4 days of storage, slips stored with a liner at 30°C exhibited significantly lower rates of water loss compared to slips stored without a liner (3.8% vs 15.5%) (P=0.0304); (Table 2-5, Fig. 2-6). For the remainder of storage (8 days), slips stored at 30°C with a liner had significantly lower rates of water loss than slips stored without (Table 2-5). On day 8 of storage, slips stored with a liner at 30°C had significantly lower rates of water loss than slips stored without a liner (19.8% vs 43.4%) (P<0.01); (Table 2-5, Fig. 2-6). During 8 days of storage, color L* and color h measurements were similar for slips stored with and without a liner at 30°C (Table 2-5). The chlorophyll content of sweetpotato slips stored with and without a liner at 30°C for 8 days was similar (Table 2-5).

Physiological Measurements - Chlorophyll fluorescence (Fv/Fm) was similar between sweetpotato slips stored at 30°C, with and without a liner and until the sixth day of storage (Table 2-5). After 6 days of storage, slips stored at 30°C, with a liner had significantly lower Fv/Fm responses than slips stored without a liner (0.532 vs 0.629) (P=.0284); (Table 2-5). However, there were no other significant differences between liner treatments throughout storage, including day 8 (Table 2-5). During 8 days of storage at 30°C, respiration rates were similar between sweetpotato slips stored with a liner and slips that were stored without a liner (Table 2-5). Color L* and color h were similar between sweetpotato slips stored with and without a liner at 30°C for 8 days (Table 2-5). Ethylene within the sample boxes remained at undetectable levels (<0.05µl/l) at every point of analysis during the trials.

Discussion

In our experiment slips stored at 16°C had the longest shelf life followed by slips stored at 22°C. While slips stored at 30°C had the shortest shelf life. The results of our experiment confirm that temperature management is the most important tool for maintaining postharvest freshness and quality of sweetpotato slips and all other horticultural commodities (Kader, 2013; Prusky, 2011). Refrigerated storage (0-2°C for commodities from temperate climates; 7-13°C for commodities from subtropical and tropical climates) is highly beneficial as it slows deterioration due to ripening, softening, and spoilage pathogens, as well as, slowing water loss and color changes (Hardenburg et al, 1986). Storage at optimal temperature decreases the rate of key metabolic processes and respiratory heat production (Bachmann and Earles, 2000; Kader and Saltviet, 2002). Some of these processes include water loss, compositional changes, and the growth rate of spoilage pathogens (Kader; 2002; Prusky, 2011). Therefore, exposure to nonoptimal temperatures can compromise the quality of fresh commodities, ultimately reducing shelf life (Kader, 2013).

Currently, slips are not shipped through temperature-controlled freight and are exposed to the ambient air temperature within a standard courier delivery vehicle or distribution warehouse, many of which do not utilize temperature management technologies. This means that slips could be subjected to a wide range of temperatures throughout the shipping process. While shipping ideally only requires two days, shipping could be delayed, or weather conditions and other circumstances could delay planting on the grower-end; prolonging exposure to non-optimum temperatures. While slips could benefit from temperature management during transit and storage, one problem is that most refrigeration units and refrigerated transport vehicles maintain temperatures around 4°C or lower as recommended by the FDA. Preliminary trials conducted in our lab indicated that storing slips at or below 10°C will produce symptoms of chilling injury. Ultimately chilling injury could compromise the slip limit its potential to establish after being planted in the field.

The results from our trials indicate that the utilization of a nylon liner had significantly reduced water loss at all temperatures. However, slips stored at 22°C benefited more from being stored in a liner than slips stored at 16°C and 30°C. Slips stored at 22°C with a liner, had significantly higher visual quality ratings than slips stored without a liner at 22°C after 6 days, and through the end of storage. Given that storage at 16°C and 30°C resulted in very few significant differences in terms of visual quality between the liner and no-liner treatments, it is likely that temperature has a stronger influence on postharvest quality than the presence of a liner during storage at more ideal and extreme temperature ranges. In the optimal temperature ranges, key metabolic processes and water loss are already being slowed even without the presence of a liner. While the liner did prevent water loss at more optimal temperatures, it did not significantly influence visual quality. Conversely, the effect of high temperatures (30°C) on slip quality during storage was more influential than the presence of a liner. We also observed the build-up of condensation inside of the liner during storage. Water condensation inside the packaging or on the surface of the commodity is detrimental because it leads to defects in the surface color and texture of the commodity (Garcia et al., 1998; Feng et al., 2003), while simultaneously encouraging microbial growth (Tapia et al., 2007). It is unclear exactly how condensation affected slip quality during storage in our experiment, but this may be an important area for future research with sweetpotato slip packaging.

One of the challenges of producing plants through vegetative propagation is wilting and death before being planted into the field (Alem, 2010). Water loss in propagates is caused by the failure to take up water through the stem or leaves after being severed from the stock plant (Loach and Whalley, 1978). Due to high surface area to volume ratios and the presence of stomata, leafy green vegetables are extremely vulnerable to high rates of water loss through transpiration (O'Hare et al., 2001). Water loss is a major contributing factor to reduction in quality of leafy greens and fresh herbs and is the main cause of wilting and weight loss; leafy greens will appear wilted after just 3-5% water loss (Holcroft, 2015). High rates of water loss lead to decreased visual quality, physiological changes, and can compromise the ability of the commodity to resist attack by spoilage pathogens (Cantwell and Reid, 1993). Water loss is caused by transpiration which involves the transport of water, primarily through the stomates of the leaves, and the subsequent evaporation of this moisture from the surface of the commodity to the surrounding environment (Holcroft, 2015)

In our trials we observed considerable yellowing of slips particularly near the end of shelf life at 20°C and 30°C. Yellowing is a physiological phenomenon related to chlorophyll degradation, which results in dramatic declines of leaf greenness, taste, and nutritive quality (Hue et al., 2011). Postharvest yellowing occurs when leafy organs are stressed due to ethylene sensitivity, high light, water loss, high temperature, or when they cease to produce carbohydrates (Cantwell and Reid, 1993). The cells of the leaf initiate a catabolic process which is designed to recover important nutrients for reutilization in other plant parts. Hydrolysis of proteins and disassembly of the chloroplast apparatus are accompanied by decreased photosynthetic activity and increases in respiration and ethylene production (Cantwell and Reid, 1993). Losses in membrane integrity and the related capacity to maintain ionic balance lead to increased yellowing and ultimately senescence (Cantwell and Reid, 1993). In our experiment yellowing was observed throughout the storage period and was one of the primary characteristics in considering slip quality and ultimately the shelf life of sweetpotato slips. We determined that the yellowing was due to plant senescence and was likely initiated by water loss, and exposure to extreme temperatures as ethylene was below detectable levels at all analysis points.

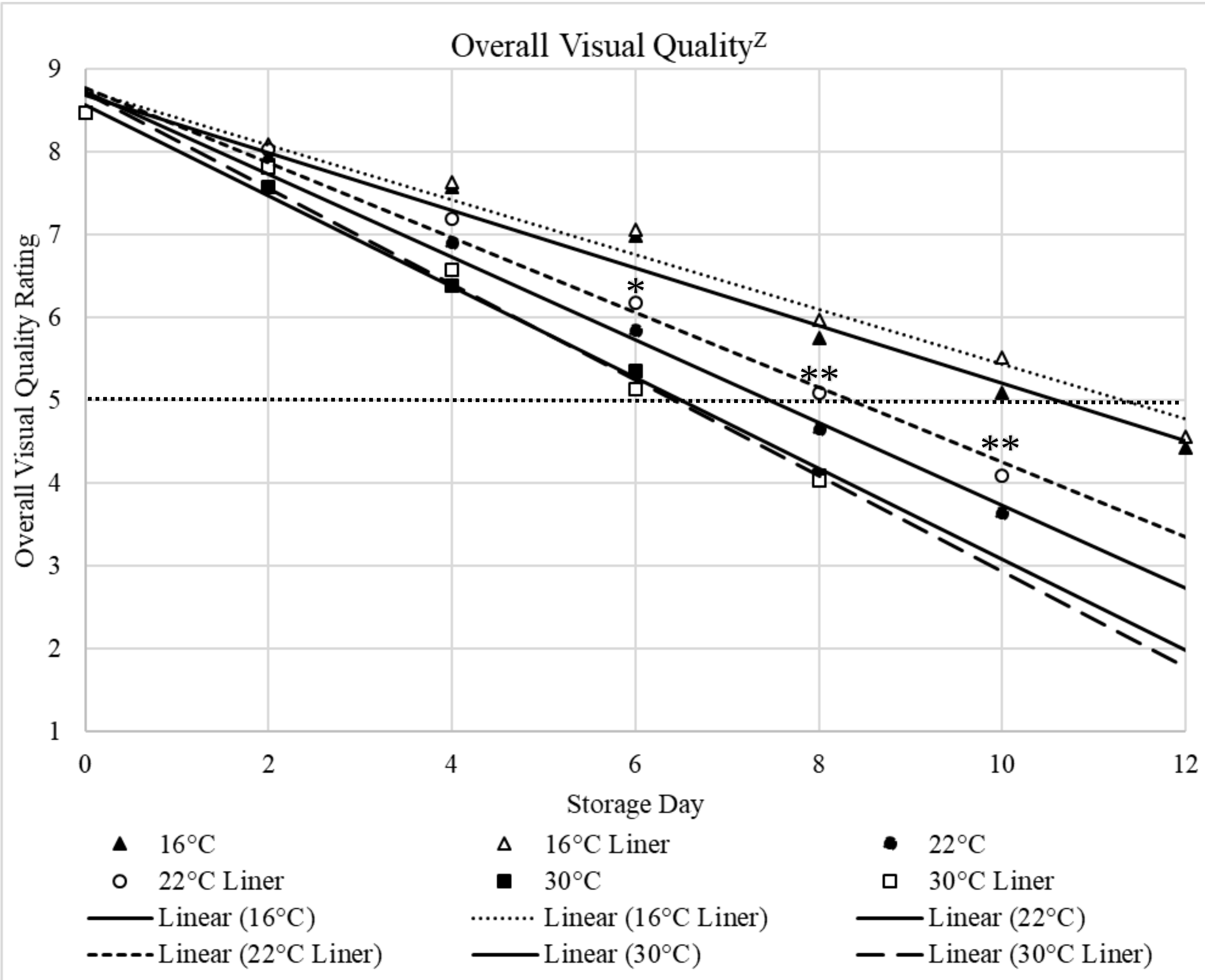
Conclusion

The results of this study indicate that temperature management is crucial for maintaining postharvest quality of sweetpotato slips. Slips stored at lower temperatures maintained higher overall visual quality, exhibited less stress, and had reduced respiration rates throughout storage. Additionally, wrapping the slips in a nylon film-liner prevented water loss during storage at all temperatures. The utilization of a nylon liner is particularly useful for maintaining quality when storing slips at temperatures consist with an indoor ambient room temperature. To our knowledge this is the first study aimed at identifying optimum storage and handling practices for sweetpotato slips. Information from this study could benefit by both large and small-scale slip produces by aiding in providing best management practices for storage and shipping of sweetpotato slips. Moreover, large-scale producers in the core production states could utilize the information provided by this study to ensure that customers in other regions of the country receive high quality propagules that will grow well in the field.

Table 2-2 - Probability values reflecting the effects of temperature, liner, and storage day on overall visual quality (OVQ), water loss, chlorophyll fluorescence, respiration rate, leaf color and chlorophyll content of sweetpotato slips stored at 16C, 22C and 30 with and without a liner.

	Probability Values						
	OVQ	Water Loss	Chlorophyll Fluorescence	Respiration	Color L*	Color hue	Chlorophyll Content
Temp	<.0001	NS	<.0001	<.0001	0.0016	<.0001	NS
Liner	0.0030	<.0001	0.0371	NS	NS	NS	NS
Day	<.0001	<.0001	<.0001	NS	<.0001	<.0001	0.0003
Temp x Liner	<.0001	<.0001	<.0001	<.0001	0.0007	<.0001	NS
Temp x Day	<.0001	<.0001	<.0001	<.0001	<.0001	<.0001	0.0019
Liner x Day	<.0001	<.0001	<.0001	NS	<.0001	<.0001	0.0039
Temp x Liner x Day	<.0001	<.0001	<.0001	<.0001	<.0001	<.0001	0.0430

Figure 2-2 - Mean overall visual quality rating of slips stored at 16°C, 22°C, and 30°C with and without a liner with trendlines applied to indicate shelf life.



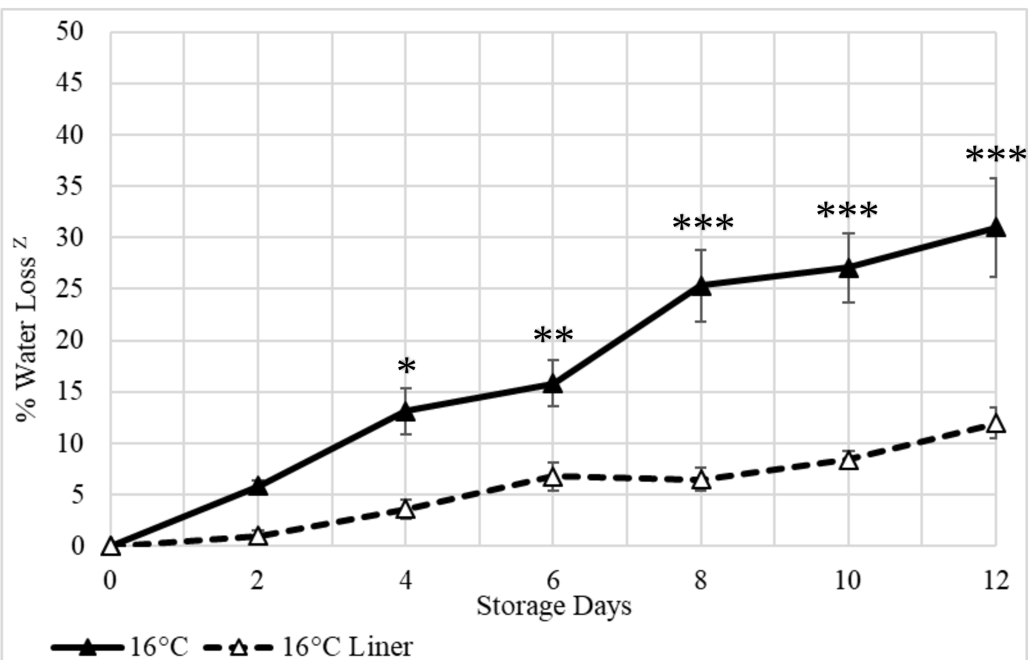
[z] Shelf life was determined using the mean overall visual quality ratings from 6 harvests of slips grown in the open field and stored at 16°C, 22°C, and 30°C, with and without a liner. Each temperature - liner treatment combination was charted and analyzed separately. Trendlines were used to determine the specific time during storage when each temperature and liner treatment reached an overall visual quality rating of 5 which we determined to be the lower limit marketability. * = $P < .05$ level of significance, ** = $P < .01$ level of significance, *** = $P < .001$ level of significance

Table 2-3 - Probability values comparing the means of sweetpotato slips , with and without a liner, during storage stored at 16°C for overall visual quality (OVQ), water loss, chlorophyll fluorescence, respiration rate, leaf color and chlorophyll content.

P-values for Sweetpotato Slips Stored at 16°C for 12 Days [Z]							
Parameter	Day 0	Day2	Day 4	Day 6	Day 8	Day 10	Day 12
OVQ	NS	NS	NS	NS	NS	0.0013	NS
Water Loss	NS	NS	0.0020	0.0033	<.0001	<.0001	<.0001
Chloro Fluor	NS	NS	NS	NS	NS	NS	NS
Respiration	NS	NS	NS	NS	NS	NS	NS
Color L	NS	NS	NS	NS	NS	NS	0.0010
Color h	NS	NS	NS	NS	NS	NS	0.0002
Chlor Cont	NS	NS	NS	NS	NS	NS	NS

[z] *P*-values comparing the means of slips stored at 16°C. Sweetpotato slips cv. 'Orleans' were grown in the open field and stored at 16°C for 12 days. All parameters were measured on day of harvest and again every 48 hours for 12 days. Each value represents a comparison of means that were averaged over 6 harvests. Instances of significance were determined using ANOVA.

Figure 2-3 - The effect of temperature, liner, and day on sweetpotato slip water loss during storage at 16°C.



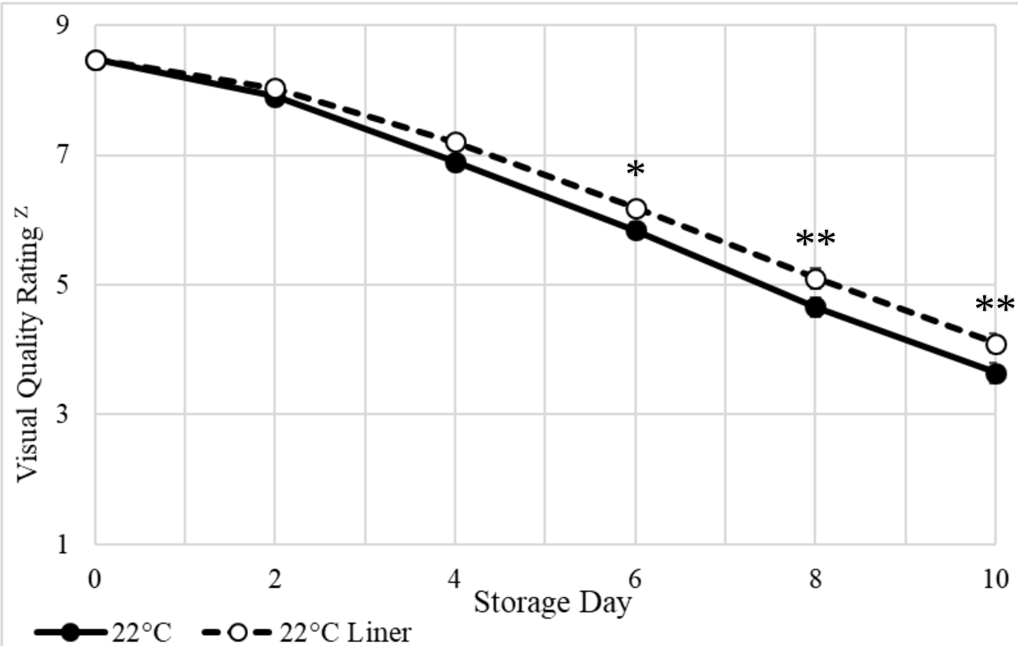
[Z] % Water loss was calculated by taking an initial weight of each sample box (sample included) on day 0, followed by a weight measurement of the sample box on the day it was selected for analysis (Day 2,4,6, etc.). After subtracting the new weight from the initial weight, this number was divided by the initial weight and multiplied by 100. Weight loss was expressed as percent water loss over the storage period. The data points from this graph represent the mean water loss values of slips stored at 16°C with and without a liner, during 12 days of storage. * = $P < .05$ level of significance, ** = $P < .01$ level of significance, *** = $P < .001$ level of significance.

Table 2-4 - Probability values comparing the means of slips stored at 22°C, with and without a liner for overall visual quality (OVQ), water loss, chlorophyll fluorescence, respiration rate, leaf color and chlorophyll content.

P-values for Sweetpotato Slips Stored at 22°C for 10 Days [Z]						
parameter	Day 0	Day2	Day 4	Day 6	Day 8	Day 10
OVQ	NS	NS	NS	0.0419	0.0087	0.0072
Water Loss	NS	NS	0.0031	<.0001	<.0001	<.0001
Chlor Fluor	NS	NS	NS	NS	0.0114	NS
Respiration	NS	NS	NS	NS	NS	NS
Color L	NS	NS	NS	NS	0.0001	NS
Color h	NS	NS	NS	NS	0.0002	NS
Chlor Cont	NS	NS	NS	NS	NS	NS

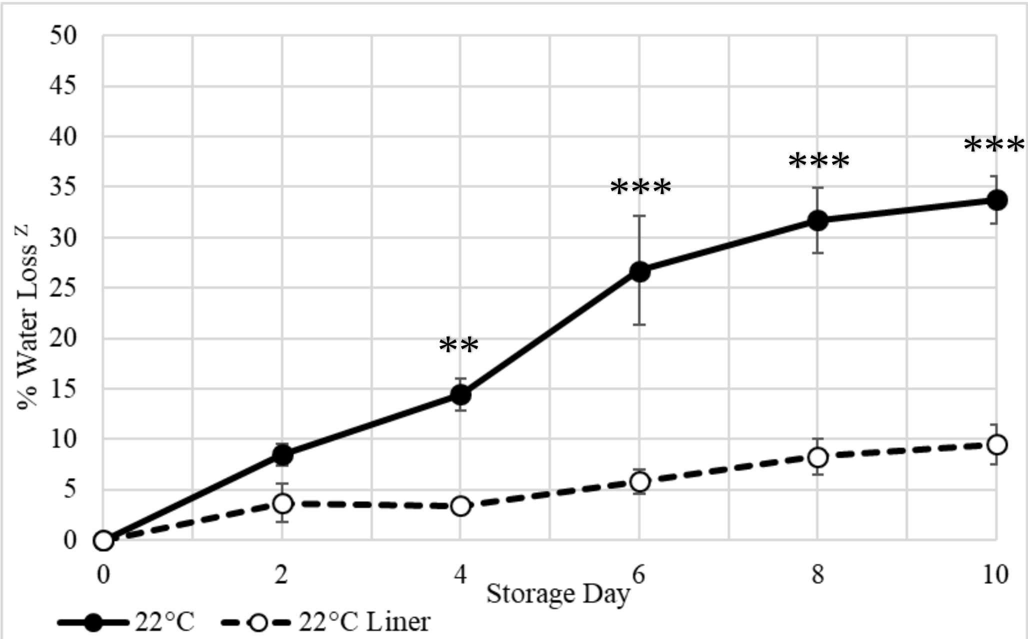
[Z] *P*-values comparing the means of slips stored at 22°C, with and without a liner for the parameters: overall visual quality (OVQ), water loss, chlorophyll florescence, respiration rate, leaf color, chlorophyll content. Sweetpotato slips cv. 'Orleans' were grown in the open field and stored at 22°C for 10 days. All parameters were measured on day of harvest and again every 48 hours for 10 days. Each value represents a comparison of means that were averaged over 6 harvests.

Figure 2-4 - The effect of temperature, liner, and day on sweetpotato slip overall visual quality during storage at 22°C.



Z % Overall visual quality was evaluated using a scale adapted for this experiment which rated sweetpotato slips from 9 (field fresh slip) to 1 (completely senesced). Visual quality of sweetpotato slips stored at 22°C was evaluated on day 0 and every 48 hours for 10 days. The data points from this graph represent the mean overall visual quality ratings of slips stored at 22°C with and without a liner, during 10 days of storage. * = $P < .05$ level of significance, ** = $P < .01$ level of significance, *** = $P < .001$ level of significance.

Figure 2-5 - The effect of temperature, liner, and day on sweetpotato slip water loss during of storage at 22°C.



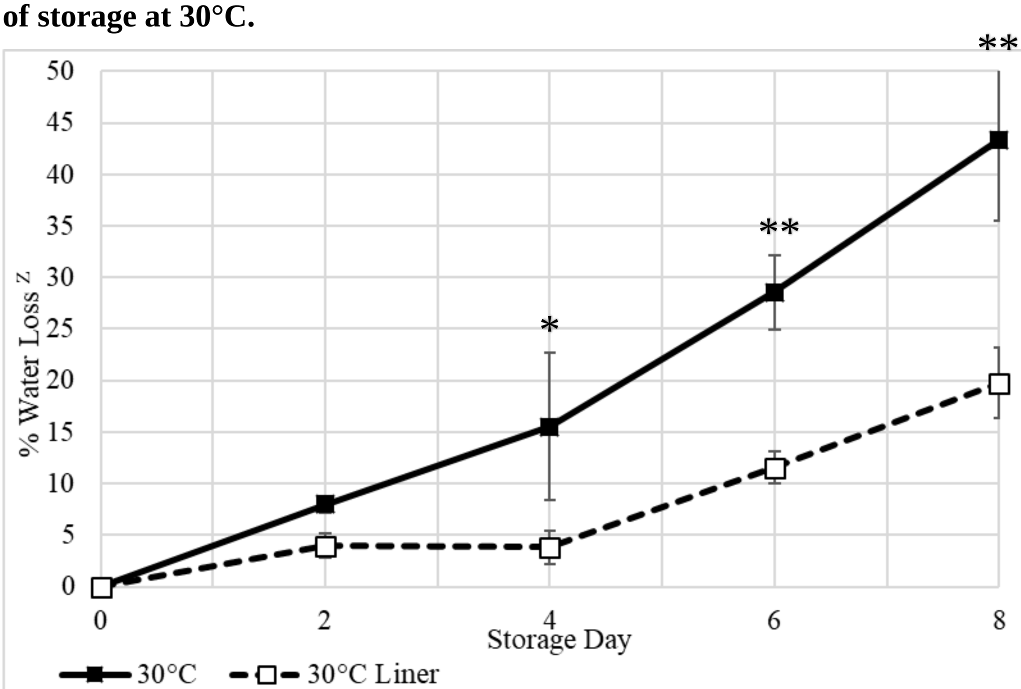
Z % Water loss was calculated by taking an initial weight of each sample box (sample included) on day 0, followed by a weight measurement of the sample box on the day it was selected for analysis (Day 2,4,6, etc.). After subtracting the new weight from the initial weight, this number was divided by the initial weight and multiplied by 100. Weight loss was expressed as percent water loss over the storage period. The data points from this graph represent the mean water loss values of slips stored at 22°C with and without a liner, during 10 days of storage. * = $P < .05$ level of significance, ** = $P < .01$ level of significance, *** = $P < .001$ level of significance.

Table 2-5 - Probability values comparing the means of slips stored at 30°C, with and without a liner for overall visual quality (OVQ), water loss, chlorophyll fluorescence, respiration rate, leaf color and chlorophyll content.

P-values for Sweetpotato Slips Stored at 30°C for 8 Days [Z]					
parameter	Day 0	Day2	Day 4	Day 6	Day 8
OVQ	NS	NS	NS	NS	NS
Water Loss	NS	NS	0.0304	0.0026	0.0014
Chlor Fluor	NS	NS	NS	0.0284	NS
Respiration	NS	NS	NS	NS	NS
Color L	NS	NS	NS	NS	NS
Color h	NS	NS	NS	NS	NS
Chlor Cont	NS	NS	NS	NS	NS

[Z] *P*-values comparing the means of slips stored at 30°C, with and without a liner for the parameters: overall visual quality (OVQ), water loss, chlorophyll florescence, respiration rate, leaf color, chlorophyll content. Sweetpotato slips cv. 'Orleans' were grown in the open field and stored at 30°C for 8 days. All parameters were measured on day of harvest and again every 48 hours for 8 days. Each value represents a comparison of means that were averaged over 6 harvests

Figure 2-6 - The effect of temperature, liner, and day on sweetpotato slip water loss during of storage at 30°C.



[Z] % Water loss was calculated by taking an initial weight of each sample box (sample included) on day 0, followed by a weight measurement of the sample box on the day it was selected for analysis (Day 2,4,6, etc.). After subtracting the new weight from the initial weight, this number was divided by the initial weight and multiplied by 100. Weight loss was expressed as percent water loss over the storage period. The data points from this graph represent the mean water loss values of slips stored at 30°C with and without a liner, during 8 days of storage. * = *P* < .05 level of significance, ** = *P* < .01 level of significance, *** = P < .001 level of significance.

Bibliography

Acedo, A.L., Weinberger, K. Best Practices in Postharvest Management of Leafy Vegetables in Greater Mekong Subregion Countries. Proceedings of a GMS workshop 25-27 October 2007 Hanoi, Vietnam

Aharoni N., Dvir, O., Reuveni, A., 1989. Modified Atmospheres in Film Packages Delay Senecsence and Decay of Fresh Herbs. Acta Hort 258: pp. 255-262

Alem, P.O., 2010. Unrooted stem cutting physiology; water use and leaf gas exchange of severed stem cuttings, All Theses. 918. URL https://tigerprints.clemson.edu/all_theses/918 Clemson University.

Bachmann J, Earles R (2000). Postharvest handling of fruits and vegetables. ATTRA Horticultural Technical Note. Pp. 19.

Bautista, O.K., Acedo, A.L. 1987. Postharvest Handling of Fruits and Vegetables. Manila: National Book Store Inc. Tech guide Series No. 4. p. 24

Ben-Yehoshua, S. and V. Rodov. 2003.Transpiration and water stress. In: J.A. Bartz and J.K. Brecht. (Eds.). Postharvest Physiology and Pathology of Vegetables. 2nd ed., Marcel Dekker, Inc., NY. pp. 111-159.

Bond, J., 2017. U.S. Sweetpotato Production Swells | USDA [WWW Document]. URL https://www.usda.gov/media/blog/2017/01/5/us-sweet-potato-production-swells (accessed 10.18.19).

Bouwkamp, J.C. 1985. Sweet Potato Products: A Natural Resource for the Tropics. CRC Press. Boca Raton, Florida, USA. ISBN 10: 0849354285 ISBN 13: 9780849354281.

Cantwell, M., Reid M. 1993. Postharvest Physiology and Handling of Fresh Culinary Herbs, Journal of Herbs, Spices & Medicinal Plants, 1:3, pp. 93-127, doi:10.1300/J044v01n03_09.

Cantwell, M., Rovelo, J. Nie, X., Rubatzky, V. 1998. Specialty salad greens: Postharvest physiology and shelf-life. Acta Horticulturae. 467. pp. 371-378. doi:10.17660/ActaHortic.1998.467.42.

Edmond, J.B., 1971. Taxonomy, History and Industry, in: Edmond, J.B., Ammerman, G.R. (Eds.), Sweetpotatoes: Production, Processing and Marketing, Major Feed and Food Crops in Agriculture and Food Series. Avi Pub. Co., Westport, Conn., p. 334.

Estes, E.A. 2009 Marketing Sweetpotatoes in the United States: a serious challenge for small-to-moderate volume growers. In: Loebenstein, G. and Thottappilly, G. (eds) The Sweetpotato Springer Science + Business Media BV, Houten, The Netherlands, pp. 269–283.

Flaccavento, A., Williams, M., Shuman, M. 2014. Food Hub Feasibility Study: Northeast Kansas.

Feng, J., Maguire, K. M., Mac Kay, B. R. 2003. Effects of package and temperature equilibration time on physiochemical attributes of ‘Hayward’ kiwifruit. Acta Horticulturae (ISHS), 599, pp. 149-155

Garcia, J. M., Medina, R. J., Olias, J. M. 1998. Quality of strawberries automatically packed in different plastic films. Journal of Food Science, 63, 6, pp. 1037-1041.

Gast, K.L.B. 1991. Containers and Packaging Fruits & Vegetables, Kansas State University, March 1991. http://www.oznet.ksu.edu

Gross, K.C., Wang, C.Y., Saltviet, M.E. 2004. The commercial storage of fruits, vegetables and florist and nursery stocks. USDA Agricultural Handbook 66, U.S. Dept. of Agriculture, Washington.

Holcroft, D. 2015. Water Relations in Harvest Fresh Produce. The Postharvest Foundation White Paper No. 15-01. ISBN978-1-62027-005-9

Hruschka HW, Wang CY. Storage and Shelf Life of Packaged Watercress, Parsley, and Mint. 1979. U.S. Dept. of Agriculture, Mktg. Res. Rept. 1102.

Hardenburg R.E., Watada A.E., Wang C.Y. 1986. The commercial storage of fruits, vegetables, and florist and nursery stocks. U.S. Department of Agriculture, AGRIC. HANDBOOK 66: pp. 47-48.

Jobling, J. 2000. Temperature Management is essential for maintaining produce quality. Sydney Postharvest Laboratory Information Sheet.

Jones, A. 1967. Theoretical segregation ratios of qualitatively inherited characters for hexaploidy sweetpotato (*Ipomoea batatas*). USDA Tech. Bul. No. 1368.

Kader, A.A., 2002: Chapter 4: Postharvest Biology and Technology: An Overview. In Kader, A.A., Postharvest Technology of Horticulture Crops. University of California Agriculture and Natural Resources, Publication # 3311.

Kader, A.A., 2013. Postharvest Technology of Horticultural Crops - An Overview from Farm to Fork. Ethiop. J. Appl. Sci. Technol. (Special Issue No.1): 1- 8

Kader, A. Saltveit, M., 2002. Chapter 2: Respiration and gas exchange of vegetables. In Bartz, J. and Brecht, J.K., Postharvest Physiology and Pathology of Vegetables. CRC Press ISBN 9780824706876 - CAT# DK1909

Kays, S.J. & Paull, R.E. 2004. Postharvest biology. Exon Press, Athens, GA

Lange, D.L. and A. Cameron. 1994. Postharvest shelf life of Sweet Basil. Hort Science 29: pp. 102-103

Lee, S., Kader A. A. 2000. Preharvest and postharvest factors influencing vitamin C content in horticultural crops. Postharvest Biology and Technology 20. pp. 207 – 220

Loach, K. and D.N. Whalley. 1978. Water and carbohydrate relationships during the rooting of cuttings. Acta Hort., 79:161-168

Loebenstein, G., Thottappilly, G., Fuentes, S., Cohen, J., 2009. Virus and Phytoplasma Diseases, In Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 105–134. https://doi.org/10.1007/978-1-4020-9475-0_8

Mitcham, E.J., Crisosto C.H., Kader, A.A. 2001. Herbs (Fresh Culinary): Recommendations for Maintaining Postharvest Quality [WWW Document]. URL http://postharvest.ucdavis.edu/Commodity_Resources/Fact_Sheets/Datastores/Vegetables_English/?uid=17&ds=799 99 (accessed 2.19.19)

Nair, A., 2018. Sweetpotato: Options for the Midwest. Acreage Living Newsletter – Iowa State Extension and Outreach. [WWW Document]. URL https://www.extension.iastate.edu/smallfarms/sweet-potato-options-midwest (accessed 7.7.19)

O'Hare, T.J., Able, A.J., Wong, L.S., Prasad, A., McLauchlan, R. 2001. Fresh-cut Asian Vegetables-Pak Choi as a Model Leafy Vegetable. Postharvest handling of fresh vegetables. Proceedings of a workshop in Beijing, P.R.C., 9-11 May 2001.

Prusky, D. 2011. Reduction of the Incidence of Postharvest Quality Losses, and Future Prospects. Food Security, Vol 3, pp. 463-474

Sommer, N.F., 1982. Postharvest Handling Practices. Plant disease 66, 5, p.357.

Tapia, M. S., Alzamora, S. M., Chirife, J. 2007.Effects of water activity on microbial stability: as a hurdle in food preservation. In G.V. Barbosa-Canovas, A.J. Fontana, S.J. Schmidt, T.P. Labuza (Eds.), Water Activity in Foods – Fundamentals and Applications, Blackwell Publishing and IFT Press, pp. 239-271.

Thompson, J.F., Mitchell, F.G., Rumsey, T.R., Kasmire, R.F., Crisosto, C.H. 2008. Commercial Cooling of Fruits, Vegetables, and Flowers. University of California, Agriculture and Natural Resources, Pub. 21567

Thottappilly, G., 2009. Introductory Remarks, In Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 3–7. https://doi.org/10.1007/978-1-4020-9475-0_1

USDA National Agricultural Statistics Service (NASS), 2018. United States Department of Agriculture National Agricultural Statistics Service Crop Production 2017 Summary

USDA National Agricultural Statistics Service (NASS), 2018. United States Department of Agriculture National Agricultural Statistics Service. Statistics by Subject: National Statistics for Sweet Potatoes

Chapter 3 - The Influence of Sweetpotato Slip Storage Duration on Vine Establishment, Growth, and Storage Root Yield

Abstract

To achieve high storage root yield and quality, it is imperative that growers follow recommended production practices. However, practices pertaining to the proper handling of freshly harvested slips are often overlooked. Because U.S. sweetpotato slip production is concentrated in the Southeast U.S. and California, shipping to other regions can require several days. During shipping, slip quality can deteriorate to varying degrees depending on the length of shipping, type of packaging, and the transportation temperature. The objective of this study was to investigate the influence of slip storage duration on slip establishment, growth and storage root yield. Trials were conducted during the 2018 growing season. Two, 4' x 15' slip propagation beds of 'Orleans' were established and harvested, every 3 days over a 12-day and period. Slips were held in storage at 22°C and 65% relative humidity and subsequently planted on July 10, 2018. The trials were planted in a randomized complete block design on hilled rows, with 48 in row centers and 1' in row spacing. Transplant establishment and growth data was collected at 10, 14, 21, 28, and 35 days after transplant. Data included survival rate, stem diameter, vine length, leaf area, shoot biomass, and root biomass. The storage roots were harvested on October 11 October (93 days after transplant). The results of this study show that slips planted the same day they were harvested established the quickest after transplant. However, slips that were stored for 6 days, often out performed slips stored for 3 days in establishment and growth measurements. Plots grown from slips planted the day of harvest (1.81lb/plant) and from slips stored for 6 days (1.68lb/plant) had significantly higher bulk yields than slips planted at a quality 1 (0.76lb/plant) with *P*-value of 0.0020 and 0.0052 respectively.

The results of this work indicate that storing slips for more than 6 days could have a detrimental effect on storage root yields.

Introduction

Sweetpotato *Ipomoea batatas (*L.) Lam. is one of the top ten most commonly consumed fresh vegetables in the United States (FAO, 2017; USDA ERS, 2015). All commercial sweetpotato production fields are typically started with slips (unrooted shoot cuttings) (Smith et al, 2009). As a result, the process of sweetpotato production can be divided into two distinct sections: slip production and storage root production (Smith et al.,2009). Slips are adventitious sprouts produced by bedding storage roots held over from the previous year's crop, commonly referred to as seed roots (Schulthies et al., 2008).

Most commercial sweetpotato growers in U.S. purchase slips annually from large-scale slip producers. The majority of slips available throughout the U.S. are shipped in from major production states such as North Carolina. Slip prices vary depending on several factors including if the slips are organic, or virus-tested, and how many generations (G) the stock is removed from micro-propagation (i.e. G1, G2, G3).

Production of storage roots predominantly takes place in the open field: in raised bed or hilled rows on 48" row centers. slips are typically planted in 12" row spacing, requiring approximately 10,890 slips per acre. The freshly harvested storage roots are graded in the field by size into the following classes: U.S. No. 1's, jumbos, and canners (La Bonte et al., 2012).

To achieve high storage root yield and quality, it is imperative that growers follow recommended production practices (Kemble et. al, 2014; Smith et al., 2009; Wilson et al., 1977). However, practices regarding the proper storage and packaging of freshly harvested slips are often overlooked (Thompson et al., 2017). Holding slips prior to transplant is practiced by many growers in North Carolina (Thompson et al., 2017). The idea behind this practice is that it allows time for the slips to initiate adventitious roots prior to transplant. Adventitious roots originate from root primordia that are visible on the aerial stem prior to slip harvest (Hahn and Hozyo, 1983; Belehu et. al, 2004). Root primordia typically form in pairs on either side of the stem just below the nodes or leaves (Togari, 1950). Adventitious root formation plays a critical role in storage root formation (Lewthwaite and Triggs, 2009). Storage root development commences from the adventitious roots that are pre-formed at nodes and can initiate prior to transplant (Lewthwaite and Triggs, 2009) Thompson et al. (2017) reported that slips that were held 1 day before planting had greater total yield (45.7 MT ha^{-1}) than slips held for 7 days before planting (40.4 MT ha^{-1}), 5 days before planting (41.5 MT ha^{-1}), and slips harvested the same day as planting (42.4 MT ha^{-1}). Slips that were held for 3 days before planting were similar to all holding treatments but produced higher marketable yields (42.6 MT ha^{-1}) than 7 and 5 days before planting. However, they also concluded that environment (location) and soil moisture played a more important role in transplant survival and root formation than holding treatments.

Interregional shipping of sweetpotato slips in the United States is ideally 2-3 days but can require more depending on distance of their destination. During that time slip quality can deteriorate to varying degree depending on the length of shipping, type of packaging, and the ambient environment where the slips are kept. In this study we are investigating how deterioration that can occur during transport or storage can ultimately affect storage root production and yield. More specifically we are studying the effect of the visual slip quality, that was determine by the quality scale that we developed (Chapter 2), on plant stand, growth, and storage root yield.

Materials and Methods

Propagation Bed Establishment

Trials were conducted during the 2018 growing season. Two, 4’ x 15’ slip propagation beds of ‘Orleans’ were established at the Olathe Horticulture Center in Olathe, Kansas [Johnson County (lat. 38.884347°N, long. 94.993426°W; USDA Plant Hardiness Zone 6A), soil type: chase silt loam (pH= 6.3)]. This was accomplished by laying seed roots at grade on the soil surface, close together, but not overlapping. The propagation beds were fitted with two drip irrigation lines that were approximately 1’ apart, lengthwise down the center of the beds. The seeds roots were then covered with approximately 2” of soil and subsequently covered with a single layer of vented clear polyethylene mulch. The plastic mulch was ultimately removed when sprouts began to breach the soil.

Slip Harvest and Storage

When slips reached a height of approximately 12”, they were harvested by hand, every 3 days over a 12-day period. After harvest, slips were transported to the Postharvest Physiology Laboratory at Kansas State University Olathe, Olathe, Kansas. All slips, except for those harvested the day of planting were stored at 22°C and 65% relative humidity in a single Forma Environmental Chamber (ThermoFisher Scientific Inc., Asheville, NC, USA). The slips were held in storage for various lengths of time to artificially influence their quality at time of transplant. The first group of samples harvested were held in storage for 12 days. The second group of slips were harvested 3 days later and were held in storage for 9 days. This process was repeated two more times to for slips held in storage for 6 days and 3 days, respectively. Slips were also harvested the same day that all slips were planted in the field, and represented 0 day storage duration.

Slip planting and Storage Root Harvest

The slips were planted on July 10, 2018 at the Kansas State University John C. Pair Horticultural Center (JCPHC) in Haysville, KS, USA [Sedgwick County (latitude 37.518928°N, longitude 97.313328°W; USDA Plant Hardiness Zone 6B)]. Soil type at JCPHC is Canadian-Waldeck fine sandy loam (pH = 6.7). The trial was replicated at the Olathe Horticulture Center but was unusable due to interference from wildlife. The experimental design was a randomized complete block design with five treatments replicated 4 times. Experimental plots were 20’ long and consisted of 20 slips that were planted by hand, on hilled rows. Rows were 48 in on center and slips were spaced 1’ in the row. Rows were 100’ long and divided into five experimental plots described above; representing each of the five storage durations (0,3,6,9, and 12 days in storage). The plots were irrigated with an overhead system as needed to prevent stress but not fertilized. Transplant establishment and growth data was collected after planting. Data included survival rate, stem diameter, vine length, leaf area, shoot biomass, and root biomass. Storage roots were harvested on 11 October (93 days after transplant). The harvested storage roots were cured for one week at 30°C and 90% relative humidity and then cleaned to remove excess dirt. A bulk weight measurement was then taken which consisted of 4 plants from each plot.

Transplant Establishment and Growth Measurements

In order to observe slip establishment and growth a series of data was collected at 10, 14, 21, 28, and 35 days after planting. Data included survival rate, transplant establishment rating, stem diameter, vine length, leaf area, shoot biomass, and root biomass. For the measurements transplant establishment, stem diameter and vine length an additional day of data was collected 42 days after planting.

Slip survival rate or plant stand was determined at days after transplant by counting the number of plants which were visibly dead or missing. In order to visually track establishment and growth in the first few weeks after transplant, a transplant establishment scale was created for this experiment. The scale, which was adapted from a visual quality scale for spinach (Medina et al., 2012) rated sweetpotato transplants from 9 to 1 as follows: (9 = transplant is alive and growing vigorously, full green, no senescence and/or yellowing; 7 = transplant is alive and growing, mild to moderate senescence and yellowing (one or two leaves); 5 = transplant is alive but growing slowly, moderate to substantial senescence and/or yellowing, 3 = transplant is a small shoot sticking out of the ground, original leaves completely senesced, 1 = transplant failed or is not visible). Each individual plant was rated in the field at points of analysis.

Stem diameter was measurements were taken on four plants that were randomly selected from every plot each on each point of analysis. Measurements were taken just above the soil line but before the first leaf petioles with a manual Vernier caliper. Vine length was measured on the same four plants randomly selected for stem diameter measurements and were also measured at each point of analysis each day measurements were taken. Vine length was measured from the soil line to the tip of the newest leaf blade on the apical shoot of the main stem using a standard ruler and meter stick.

Root and shoot biomass were analyzed by carefully removing two plants from each plot. The below and above ground, portions of the plant were separated into root and shoot biomass, respectively. Both the root and shoot material were dried using a Thermo Scientific Precision™ Gravity Convection Compact Oven (Thermo Scientific, Waltham, Massachusetts, USA). Fresh root and shoot samples were dried in the oven for 24 hours at 100°C. The samples were then left in the ambient room temperature of the lab (Approximately 20°C) for an additional 12 hours and weighed to ensure that all water weight had been removed. The samples were weighed a final time to determine root and shoot which were expressed in grams (g).

Leaf area was measured on six total leaves from each plot that were harvested at each point of analysis. The most mature leaves were selected for analysis, especially in latter weeks. Leaf area was measured using a CI-202 Portable Laser Leaf Area Meter (CID Bio-Science, Inc., Camas Washington, USA).

Statistical Analysis

All statistical calculation and analysis were facilitated by using Statistical Analysis Software (SAS), Cary, North Carolina, USA. SAS procedure *Generalized Liner Model* (GLM) was employed to conduct regression and ANOVA to determine the affect(s) of: storage time, and days after transplant on: transplant visual quality, stem diameter, vine length, leaf area, root biomass, shoot biomass, survivability yield (bulk weight), and percent marketable yield. All models were conducted by separating storage duration, and days after planting to obtain a more accurate depiction of their effects and data on each day after transplant was analyzed independently. For yield data, *Generalized Liner Model* (GLM) was employed to conduct regression and ANOVA to determine the affect(s) of: storage duration on survivability, and storage root yield (bulk weight), marketable yield. Each test-response parameter pair was independently analyzed, and the subsequent model generated was normal, independent and homoscedastic with P value < 0.05 and appropriate F value. Therefore, any claims and/or statements for or against the efficacy of the test parameters are made with statistical significance.

Results

For all establishment and growth parameters measured, days after planting had a significant effect ($P<.0001$) (Table 3-1). Additionally, the interaction of storage time and days after transplant had a significant effect (*P*<.0001) for all establishment and growth measurements (Table 3-1).

Transplant Establishment and Growth Measurements

The survival rate was highest among slips planted the day of slip harvest (100%) followed by slips stored for 3 days (99%), slips stored for 6 days (93%), slips stored for 9 days (86%), and slips stored for 12 days (78%). However, storage time affected survivability at the 0.0525 level if significance. Therefore, we could not make conclusions based on significant differences in survivability between slip with different storage times.

Transplant establishment ratings were significantly lower ($P < 0.05$) during the first 5 weeks after transplant for slips planted after 9 and 12 days of storage compared to those stored for 0, 3, and 6 days (Figure 3-1). By 28 days after planting, no significant differences were observed between the transplants that were planted after 0, 3, and 6 days of storage with ratings 8.4, 7.6, and 7.9 respectively. However, on day 28 these three treatments had higher transplant establishment ratings than transplants planted after 9 days (6.1) and 12 days in storage (5) at the *P*<.0001 level of significance. Furthermore, slips planted after 9 days of storage had significantly higher transplant establishment ratings than those planted after 12 days in storage in the first 5 weeks after transplant at the $P<0.005$ level of significance.

At 35 days after planting slips that were planted after 6 days of storage had the largest stem diameter (9.3mm), followed by slips stored for 0 days (8.6mm) (Figure 3-2). Both groups had significantly larger stem diameters than slips planted after 12 days of storage ($P < 0.05$). However, this was the only day of the trial where significant differences in stem diameter were observed for slips planted after different storage times. For all other days including 42 days after transplant, stem diameter was similar among slips planted after different storage times.

Slips planted after 6 days and 0 days in storage had the longest vine lengths and had similar vine lengths throughout the trial (Figure 3-3). However, 42 days after planting, slips planted after 0, 6, and 9 days in storage had similar vine lengths (140.2cm, 140.4cm, and 132.3 cm) (Figure 3-3). All of which had significantly longer vine lengths than slips planted at after 12 days in storage ($P < 0.05$). Leaf area was greatest for plants grown from slips stored for 0, 3, and 6 days (Figure 3-4). However, on the last day of measurement, 35 days after planting, all quality treatments had similar leaf area.

During the trial root biomass dry weights were similar between all storage treatments until 35 days after planting (Figure 3-5). After 35 days, plants grown from slips harvested the day of planting had significantly greater root biomass than all the other storage treatments ($P < 0.05$). Shoot biomass was similar between all quality treatments at 14 and 21 days after planting (Figure 3-6). At 28 and 35 days after planting, plants grown from slips stored for 0 and 6 days remained similar but had significantly greater shoot biomass than all other storage treatments ($P < 0.05$).

Storage duration had a significant effect on storage root yield ($P < 0.05$). Bulk storage root yield was highest among plots that were planted with slips stored for 0 days (7.2lbs/plot) followed by 6 days(6.7lbs/plot), 9 days (4.8lbs/plot), 3 days (4.7lbs/plot), and 12 days (3lbs/plot) (Figure 3-7). Slips planted after 0 and 6 days in storage, had significantly higher yields in terms of bulk weight than those planted at after 12 days in storage. However, they were both similar in bulk yield to slips planted after 3, 6, and 12 days in storage (Figure 3-7).

Discussion

The objective this study was to investigate the influence of slip storage duration on slip establishment, growth, and storage root yield. Thompson et al. (2017), reported that slips held 1 day before planting had greater total yield than slips held for 7 days, 5 days, or slips harvested the same day as transplant. While our treatments were slightly different, we found that slips that were planted after 0 and 6 days in storage produced the highest storage root yield. Both groups had similar storage root yields to plots that were planted with slips stored for 3 and 9 days but had significantly higher yields than slips that were planted after 12 days in storage. Plants have naturally evolved energy allocation strategies in order to increase their survival potential (Kays, 1985). The energy acquired through photosynthesis can be utilized in two ways: as energy needed for general maintenance of the plant, and energy allocated for production, including foliage and reproductive organs (Kays, 1985). It is possible that slips planted after 12 days in storage, were planted at such a low-quality condition that they simply required so much maintenance energy to initially survive and had to little energy to allocate to storage root formation.

In this research, sweetpotato slips stored for longer periods of time prior to planting had shorter vine lengths in the first several weeks after transplant and were growing at a slower rate. However, by 42 days after planting, the vine lengths were similar to plants started with slips stored for 0, 3, and 6. Regardless of the parity in vine lengths, storage root yields were lowest for plants started from slips stored for 12 days. This could be explained by the allocation of energy (Kays, 1985) into vine growth and canopy development rather than storage root production among plants started from slips stored for longer periods of time (Kays, 1985).

Thompson et al. (2017), reported that many growers in North Carolina believe that the formation of adventitious roots at the nodes prior to transplant leads to higher storage root yields. Villordon et al. (2009) reported that most adventitious roots initiated as early as 5 - 7 days after planting possessed the anatomical characteristics consistent with the development of storage roots for in cv. ‘Beauregard’ and ‘Georgia Jet’. Newly initiated adventitious roots accounted approximately 86% - 89% of the total storage root count at 60 - 65 days after transplant. In our study, although not included in our measurements, we observed the initiation of adventitious roots as early as 3 days after the slips were harvested and prior to transplant. After 6 days in storage, adventitious roots were much more prominent. We also observed that the adventitious roots that initiated during storage prior to harvest were very fragile and often broke when the slips were removed from the packaging; it is unclear if they would remain intact during transplant. However, slips stored for six days out performed slips that were stored for 3 days in many parameters, most importantly, bulk yield. Therefore, it is possible that formation of adventitious roots prior to transplant could play a role in plant establishment and yield.

La Bonte et al. (2012) reported that ‘Orleans’ produced yields comparable to a popular cultivar ‘Beauregard’ in their trials. Storage root yields in sandy loam soils (similar to the soil at John C Pair Horticulture Center) ranked higher in yield of U.S. #1 grade storage roots in comparison to ‘Beauregard’. ‘Orleans’ has been shown to have consistent yields for early, middle, and late season plantings (La Bonte et al., 2012). In our study, ‘Orleans’ slips produced more “canner” grade storage roots than any other marketable grades. La Bonte et al. (2012) reported that cv. ‘Orleans’ requires 115-120 days to developed harvestable storage roots. One of the limitations of our study is that the storage roots were harvested 93 days after the slips were planted. This likely led to a lower than average overall storage root yield in our trial. In the temperate climate of Northeast Kansas, cool springs often prevent adequate time to bed slips and subsequently plant them in the field to produce storage roots. This is one of the reasons why slips are often shipped into the region from core production states in the southeastern U.S.

Another limitation of our study is that data was collected from a single location, over the course of a single growing season (2018). The trial was replicated at the Olathe Horticulture Research and Extension Center however the trial plots were compromised and rendered unusable due to interference from wildlife.

Conclusions

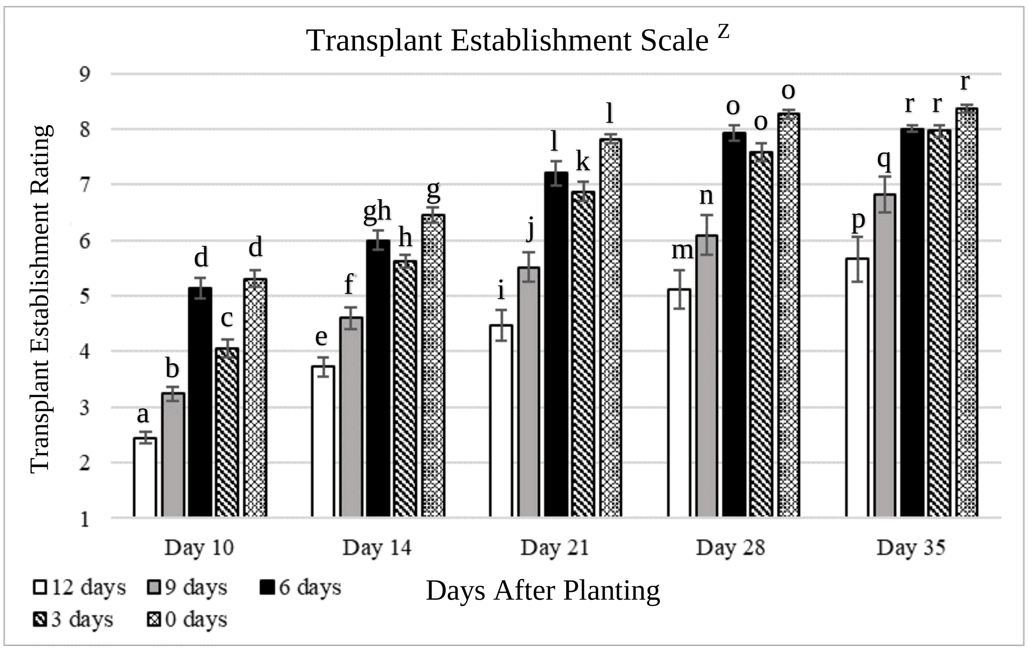
The results of this study show that slips planted the same day of harvest and those stored for 6 days prior to planting established quickest and had the highest storage root yields. However, storage root yield was similar between slips planted the day of harvest and slips stored for up to 9 days. Storing sweetpotato slips for more than 9 days can have a detrimental effect on storage root yield. Future research could conduct similar trials repeated over multiple years and locations and using multiple cultivars to get a more generalizable idea about the effect of slip storage duration time on slip establishment, growth and storage root yield of sweetpotato. In addition to more extensive trials, research should focus on the initiation of adventitious roots and potential influence of the emergence of adventitious root on storage root yield.

Table 3-1 – Probability values for the effect of storage duration, days after planting, and the interaction of storage time x days after planting on transplant visual quality (TVQ), stem diameter, vine length, leaf area, root biomass and shoot biomass of treatments every 7 days for the first 5 -6 weeks after slips were planted.

ANOVA Table for Establishment and Growth Parameters [Z]						
	Transplant Est.	Stem Diameter	Vine Length	Leaf Area	Root Biomass	Shoot Biomass
Storage Duration	<.0001	0.0244	<.0001	0.0015	0.0191	0.0368
Days	<.0001	<.0001	<.0001	<.0001	<.0001	<.0001
Quality x Day	<.0001	<.0001	<.0001	<.0001	<.0001	<.0001

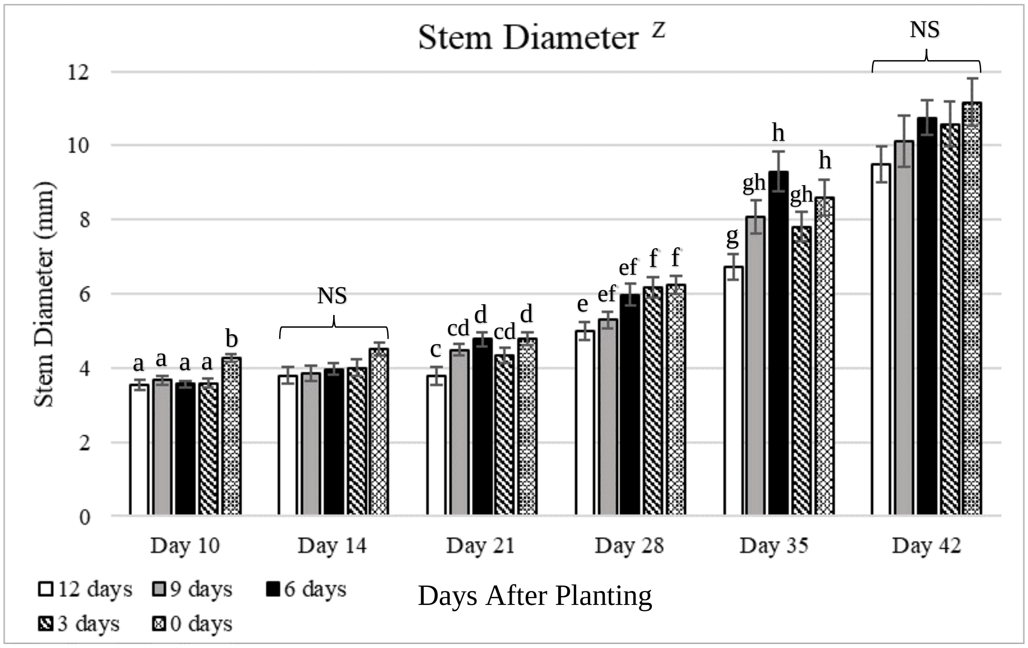
[Z] Table 8 – *P*-values based on the influence of storage duration and days after planting on the parameters: transplant establishment, stem diameter, vine length, leaf area, root biomass, and shoot biomass. *P*-values based on model fit from ANOVA.

Figure 3-1 - The effect of Storage Duration and Days After planting on Transplant Establishment.



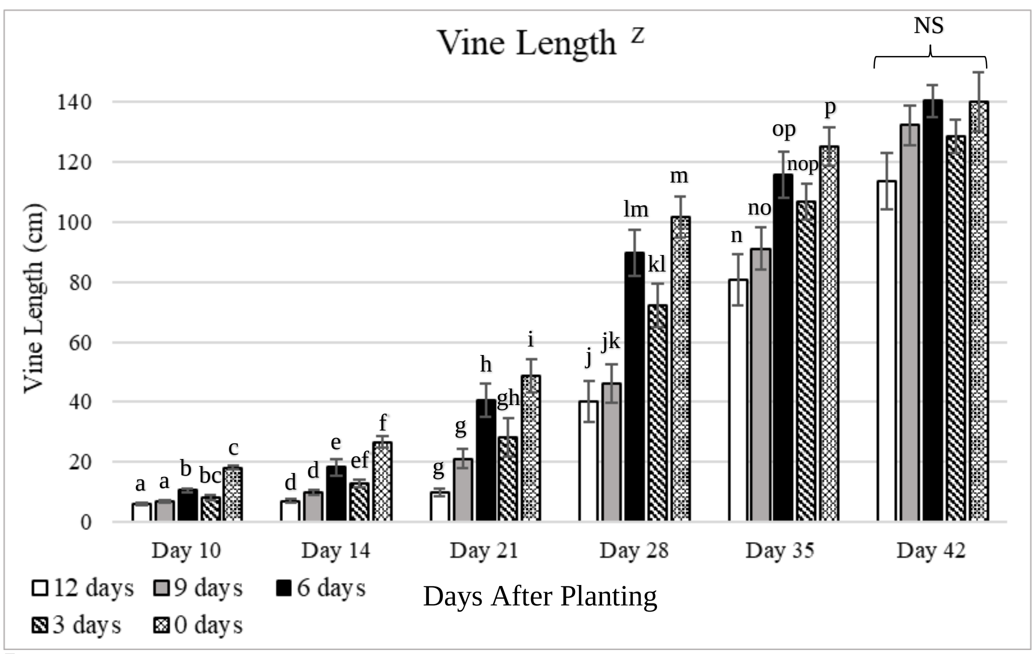
[Z] The transplant establishment scale rated growing sweetpotato plants from 9 = "vigorous growth" to 1 = "dead or not visible". Each individual plant was rated in the field at 10, 14, 21, 28, 35, and 42 days after. The data in this figure is based on the means of each storage treatments on each day after transplant. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently.

Figure 3-2 - The effect of Storage Duration and Days After planting on Stem Diameter.



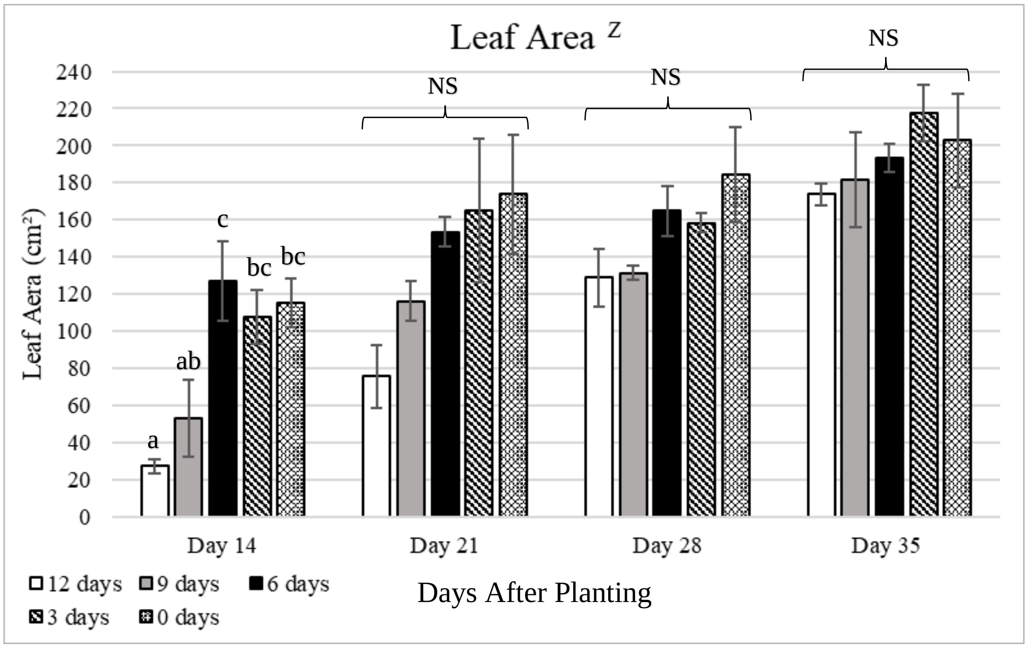
Z Stem diameter was measured at 10, 14, 21, 28, and 35 days after planting using Vernier caliper. Four plants were randomly selected from every plot each day of analysis. The data in this figure is based on the means of each storage treatment on each day after transplant. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently

Figure 3-3 - The effect of Storage Duration and Days After planting on Vine Length.



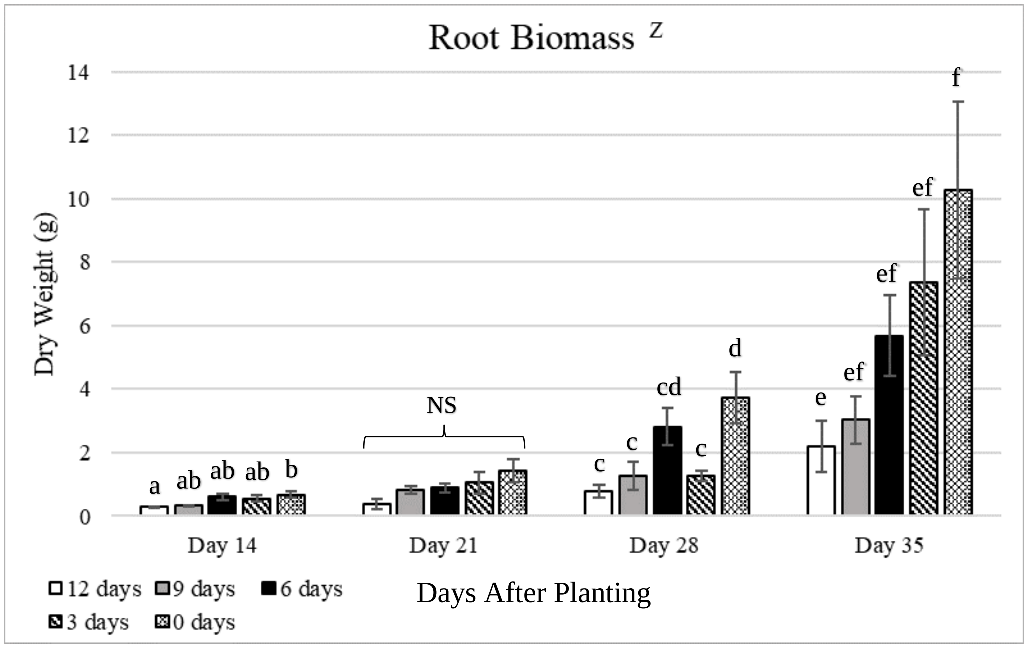
Z Vine length was measured at 10, 14, 21, 28, and 35 days after planting using a standard ruler and meter stick. Four plants were randomly selected from every plot each measurement. The data in this figure is based on the means of each storage treatment on each day after planting. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently.

Figure 3-4 – The effect of Storage Duration and Days After planting on Leaf Area.



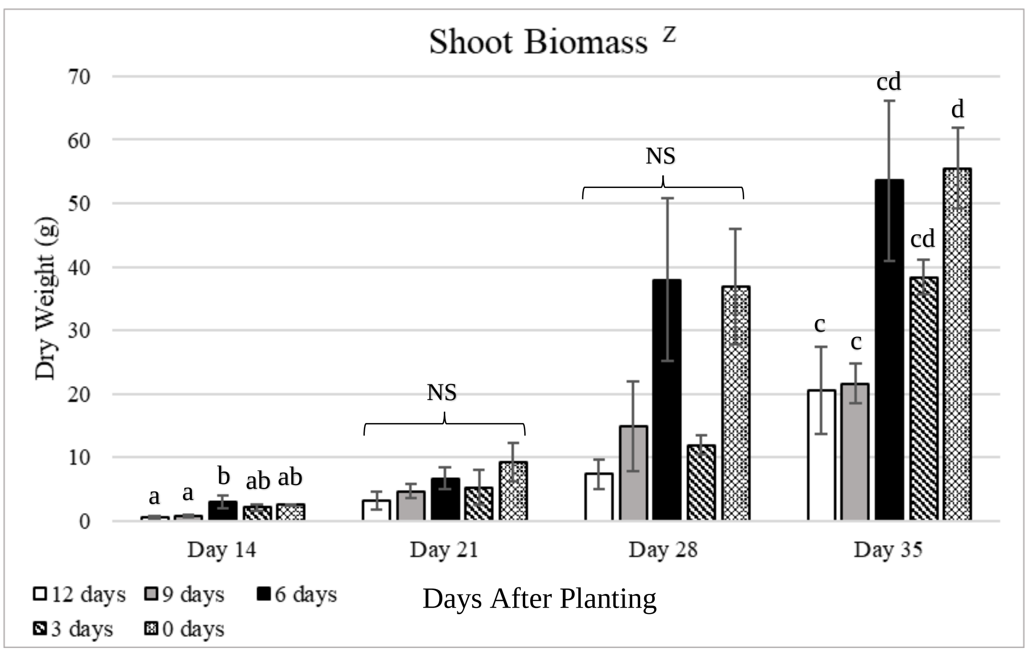
Z Leaf area was analyzed at 14, 21, 28, 35 days after planting. Three green, undamaged mature, leaves were removed from two slips (6 total). The data in this figure is based on the means of each storage treatment on each day after planting. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently.

Figure 3-5 – The effect of Storage Duration and Days After planting on Root Biomass



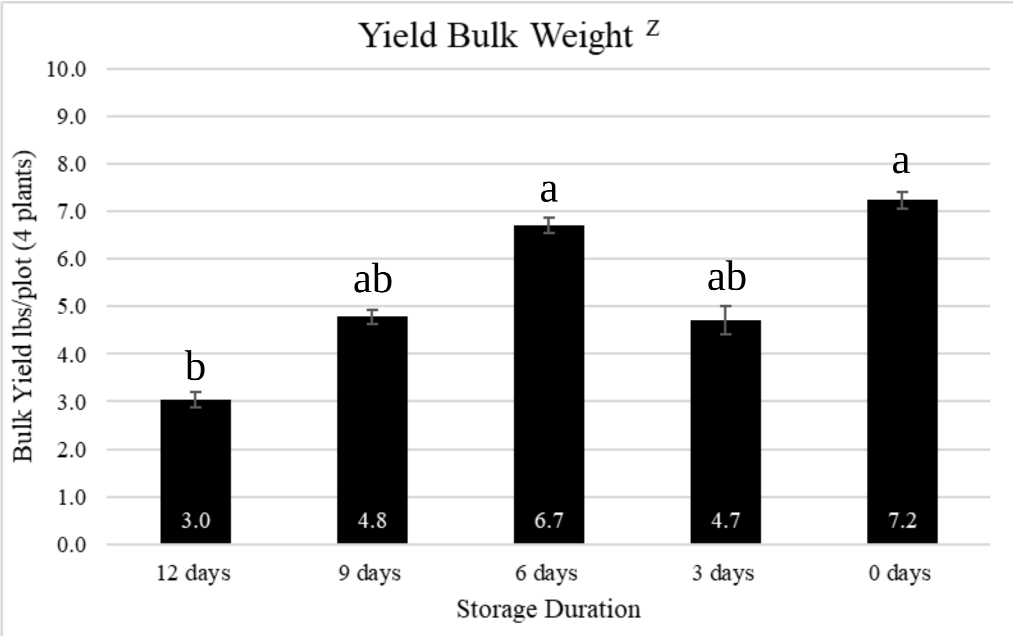
Z Root biomass was analyzed at 14, 21, 28, and 35 days after planting. plants from each storage treatment were removed from the field and dried. The data in this figure is based on the means of each storage treatment on each day after planting. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently.

Figure 3-6 – The Effect of Slip Storage Duration and Days After Planting on Shoot Biomass



Z Shoot biomass was analyzed at 14, 21, 28, and 35 days after planting. plants from each storage treatment were removed from the field and dried. The data in this figure is based on the means of each storage treatment on each day after transplant. Instances of significance were determined using ANOVA and were analyzed on each day after planting independently.

Figure 3-7 – The effect of Slip Storage Duration on Bulk Yield



Z Bulk yield was determined by averaging the weight of storage roots harvested from 4 plants from each plot representing different storage treatments, 93 days after planting. The data in this figure represents the mean bulk weight from each storage treatment. Instances of significance were determined using ANOVA

Bibliography

Belehu, T., Hammes, P. S., Robbertse, P. J. 2004.The origin and structure of adventitious roots in sweetpotato (*Ipomoea batatas*). Aust. J. Botany 52: 551-558.

Food and Agriculture Organization of the United Nations (FAO), 2017. Value of Agricultural Production [WWW Document]. Food Agric. Organ. United Nations. URL http://www.fao.org/faostat/en/#data/QV (accessed 10.17.18).

Hahn, S. K., Hozyo, Y. 1983. Sweetpotato and yam. Symposium on potential productivity of field crops under different environments. IRRI, Los Banos, Philippines.

Kemble, J. M., Quesada-Ocampo, L. M., Ivors, K. L., Jennings, K. M., Walgenbach, J. F. 2013. 2013 Southeastern US Vegetable Handbook. Lincolnshire, IL: Vance Publishing Corp: pp. 91-92.

Kays S. J., 1985. Chapter 4: The physiology of yield in the sweetpotato. In Bouwkapmp, J. C., Sweet Potato Products: A Natural Resource for the Tropics. CRC Press, Boca Raton, Florida. pp. 79-132

La Bonte, D.R., Clark, C.A., Smith, T.P., Villordon, A.Q. 2012. Orleans Sweetpotato. HortScience 47, 1817–1818.

Lewthwaite, S. L., Triggs, C. M. 2009. Preliminary study of the spatial distribution of sweetpotato storage roots. Agronomy New Zealand 39: 111-122

Schultheis, J. R., Boudreaux, J. E., Shankle, M.W., Smith, Tara P., et al. 2008. In G. J. Holmes and J. M. Kemble (eds.) Vegetable Crop Handbook for the Southeastern US (Sweetpotato Section).

Smith, T.P., Stoddard, S., Shankle, M., Schultheis, J., 2009. Sweetpotato Production in the United States, In Loebenstein, G., Thottappilly, G. (Eds.), The Sweetpotato. Springer Netherlands, Dordrecht, pp. 287–323. https://doi.org/10.1007/978-1-4020-9475-0_14

Thompson, W.B., Schultheis, J.R., Chaudhari, S., Monks, D.W., Jennings, K.M., Grabow, G.L., 2017. Sweetpotato Transplant Holding Duration Effects on Plant Survival and Yield. HortTechnology 27, 818–823. https://doi.org/10.21273/HORTTECH03808-17

Togari, Y., 1950. A study in the tuberous-root formation of sweetpotato. Bull. Natl. Agric. Exp. Stn. 68, 1–96.

Villordon, A. Q., D. R. La Bonte, N. Firon, Y. Kfir, E. Pressman, and A. Schwartz. 2009. Characterization of Adventitious Root Development in Sweetpotato. Hort Science 44(3): 651655.

Wilson, L.G., Covington, H.M., Averre, C.W., 1977. Sweetpotato production, handling, curing, storage, and marketing in North Carolina. Proc. Symp. Int. Soc. Trop. Root Crop.