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NON-CONTACT AND EARLY DETECTION OF PLANT WATER STRESS USING INFRARED THERMOMETRY AND IMAGE PROCESSING

DISSERTATION

Presented in Partial Fulfillment of the Requirements for

The Degree Doctor of Philosophy in the Graduate

School of The Ohio State University

By

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* * * * *

The Ohio State University
2000

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A methodology for early, non-contact, non-destructive, and quantitative detection of plant water stress for plants grown in controlled environments was developed with applications of infrared thermometry using crop water stress index (CWSI) and image processing using top projected canopy area (TPCA) of the plants.

A computer-controlled system was designed and built for continuous monitoring of plant health and growth. A crop water stress index model for plants grown under controlled environments was developed using basic thermodynamic principles and the energy balance of the plant. Model predicted CWSI values were correlated with measured CWSI values with $R^2$ values of 0.83, 0.50, 0.79, and 0.76 for the experiments conducted. An inverse and linear correlation was found between crop water stress index and measured evapotranspiration rates. The leaf temperatures of the stressed plants were found to be 1-3 °C higher than the air temperature. The leaf temperatures of well-watered plants were consistently lower (1-4 °C) than air temperature during the experiments.

Top projected canopy area (TPCA) of the plants was extracted from plant images using machine vision and image processing techniques. TPCA expansion of the plants in the treatment group was temporarily inhibited as the plants experienced water stress. Following the irrigation, as the plants recovered from water stressed condition, the TPCA expansion continued to increase. TPCA gains of the plants in the treatment group were
affected by water stress and they were less than the TPCA gains of the plants in the control group. Baselines were established with a parametric approach using CWSI and coefficient of variation of TPCA (COV of TPCA) of the plants for early detection of the water stress. The baselines using only CWSI as an indicator for early water stress detection were found to be 0.14, 0.12, 0.20, and 0.10, and were 0.40, 0.55, 0.70, and 0.36 when only COV of TPCA was used as an indicator. The effectiveness of the sensing techniques was evaluated using timing of the stress detection by human. The CWSI based technique was able to detect the stress one to two days prior to the time of stress detection by human while the detection with TPCA based approach was found to be mostly 5 hours prior to the stress detection by human. Overall results of this study suggested that early and non-contact detection of plant water stress using CWSI was more successful and was quicker compared to the TPCA based water stress detection.
Dedicated

to My Wife, Özlem.
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Computer Data Acquisition and Control
Image Processing and Machine Vision
Computational Fluid Dynamics Applications
Plant Health Monitoring
Plant Physiology
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<td>AW</td>
<td>Available water</td>
<td>%</td>
</tr>
<tr>
<td>COV&lt;sub&gt;TPCA&lt;/sub&gt;</td>
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<td>C&lt;sub&gt;p&lt;/sub&gt;</td>
<td>Specific heat of the air at constant pressure</td>
<td>J kg&lt;sup&gt;-1&lt;/sup&gt; °C&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>CWSI</td>
<td>Crop water stress index</td>
<td>-</td>
</tr>
<tr>
<td>C&lt;sub&gt;i1,2&lt;/sub&gt;</td>
<td>Concentration entity i at location 1 and 2</td>
<td>mol m&lt;sup&gt;-3&lt;/sup&gt;</td>
</tr>
<tr>
<td>C&lt;sub&gt;1,2&lt;/sub&gt;</td>
<td>Concentration of substance in state 1 and 2</td>
<td>mol kg&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>C&lt;sub&gt;t&lt;/sub&gt;, C&lt;sub&gt;2&lt;/sub&gt;</td>
<td>Coefficients</td>
<td>-</td>
</tr>
<tr>
<td>D&lt;sub&gt;i&lt;/sub&gt;</td>
<td>Diffusion coefficient</td>
<td>m&lt;sup&gt;2&lt;/sup&gt;s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>d&lt;sub&gt;vt&lt;/sub&gt;</td>
<td>Absolute humidity at leaf liquid-vapor phase transition</td>
<td>kg&lt;sub&gt;vapor&lt;/sub&gt; m&lt;sup&gt;-3&lt;/sup&gt; air</td>
</tr>
<tr>
<td>d&lt;sub&gt;va&lt;/sub&gt;</td>
<td>Absolute humidity of air surrounding the leaf</td>
<td>kg&lt;sub&gt;vapor&lt;/sub&gt; m&lt;sup&gt;-3&lt;/sup&gt; air</td>
</tr>
<tr>
<td>e&lt;sup&gt;*(T&lt;sub&gt;c&lt;/sub&gt;)&lt;/sup&gt;</td>
<td>Saturation vapor pressure at leaf temperature</td>
<td>Pa</td>
</tr>
<tr>
<td>e&lt;sup&gt;<em>(T&lt;sub&gt;a&lt;/sub&gt;)&lt;/sup&gt;, e&lt;sup&gt;</em>&lt;/sup&gt;</td>
<td>Saturation vapor pressure at air temperature</td>
<td>Pa</td>
</tr>
<tr>
<td>e(T&lt;sub&gt;a&lt;/sub&gt;), e&lt;sub&gt;a&lt;/sub&gt;</td>
<td>Vapor pressure at air temperature</td>
<td>Pa</td>
</tr>
<tr>
<td>e&lt;sup&gt;0&lt;/sup&gt;</td>
<td>Vapor pressure of water in the system</td>
<td>Pa</td>
</tr>
<tr>
<td>e&lt;sub&gt;sa&lt;/sub&gt;</td>
<td>Vapor pressure of soil air</td>
<td>Pa</td>
</tr>
<tr>
<td>e&lt;sup&gt;*&lt;/sup&gt;</td>
<td>Vapor pressure of air at soil temperature</td>
<td>Pa</td>
</tr>
<tr>
<td>E</td>
<td>Evaporation</td>
<td>W m&lt;sup&gt;-2&lt;/sup&gt;</td>
</tr>
<tr>
<td>E&lt;sub&gt;I&lt;/sub&gt;</td>
<td>Radiation from both long wave and short wave</td>
<td>W m&lt;sup&gt;-2&lt;/sup&gt;</td>
</tr>
<tr>
<td>E&lt;sub&gt;O&lt;/sub&gt;</td>
<td>Heat loss by convection, conduction, and latent heat</td>
<td>W m&lt;sup&gt;-2&lt;/sup&gt;</td>
</tr>
<tr>
<td>E&lt;sub&gt;S&lt;/sub&gt;</td>
<td>Stored energy in the leaf resulting from photosynthesis and metabolic activity</td>
<td>W m&lt;sup&gt;-2&lt;/sup&gt;</td>
</tr>
<tr>
<td>ET</td>
<td>Actual evapotranspiration rate</td>
<td>kg m&lt;sup&gt;-2&lt;/sup&gt;s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>E&lt;sub&gt;TP&lt;/sub&gt;</td>
<td>Potential evapotranspiration rate</td>
<td>kg m&lt;sup&gt;-2&lt;/sup&gt;s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>f&lt;sub&gt;1,2&lt;/sub&gt;</td>
<td>Free energy of substance in state 1 and 2</td>
<td>J</td>
</tr>
<tr>
<td>g&lt;sub&gt;t&lt;/sub&gt;</td>
<td>Total leaf conductance to water vapor</td>
<td>m s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>g&lt;sub&gt;c&lt;/sub&gt;</td>
<td>Residual cuticular conductance</td>
<td>m s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>g&lt;sub&gt;max&lt;/sub&gt;</td>
<td>The stomatal conductance at maximum aperture</td>
<td>m s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>I&lt;sub&gt;s&lt;/sub&gt;</td>
<td>Shortwave radiation</td>
<td>W m&lt;sup&gt;-2&lt;/sup&gt;</td>
</tr>
<tr>
<td>J&lt;sub&gt;i&lt;/sub&gt;</td>
<td>Flux density or rate of mass transfer</td>
<td>mol m&lt;sup&gt;-2&lt;/sup&gt;s&lt;sup&gt;-1&lt;/sup&gt;</td>
</tr>
<tr>
<td>LAI</td>
<td>Leaf area index</td>
<td>-</td>
</tr>
<tr>
<td>LVPD</td>
<td>Vapor pressure deficit between leaf and air</td>
<td>Pa</td>
</tr>
<tr>
<td>n</td>
<td>Number of moles of substance</td>
<td>-</td>
</tr>
<tr>
<td>Symbol</td>
<td>Description</td>
<td>Unit</td>
</tr>
<tr>
<td>----------</td>
<td>-------------------------------------------------------------------------------</td>
<td>--------</td>
</tr>
<tr>
<td>( n_w )</td>
<td>Number of moles of water</td>
<td>-</td>
</tr>
<tr>
<td>( P )</td>
<td>Atmospheric pressure</td>
<td>Pa</td>
</tr>
<tr>
<td>( P_k )</td>
<td>Weather factor</td>
<td>-</td>
</tr>
<tr>
<td>PLWP</td>
<td>Plant leaf water potential</td>
<td>kPa</td>
</tr>
<tr>
<td>( P_{1,2} )</td>
<td>Pressure of substance in state 1 and 2</td>
<td>Pa</td>
</tr>
<tr>
<td>( Q_E )</td>
<td>Latent heat flux</td>
<td>W m(^{-2})</td>
</tr>
<tr>
<td>( Q_G )</td>
<td>The energy stored or released by biochemical reactions</td>
<td>W m(^{-2})</td>
</tr>
<tr>
<td>( Q_H )</td>
<td>Convective heat flux between plant leaves and the air</td>
<td>W m(^{-2})</td>
</tr>
<tr>
<td>( Q_{RAD, R} )</td>
<td>Incident solar radiation</td>
<td>W m(^{-2})</td>
</tr>
<tr>
<td>( R )</td>
<td>Standard gas constant (8.314)</td>
<td>J mol(^{-1}) K(^{-1})</td>
</tr>
<tr>
<td>( r_a )</td>
<td>Aerodynamic resistance</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_{ah} )</td>
<td>Air resistance for heat diffusion</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_{av} )</td>
<td>Resistance to water vapor transfer due to air around the leaf</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_c )</td>
<td>Canopy resistance</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_i )</td>
<td>Resistance to diffusion of entity i in a medium</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r^*_i )</td>
<td>Internal resistance of the canopy</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( \bar{r}_i )</td>
<td>Represents functions larger than unity</td>
<td>-</td>
</tr>
<tr>
<td>( r_m )</td>
<td>Minimal canopy resistance under potentially transpiring conditions</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_{min} )</td>
<td>Minimum possible canopy internal resistance</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_s )</td>
<td>Resistance to water vapor transfer due to the leaf or stomata</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( r_v )</td>
<td>Resistance to vapor diffusion</td>
<td>s m(^{-1})</td>
</tr>
<tr>
<td>( T )</td>
<td>Temperature</td>
<td>°K</td>
</tr>
<tr>
<td>( T_A )</td>
<td>Air temperature</td>
<td>°C</td>
</tr>
<tr>
<td>( T_C )</td>
<td>Plant canopy leaf temperature</td>
<td>°C</td>
</tr>
<tr>
<td>( T_C - T_A )</td>
<td>Leaf to air temperature differential</td>
<td>°C</td>
</tr>
<tr>
<td>( T_{LL} )</td>
<td>Theoretical lower limit to canopy temperature</td>
<td>°C</td>
</tr>
<tr>
<td>( T_{UL} )</td>
<td>Theoretical upper limit to canopy temperature</td>
<td>°C</td>
</tr>
<tr>
<td>TPCA(_i)</td>
<td>Top projected canopy area at time i</td>
<td>Pixels</td>
</tr>
<tr>
<td>TPCA(_{ave})</td>
<td>Daily average top projected canopy area</td>
<td>Pixels</td>
</tr>
<tr>
<td>V(_{PD, air})</td>
<td>Vapor pressure deficit of air</td>
<td>Pa</td>
</tr>
<tr>
<td>V(_S)</td>
<td>Total volume of the soil sample</td>
<td>m(^3)</td>
</tr>
<tr>
<td>V(_w, V_M)</td>
<td>Molal volume of water</td>
<td>m(^3) mol(^{-1})</td>
</tr>
<tr>
<td>( \bar{V}_w )</td>
<td>Partial molal volume of water</td>
<td>m(^3) mol(^{-1})</td>
</tr>
<tr>
<td>( x, y )</td>
<td>Image coordinates</td>
<td>-</td>
</tr>
<tr>
<td>( \alpha )</td>
<td>Parameter</td>
<td>-</td>
</tr>
<tr>
<td>( \alpha_{th} )</td>
<td>Heat diffusion coefficient</td>
<td>-</td>
</tr>
<tr>
<td>( \alpha_w )</td>
<td>Water vapor diffusion coefficient</td>
<td>-</td>
</tr>
<tr>
<td>( \varepsilon )</td>
<td>Molar conversion from water to dry air</td>
<td>-</td>
</tr>
<tr>
<td>( \gamma )</td>
<td>Thermodynamic psychrometric constant</td>
<td>Pa °C(^{-1})</td>
</tr>
<tr>
<td>( \lambda )</td>
<td>Latent heat of vaporization</td>
<td>J kg(^{-1})</td>
</tr>
<tr>
<td>Symbol</td>
<td>Definition</td>
<td>Unit</td>
</tr>
<tr>
<td>--------</td>
<td>------------------------------------------------------------------------------------------------------</td>
<td>---------</td>
</tr>
<tr>
<td>$\lambda_{1,2}$</td>
<td>Image threshold levels 1 and 2</td>
<td>-</td>
</tr>
<tr>
<td>$\Delta, \delta$</td>
<td>The slope of saturated vapor pressure-temperature curve</td>
<td>Pa °C$^{-1}$</td>
</tr>
<tr>
<td>$\rho, \rho_a$</td>
<td>Density of the air</td>
<td>kg m$^{-3}$</td>
</tr>
<tr>
<td>$\rho_s$</td>
<td>Dry bulk density of the soil sample</td>
<td>g m$^{-3}$</td>
</tr>
<tr>
<td>$\mu_w$</td>
<td>Chemical potential of water in the system</td>
<td>J kg$^{-1}$</td>
</tr>
<tr>
<td>$\mu_w^0$</td>
<td>Chemical potential of pure water at atmospheric pressure at the same temperature as the system which is under consideration</td>
<td>J kg$^{-1}$</td>
</tr>
<tr>
<td>$\mu_{TPCA}$</td>
<td>Mean of top projected canopy area</td>
<td>Pixels</td>
</tr>
<tr>
<td>$\psi$</td>
<td>Water potential</td>
<td>Pa</td>
</tr>
<tr>
<td>$\psi_{SWP}$</td>
<td>Soil water potential</td>
<td>kPa</td>
</tr>
<tr>
<td>$\sigma_{TPCA}$</td>
<td>Standard deviation of top projected canopy area</td>
<td>Pixels</td>
</tr>
<tr>
<td>$\theta_G$</td>
<td>Gravimetric soil moisture content</td>
<td>g g$^{-1}$</td>
</tr>
</tbody>
</table>
CHAPTER 1

INTRODUCTION

Plant growth and productivity are directly related to the availability of water to the plants, while only 1-2% of the water taken up by plants is utilized for metabolic activity. Most water evaporated at plant surface is water that passes through the plant, entering at the roots, passing through the vascular tissue to the leaves or other plant parts and exiting into the surrounding air via stomata. The process of evaporation of water from plant to the surrounding air is called transpiration. Transpiration furnishes the driving force for water movement through the plant. It is "the evaporative cooling mechanism of plants." Because large quantities of energy are required in the change of phase from liquid to vapor, evaporation provides a very efficient mechanism for the dissipation of heat. Under greenhouse conditions, about one-third of incoming solar energy is converted to latent heat in the process of evapotranspiration (Mankin et al., 1998). Plants must withdraw enough water to meet the demand for transpiration. Plant water stress and eventually wilting occur when the demand (transpiration) exceeds the supply (available water for the plant). In order to accomplish a proper water management practice in controlled environment plant production, one must know when to irrigate the
plants and how much to irrigate. To answer the question "when," it is necessary to detect plant water stress as early as possible.

Frequent and excessive irrigation to the plants may increase the relative humidity in the greenhouses. Thus, growers sometimes need to heat the greenhouse to reduce the relative humidity. This increases the cost for heating. When early water stress detection is achieved and irrigation is based on measurements obtained from the plants, the frequency and the amount of water applied to the plants might be less compared to the amounts when the plants are irrigated several times during the day. Therefore, the cost for heating will be reduced as well as the cost of pumping energy for water.

The importance of early and quantitative plant water stress detection can also be emphasized from a different perspective. More and more people are consuming nutritional supplements as a way of meeting their daily dietary needs, preventing disease and improving their quality of life. Traditional medicines derived from plants, herbs and other natural sources are being sought as alternatives to modern medicine and pharmaceuticals. There are many herbs and ornamental plants used as medicinal plants. Among many are English Daisy (*Bellis Perennis*) flowers good for the liver, Sweet Basil (*Ocimum Basilisum*) used for kidney troubles, Pepper Mint (*Mentha Piperita*) leaves used for loosening uterine cramps and strengthening the nerves, and Pot Marigold (*Calendula Officinalis*) heals wounds as well as internal and external ulcers (Gabriel, 1979; Oster, 1991; Herbal Guide, 1999). Most of these plants are also rich in oil and fat content, starch, sugar, and protein content. Phytochemicals (bioactive plant chemicals) and enzymes used as raw material in nutraceutical and pharmaceutical industry, and oil,
sugar, starch, and proteins used in food and feed industry, are the products of secondary metabolisms of the plants.

Secondary metabolism can be promoted through cultural practices. Evidences of increased secondary metabolism due to lower resource availability (water, nutrient, light), was reported by Herms and Mattson, 1992. They illustrated that the change of secondary metabolism of the plants under low resource availability.

The most common metabolic response to water stress appears to be the accumulation of amino acids, prominently proline, sugars, cyclitols, betaine, choline, and inorganic ions (Waterman and Mole, 1989). Controlled experiments confirmed that water stress has been shown to increase alkaloid percentages in a variety of plants. Experiments on cucumber (Cucumis Sativus) seedlings showed that water stress increased cotyledon flavonoid content. Water stress in peppermint (Mentha Piperita) have been shown to increase the concentration of essential oils (Gershenzon, 1984; Charles et al., 1990). Singh-Sangwan et al. (1994) reported that the major oil constituents, geraniol and citral, increased as water stress increased on lemongrass. Phenolic content of pepper leaves (Capsicum Annuum) was increased 50-60% by mild water stress (Estiarte et al., 1994). Furthermore, Rhizopoulou and Diamantoglou (1991) showed that soil moisture deficit significantly increased essential oil content and total lipids in perennial herb, Origanum Majorana.
Water stressing plants to a limit that will not affect plant growth and yield may help increase the amount of useful phytochemicals, oil, sugar, starch, vitamins, proteins and carbohydrates extractable from the plants, and thus nutritional quality and marketability of the plants (Sorensen et al., 1997; Foyer et al., 1998; Barron and De Mejia, 1998). However, maintaining a certain water stress level to benefit the secondary metabolite production without killing the plants requires development of an early, plant-response-based, and most importantly a quantitative stress detection technique.

Chemical growth retardants used to control plant growth are either naturally occurring plant hormones or synthetically produced compounds. However, there are strict regulations for the use of chemical growth retardants in plant production. Water stress can modify plant development and morphology. Water stress may reduce cell enlargement of the plants causing inhibited growth and canopy expansion. Water stress also thickness cell walls making the cell wall structure of the plant stronger. There is also some evidence that water stress may result in flower initiation. Therefore, withholding water from the plants to a certain extend as a growth regulator may be a cheaper and more environmentally friendly alternative to some of the chemical growth regulators.

There has been much interest in developing quick methods for evaluating water stress level to which a plant is subjected. Methods such as measurement of soil water tension, leaf water potential, and sap flow have been widely used. While soil water level indicates the water status of growing media rather than providing the water status of the plants, it is only a measure of water supply. Leaf water potential and sap flow measurements do provide direct information about plant’s water status, nevertheless, contact measurements are required and only limited samples may be collected. These
approaches may be scientifically sound, yet it will be very difficult to implement for large-scale commercial productions. Therefore, quantitative, non-contact, and plant-response-based water stress detection is desirable.

The main focus of this dissertation was to establish a methodology for early, non-contact, non-destructive, and quantitative detection of plant water stress with applications of infrared thermometry using crop water stress index (CWSI) and image processing using top projected canopy area (TPCA) of the plants.

Chapter 1 provides the impetus of this dissertation. Problems and concerns with the existing water stress detection techniques are discussed. Chapter 2 presents the specific objectives of this study. Chapter 3 reviews the previous and current state of the plant water stress detection. Leaf temperature and its relation to the water status of the plants, measurement techniques for determining water stress and plant water status is reviewed. Applications of image processing and machine vision techniques to detect plant water stress are also presented. Chapter 4 provides the theoretical background, methodology and experimental procedures used in this study. Chapter 5 presents the results and discusses the data obtained. Chapter 6 summarizes the major conclusions of this study especially with reference to the objectives, and recommendations and concerns are made for future research work. Appendix A provides experimental procedures and the results for determination of soil water release curve of soil medium used in the experiments. Experimental procedures for Theta-probe (soil moisture sensor) calibration and results are furnished in Appendix B. Appendix C provides computer source codes for continuous plant health monitoring system. Source code for image processing of experimental plant images is presented in Appendix D. Appendix E provides two sample
T-test results for evaluation of water stress effect on TPCA gains of the plants. The results of the normality tests for crop water stress index (CWSI) and coefficient of variation of top projected canopy area (COV of TPCA) are provided in Appendix F. The descriptions and units for the abbreviations and symbols used throughout the dissertation are provided in the list of symbols section.
CHAPTER 2

OBJECTIVES

The primary goal of this study was to establish methodology for early, non-contact, and quantitative detection of plant water stress using crop water stress index (CWSI) and the changes in top projected canopy area (TPCA). To achieve this goal, the specific objectives of this study were to:

1. Design and develop an automated computer-controlled continuous plant health and growth monitoring system to conduct the experiments,
2. Develop a crop water stress index model for plants grown under controlled environment conditions,
3. Develop a methodology using top projected canopy area change of the plants for early water stress detection,
4. Establish quantitative baselines with crop water stress index and top projected canopy area change of the plants for early water stress detection, and
5. Evaluate the effectiveness of using crop water stress index and top projected canopy area for early water stress detection.
CHAPTER 3

LITERATURE REVIEW

3.1. Water transport in the plant

The maintenance of adequate water supply to plants is crucial to obtain maximum productivity. This is because water is an essential element for maintaining a normal physiological activity and membrane transport processes. Water is also the medium for long distance transport of nutrients.

The water potential of the atmosphere is usually lower than the water potential of the soil. This difference in water potential is the driving force which causes the translocation of water from the soil solution through the plant to the atmosphere. In general, water potential in the leaf is not much lower than that in the soil. Nonetheless, a large potential difference occurs across the boundary layer around the leaf and stomatal cavities and the atmosphere. The rate of water transfer across the leaf and atmosphere interface is proportional to the vapor pressure difference between both sides of the boundary. Water has to overcome a number of resistances on its path from the soil through the stem to the leaves.
Water exists as a liquid during the transport from the soil through the cortex of the roots and stem to the substomatal cavities, and then as vapor across the leaf epidermis and through the boundary layer to the atmosphere above the plant's canopy (Figure 3.1).

Figure 3.1: Water transport pathway in the plant (Mengel and Kirkby, 1982)
When radiation energy strikes the leaves, the stomata open to promote CO₂ assimilation and water is lost by transpiration process. As water loss accelerates, leaf water content is reduced. This reduction causes a reduced or lowered leaf water potentials. The lowered leaf water potential then causes the withdrawal of water from other plant tissues and soil.

During the dark period, water is absorbed by well-watered plants only in small quantities which provides enough water for cell enlargement and for replacement of water lost through cuticular flows of vapor or through pressure-induced flows, called *guttation*.

Transpiration rate is controlled by the rate at which energy is supplied to the leaf. During the midday, the plants rehydrate because the uptake of water from the soil is at greater rate than the water loss through transpiration process. If the plants are under moderate water stress conditions, transpiration is reduced to the available supply rate and it increases the leaf temperature.

3.2. Plant temperature and water status of plants

Plant temperature is an important indicator of the response of the plant to environmental factors such as solar radiation, air temperature, air movement, and water availability. Plant leaf temperatures have long been related to plant transpiration, leaf water potential, and soil water deficit.
Gates (1964) determined the energy balance for a single leaf and concluded that transpiration was important for keeping the temperature of the fully sunlit leaves within the terminal limit. It was indicated that a small amount of transpiration could make a difference of a few degrees in plant temperature which can, under hot conditions, mean the difference between survival or thermal death. It was reported that for each $1.7 \times 10^{-4}$ g cm$^{-2}$ min$^{-1}$ of transpiration, the effective radiation load on the leaf is reduced by 0.10 cal cm$^{-2}$ min$^{-1}$. The author claimed that this amount of transpiration would drop the leaf temperature by 5 °C below that which would occur in the absence of transpiration in still air by 4 °C in wind at 1 mph, 2.5 °C at 5 mph, or by about 1 °C at 15 mph. This study showed good agreement between the energy absorbed by the leaf and the energy consumed by dissipative mechanism. According to Reicosky et al. (1980), the relative water content of plant leaves can be reduced 60 to 85% by plant water stress resulting in increased leaf to air temperature differences as high as 6 °C depending on the level of water stress.

Wiegand and Namken (1966) studied the influences of cotton plant relative turgidity (difference between fresh field weight and 60 °C oven-dry weight of leaves divided by the weight difference between same leaves after they had been floated on water overnight under illumination and their 60 °C oven-dry weight), solar radiation, and air temperature on plant leaf temperature and leaf to air temperature differential. It was pointed out that leaf temperature of the plant varied as environmental conditions and the availability of moisture for transpiration varied. They indicated that variations in plant moisture stress significantly altered leaf temperature and leaf to air temperature differential. The study showed that a decrease in relative turgidity from 83 to 59 percent
resulted in a 3.6 °C increase in leaf temperature, and the same change resulted in 2.7 to 3.7 °C increase in leaf to air temperature differential.

Leaf water potential, leaf diffusion resistance, and leaf-air temperature differences (measured with infrared thermometer) of well-watered and water-stressed peas were compared (Clark and Hiler, 1973). Their study showed that leaves of well-watered peas were slightly cooler than the air temperature whereas leaves of stressed plants were 2 to 3 °C warmer than leaves of well-watered plants. They indicated that when leaf water potential decreased, leaf diffusion resistance increased because of loss of turgor in the guard cells and leaf to air temperature differential increased due to the reduction in transpiration rate.

Ehrler (1973) investigated feasibility of using leaf to air temperature difference measurement (with thermocouples) as a guide for irrigation scheduling. The study reported that leaf to air temperature difference varied from -3 to 2 °C depending on the degree of soil water depletion. His data also showed an inverse linear relationship between the leaf-air temperature difference and the vapor pressure deficit of the air. However, the study indicated that the measurement of individual leaf temperatures with thermocouples was awkward and tedious, and many leaves must be sampled to obtain an average for the whole canopy.

Ehrler et al. (1978a) studied the relationship between wheat canopy temperature and plant water potential to investigate the true nature of the leaf to air temperature differential response. Their data showed that leaf-air temperature difference responded specifically to changes in plant water potential which in turn were elicited by changes in volumetric soil water content in the root zone. Their study reported that transpiration
began to decrease significantly at volumetric soil moisture levels lower than 0.20 resulting in rapid changes in leaf to air temperature difference as leaf water potentials decreased below -19 bars (the leaf to air temperature difference was 0 °C at -19 bar leaf water potential). He claimed that the leaf to air temperature difference method for monitoring plant stress in wheat would be reliable provided that frequent measurements of leaf-air temperature difference be made. The results from Ehrler et al. (1978b) also supported these conclusions.

Changes in stomatal conductance may cause alterations in leaf water potential by changing the transpiration rate. Farquhar and Sharkey (1982) stated that the dependence of leaf temperature on stomatal conductance was via transpiration rate. It was stated that attention should be paid to heat balance in assessing the effect of transpiration rate on assimilation rate at high temperatures and low wind speeds because electron transport and photo-phosphorylation are sensitive at high temperatures. They suggested that advances in infrared thermometry might aid in this direction.

Jones and Sutherland (1991) proposed that the prime role of stomata might be to avoid damaging plant water deficits. Jarvis and Davies (1998) denoted that the control of photosynthesis and transpiration processes represents a central role for stomata and stomatal aperture. Jones (1998) argued that another possibility is that stomatal control of transpiration has a role in maintaining leaf temperatures within an optimal range and the author claimed that stomatal closure particularly because of variation in leaf conductance may be detected as variations in leaf temperatures.
Choudhury (1983) and Choudhury and Idso (1984) investigated the effect of soil water status and root zone water potentials on canopy temperature of the corn and sunflower, respectively. It was reported that leaf temperature of the crop increased as available soil moisture decreased. The studies concluded that leaf temperature could be used as an indicator of soil water status.

Hatfield *et al.* (1985) reported good agreement between leaf to air canopy temperature differential of cotton crop and vapor pressure deficit of the air (VPD_{air}) under unlimited water supply conditions. The leaf-air temperature difference was inversely and linearly related to VPD_{air} owing to the fact that as the transpiration rate increased the temperature of the leaf was decreased due to latent heat loss from the leaf and caused more cooling effect.

Yang (1988) studied the transport processes, plant responses to environmental factor with a dynamic model for greenhouse grown cucumbers. He found that leaf temperatures of cucumbers grown under unlimited water supply conditions were consistently lower than the air temperature due to the high transpiration rate of the plants.

Al-Shooshan *et al.* (1991) evaluated the evapotranspiration of greenhouse grown Chrysanthemum crop under shaded and unshaded conditions. They reported that major cultural differences caused by the two cultural approaches were in evapotranspiration and leaf temperature. The data showed that evapotranspiration of the unshaded crop was approximately three times than that of the shaded crop. The leaf temperature of the shaded crop was found to be 3 to 4 °C below the air temperature and generally equal to the air temperature when unshaded. Al-Arifi (1999) reported that both leaf temperatures of Chrysanthemums at the top and within plant canopy were consistently lower than the
above air temperatures under 50% shade while top leaf temperatures were approximately 1 °C higher than air temperatures during high solar radiation and with air velocities of 0.1 m s⁻¹ above plant canopy. It was also reported that top leaf temperatures were nearly or slightly lower than air temperatures when air velocity was 0.6 m s⁻¹ above the canopy.

Dynamic analysis of water uptake by New Guinea Impatiens was investigated by Pang (1992). He indicated that New Guinea Impatiens were light saturated at solar irradiance above 250 W m⁻². As a result of this, at higher levels of irradiance, the exposed leaf temperature was up to 2-9 °C higher than the surrounding air temperature. The study concluded that the maximum stomatal opening and the ability of the system to move water through the plant when grown under high light intensity conditions limited the transpiration rate.

Seginer (1994) evaluated the transpirational cooling of a greenhouse crop with partial ground cover. He concluded that if water supply to the stomata was not limiting, for given ventilation rate, the canopy temperature of a sparse crop should normally not be higher than that of a dense crop. He pointed out that for a given canopy temperature, sparse plants transpire more per unit leaf area, due to micro-advection of energy surplus from the surrounding bare and dry ground.
3.3. Measurement techniques to determine plant water status and water stress

Water is an indispensable medium for plants as well as any life form and its importance to plant growth and yield is well recognized. Water requirement is the minimum amount of water required to provide optimal yield. Plant water status is the quantification of the condition of water in a plant relative to its requirement (Spomer, 1985). There has been much interest in developing quick methods for evaluating the level of water stress on plants. The measurement of plants water status and water stress has advanced from visual measurements to a variety of increasingly complex instrumental measurements and methods.

3.3.1. Visual observations

Rolling of leaves and leaf color change are visual indicators of plant water stress. O'Toole and Cruz (1980) reported that a good correlation existed between leaf rolling, stomatal resistance, and leaf water potential in rice.

3.3.2. Water content measurement

Water content measurement based either on fresh or dry weight basis was the first quantitative method to measure water stress. Water content measurement was found to be inadequate because fresh weight often varies widely during the day and dry weight also varies measurably because of photosynthesis (Kramer, 1990). The difficulties in these methods resulted in development of other methods of expressing the water content. One of the alternatives was using relative water content (Sinclair and Ludlow, 1985).
3.3.3. **Leaf water potential measurement**

Attempts have been made to monitor changes in leaf water potentials. Clark and Hiler (1973) compared plant measurements including leaf-water potential and leaf diffusion resistance as indicators of water deficits in pea. The results of the study indicated that leaf-water potential as measured by pressure chamber method was more responsive to changes in plant water status than leaf diffusion resistance. They reported that leaf-diffusion resistance was an awkward measurement because two different determinations were necessary and its value varied considerably with leaf temperature. It was said that the leaf-water potential measurement approach was found to be better. Furthermore, using this method, change in water deficits was more pronounced during vegetative stage than the pod development stage.

Oosterhuis *et al.* (1987) studied the relationship between pea leaf water potential, as measured with a pressure chamber, and a change in leaf coloration as an indication of water stress. Green color and bluish color were rated to be classifiers of well-watered and water stressed pea plants. The study indicated that the blue coloration of the crop was associated with the decrease in leaf water potential. They reported that the color change on plants in the stressed treatment occurred six days after irrigation was withheld.

Pre-dawn leaf water potential as an indicator of plant water stress on maize crop was studied by Laker *et al.* (1987). Their study showed that under moderate climatic conditions, pre-dawn leaf water potentials were found to be a suitable method for detection of water stress. However, under conditions of very high evaporative demand, plant wilted early in the morning even in soils that were almost at field capacity. They

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also reported that pre-dawn leaf water potentials showed very large fluctuations especially in the relatively dry soil water range.

Rana et al. (1997) investigated the effect of environmental, soil, and plant parameters to model crop evapotranspiration. Plant leaf water potential (PLWP) was evaluated by collecting ten leaves from each plant used. It was found that the decrease in PLWP with available water (AW) depletion, (AW was defined as the amount of water in the soil normalized by the total water availability between field capacity and wilting point), was clear for the crops used. The results also revealed that PLWP started to decrease when AW was about 50%. PLWP was indicated to be a plant-soil interface characteristic and it was only species dependent while canopy resistance depends on the soil moisture, a physical parameter depend on the soil characteristics. Thus, it was suggested that canopy resistance could be linked to soil water availability through PLWP (Rana et al., 1998). The study indicated that measurement of plant leaf water potential (PLWP) is not very easy to measure and the researchers commented the method may only be suitable for research centers.

3.3.4. Soil moisture content and tension measurement

Soil moisture content is a measure of the actual water content, and is defined as the percentage volume of a moist soil occupied by water. Soil moisture potential (soil water tension) on the other hand is an indirect measure of water content, and it is the energy necessary to extract water from the soil matrix. Soil moisture content measurements have been made in different ways and its relation to plant water status, water use, and plant growth has also been evaluated. Van Bavel (1966) evaluated the
changes in canopy resistance to water loss by soil water depletion. The results of the study showed that the soil water potential in the root zone was estimated at -4 bars when stomatal control of evaporation became first noticeable. It was pointed out that further decrease of soil water potential caused sharply rising daytime canopy resistance values that was supported by the evidence of the relationship between soil water availability and water use by the plants.

Acevedo et al. (1971) found that maize leaves showed reduced elongation at soil moisture tension as low as 20 kPa. They reported, however, that mild and short stress resulted in transitory, rapid growth after irrigation that made up for the reduced growth which occurred under water stress conditions. It was also suggested that the metabolic processes necessary for growth were not affected by the water stress and that growth after watering was only the result of a delayed event. The growth of maize leaves was found to be irreversibly reduced at 250-300 kPa soil moisture tensions.

Karlovich and Fonteno (1986) studied the effect of soil moisture tension and soil water content on the growth of chrysanthemum plant. The results revealed that soil water content might be more significant than soil moisture tension in affecting chrysanthemum plant growth under greenhouse conditions. They implied that the tension at which plants should be watered might vary depending on growth medium.

Growth of chrysanthemum and the amount of water applied to plants throughout the experiment was analyzed by comparing irrigation systems controlled by soil moisture tension and by time (5 min day⁻¹) (Lieth and Burger, 1989). The study revealed that soil moisture tensions of -7.5 to -15 kPa caused reductions over the time based treatment in fresh and dry weights of leaves and stem, but had no effect on cropping time. It was
found that stem length was reduced in those plants exposed to -7.5 to -15 kPa tension levels, however the resulting cut flowers were still long enough to satisfy commercial demands. The amount of water usage in the tension-based watering was 8 to 24% of the amount of water used in the time based irrigation operation.

Abdel-Rahman et al. (1994) studied the effect of soil moisture tension on the growth of greenhouse tomato plants. The data showed that even though the selected soil moisture tension levels (8, 13, 23 kPa) were in within the allowable range for water field capacity, there was significant difference between the dry weights of the plant, suggesting that differences in soil moisture tension affected the rate of tomato growth even if the soil moisture tension was in the field capacity range.

Sinclair et al. (1998) studied extractable soil water content and its relationship to transpiration rate of soybean on sandy soil. It was pointed out that plant gas exchange is unaffected by soil dehydration until the soil dries to less than 30% of the extractable soil water.

### 3.3.5. Sap flow measurements

Water use and transpiration rates for whole plants can also be determined by techniques which measure the rate at which sap ascends stems. These methods use heat as a tracer for sap movement, but they are different in their operation principles. There are two methods commonly used, the stem heat balance and trunk sector heat balance methods, use the heat balance principle (Smith and Allen, 1996).
Cohen and Li (1996) mentioned that sap flow measurement has been widely used recently in studying plant response to the environment, but added that results have not been satisfactory. Possible reason for this was suggested to be the non-uniform distribution of conducting elements in the stem cross-sectional area introducing error in the measurements.

3.3.6. Stem diameter change measurements

Another way of determining the plant water status is to measure the change of the stem diameter. McBurney and Costigan (1988) elaborated on the use of displacement transducers to measure the change in stem diameter in relation to plant water potential changes and water stress conditions. It was pointed out that a major difficulty with this technique was how to account for any growth component in stem diameter measurement. The study recommended the use of mathematical relationship to correct stem diameter changes related to growth. However, they recommended that further work was necessary to test if the growth correction procedure needed to be modified to account for any possible temporary inhibition of growth due to water stress when evaporative demand was high.

Calado et al. (1990) also investigated the use of stem diameter change with displacement transducers to determine its relation to the water status of tomato plants. The results showed good relationship between diurnal stem diameter changes and leaf water potentials. However, it was indicated that a considerable number of sensors did not work well because they were inadvertently left on the unprotected ground for several
days before their installation on the tomato plants. Therefore, it was suggested that dust increased friction in the slider probably causing inaccuracies.

3.4. What should be measured to assess plant water stress?

Methods such as soil water tension, leaf water potential, and sap flow have been widely used among others to assess plant water status and water stress. However, soil water content indicates the water status of growing media rather than providing the water status of the plants, it is only a measure of water supply. Plants in moist soil often develop temporary water stress and wilt on hot, sunny days when transpiration is rapid, but may exhibit only slight stress on cool, cloudy days, even when the soil is relatively dry. Therefore, measurement of soil water content to determine plant water stress cannot account for the differing abilities of plant roots to penetrate the soil and extract water from it nor predict the influence of temporary rapid increases in evaporative demand occurring during the course of each day (McBurney, 1988; Kramer, 1990).

Leaf water potential and sap flow measurements to determine plant water stress do provide direct information about plant’s water status, nevertheless, contact and destructive measurements are required and only limited samples can be collected. Such approaches may be scientifically sound, but are very difficult to implement in large-scale commercial plant production systems.

One alternative of detecting plant water stress is to develop a sensing technique to monitor the plants themselves. A desirable sensing technique shall be non-contact, non-destructive, and plant-response-based. Possible sensing techniques include an infrared
thermometry based plant temperature measurement for crop water stress index determination and top projected canopy area change using image processing applications. These two techniques may be used to continuously monitor plant temperature and biotic movement of plants for plant water stress assessment.
3.5. Crop water stress index research

3.5.1. Empirical approach

Plant leaf temperature have long been conceded as a potential information about plant water stress (Tanner, 1963; Ehrler et al., 1978a; 1978b). Earlier work in this field related leaf temperatures to ambient temperatures by suggesting if leaf-ambient air difference was negative, the plants were well-watered, if positive, the plants were to be irrigated. This way of addressing plant water stress was called stress-degree-day (Idso et al., 1977). However, the effects of transient and spatial variations in plant microclimate were not easily explained by the leaf-air differential. Therefore, Idso et al. (1981a) developed an alternative plant water stress index called crop water stress index (CWSI), which normalized the stress-degree-day parameter for environmental variability. The concept of the index depends upon the relationship which exists between the canopy-air temperature difference and the air vapor pressure deficit under non-water limiting conditions which means that the plants are transpiring at a potential rate. This relationship was denoted "non-water-stressed baseline" because of its linearity and the fact that it was obtained from well-watered crops. Ehrler (1973) first reported this linear relationship between leaf-air temperature difference and vapor pressure deficit of the air for a well-watered canopy.

The empirical determination of CWSI is based upon defining baselines that symbolize a crop completely stressed and a crop that is well-watered as illustrated in Figure 3.2. From the figure, it can be seen that the lower sloping-baseline, denoted "well-watered," represents the maximum rate of transpiration of a well-watered plant and it has
been shown that $T_C - T_A$ declines as $VPD_{air}$ increases. $T_C - T_A$ of a totally water stressed plant (theoretically speaking, with no transpiration) as illustrated with upper baseline, denoted "stressed" in the figure, is independent to $VPD_{air}$. Therefore, having identified these baselines, one can gauge water stress level from measured $T_C - T_A$, which falls between these two baselines, for a plant or a canopy and its deviation from its “ideal” counterparts. The consequent index deviates from 0 (potentially transpiring plant) to 1 (plant having no transpiration).

Figure 3.2. A schematic representation of empirical calculation of crop water stress index (Gardner et al., 1992).
In order to calculate CWSI, deviation of actual $T_C - T_A$ from lower baseline is computed relative to the upper baseline as:

$$CWSI = \frac{LM}{KM} = \frac{(T_C - T_A) - (T_{UL} - T_A)}{(T_{UL} - T_A) - (T_{UL} - T_A)} = \frac{T_C - T_{UL}}{T_{UL} - T_{LL}}$$  \hspace{1cm} (3.1)

Lower baseline is determined from temperature measurements of a well-watered plant throughout a clear sky day to obtain a range in $VPD_{air}$, and then a linear regression line is fit through the data to acquire the baseline. The upper limit is determined from working with the lower line as a following manner (Idso et al., 1981a; 1981b; Idso, 1982). As it is expected from Figure 3.2, when $VPD_{air}$ decreases, the driving force for transpiration, which is the vapor pressure gradient between leaf and air ($LVPD$), also decreases resulting in reduced transpiration rate and raise in $T_C - T_A$. If the intercept of $T_C - T_A$ vs. $VPD_{air}$ is known, $T_C$ can be calculated for a specified $T_A$. And, then vapor pressure difference between a plant and air ($LVPD$) is calculated for the empirically observed temperature difference at $VPD_{air}$=0. When $LVPD$ is zero, transpiration must be ceased. One way of achieving this is to supersaturate the air. If the $VPD_{air}$ is extended into the negative region to the point where the absolute value of $LVPD$ is matched, the point can be read as $T_{UL} - T_A$ at which the plant experiences under no transpiration conditions.
Empirically derived CWSIs have been used by researchers to detect plant water stress, to predict yield, plant quality, plant water use, and used for irrigation scheduling.

Pinter et al. (1983) used an empirically derived crop water stress index and its relation to yield for cotton production management. They indicated that CWSI responded as expected dropping to low levels following irrigation and increasing gradually as the cotton plants depleted soil moisture reserves. The data also showed that the final yield of seed cotton was significantly inversely correlated with the average CWSI observed.

O’Toole et al. (1984) compared different crop water stress measurement methods including empirically calculated CWSI, leaf water potential, stomatal resistance, net photosynthesis, and transpiration rate for rice crop. The data showed that the CWSI method was nearly three times faster than the gas exchange rate technique and about two and half times faster than the leaf water potential measurement method. They also reported that the CWSI values were highly correlated with mean photosynthetic rate measurements.

Kanemasu and Asrar (1985) incorporated remotely sensed crop canopy temperatures into a model for wheat growth and yield. They indicated that a continuous daily monitoring of the canopy temperature could provide information as the onset of stress and a quantitative estimate of the extractable soil water content, and the data could supply valuable feedback information for computer simulation models. However, plant leaf temperature measurements along with environmental measurements can be incorporated into a model which estimates the level of plant water stress. Then, it would be more valuable relating the model results to the extractable soil water content.

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Using infrared thermometry to measure single leaf temperatures and empirically derived CWSI to quantify water stress in sunflowers were reported by Nielsen and Anderson (1989). Plants were grown under an automated rainout shelter and subjected to four water treatments ranging from 0 to 100% replacement of evapotranspirational losses. The study also investigated the relation between calculated CWSI and stomatal conductance, CO₂ exchange rate, leaf water potential, transpiration rate, and percent available water in the active root zone. The data showed statistically significant correlations between CWSI and these parameters. They indicated that as the stomata closed in response to decreased availability of soil water and lower leaf water potential, transpiration rates declined, leaf temperature increased, and CWSI increased. Furthermore, in response to increased water stress, leaf CO₂ exchange rate declined. Finally, they concluded that this technique has potential to rapidly determine presence and severity of water stress for broad leaf plants that are closely or widely spaced.

Hattendorf et al. (1990) used an empirically derived CWSI and evaluated the relationship between CWSI and stomatal conductance of water stressed alfalfa. The study showed that stomatal conductance (reciprocal of stomatal resistance) decreased as crop water stress index increased. Jalali-Farahani et al. (1994) also found a well defined curvilinear relationship between canopy resistance and CWSI for turf. Canopy resistance was increased as CWSI increased.

Calado et al. (1990) studied the effects water stress on drip-irrigated processing tomatoes. CWSI was also implemented in the experiment to automate irrigation. The data showed that even though there was variability in the calculated CWSI, this method appeared to be useful to schedule and control irrigation, and also characterize water stress
in tomato plants. Furthermore, their data also showed that as water stress index increased, soluble sugar increased by nearly two degrees Brix.

Garrot et al. (1994) investigated the relationships among CWSI and irrigation scheduling levels, grain production, grain quality, and water applied. Irrigation of wheat crops was scheduled with different CWSI values ranging from 0.2 to 0.8. The results revealed that highest grain production was attained when irrigation was scheduled at CWSI values of 0.37. It was reported that scheduling irrigations with lower CWSI values did not increase grain production, but required more water. Furthermore, the study showed that scheduling irrigation at CWSI values higher than 0.37 reduced grain production.

Anconelli et al. (1994) evaluated a CWSI value which processing tomatoes may withstand. They investigated if this value, as a threshold, could serve for irrigation scheduling so as to obtain high yields of products suitable for processing. CWSI was inversely correlated with marketable yield, average fruit weight. Quantitatively, the data also showed that there was a significant regression between CWSI and the weight of the vegetative part of the plant. This data demonstrated a close relationship between leaf temperature, transpiration capacity and therefore organic matter build-up. They concluded that infrared temperature measurement employed as a guideline for irrigation scheduling permitted overall to obtain higher marketable yields with a moderate decrease in optical residue, thus giving an improved tomato paste yield.
Olufayo et al. (1996) evaluated sorghum yield, water use, and remotely sensed canopy temperatures under different levels of irrigation regimes. They evaluated the effect of solar radiation and wind speed in determination of non-water stressed baselines. They reported that a scatter was observed in leaf-air temperature vs. air vapor pressure deficit relationship. They explained the reason for this scatter as mostly the effect of wind speed, but not solar radiation. Higher correlation was obtained for low wind speeds.

Wanjura and Upchurch (1998) compared different procedures for quantifying crop water stress that included the use of canopy temperature for corn, soybean, and cotton, and related CWSI to amount of applied water and crop yield. They used both theoretically derived and empirically determined CWSI values. The data showed that the most accurate values of CWSI were computed with the empirical procedure that used the measured canopy temperature of plants in the highest water regime to represent a non-water-stressed crop. The study reported that all CWSI values and all water level treatments were linearly related with yield in cotton and soybeans, but nonlinear relationship was observed for corn.

Empirical determination of crop water stress index is based on defining upper and lower baselines which symbolizes a completely water stressed and a well-watered plant. Literature showed that leaf to air temperature differential is inversely and linearly related to $\text{VPD}_{\text{air}}$ when the plant is well watered and it is independent to $\text{VPD}_{\text{air}}$ when the plant is totally water stressed meaning that with no transpiration. Empirical approach by determining lower and upper baselines for leaf to air temperature difference to assess crop water stress index does not account for the environmental variability and its effect
on determination of the baselines. This is a shortcoming in the empirical CWSI approach. Therefore, it is necessary to account for environmental variables in the calculation of crop water stress index.
3.5.2. Theoretical approach

Shortly after the empirical approach was proposed, Jackson et al. (1981) introduced a theoretical approach that accounted for the influence of individual environmental variables on determination of baselines and CWSI. The theoretical approach evaluated the upper and lower baselines for canopy to air temperature differentials by accounting for some of the environmental and plant parameters. Jackson et al. (1981) proposed the following relationship to calculate canopy to air temperature differential:

\[
T_c - T_a = \frac{\frac{r_c Q_v}{\rho v C_v} \frac{\gamma (1 + r_c / r_v)}{\Delta + \gamma (1 + r_c / r_v)}}{\Delta + \gamma (1 + r_c / r_v)} (e^*_c - e^*_a) \]

(3.2)

From Eq 3.2, it is seen that the temperature difference between canopy and the air is a function of solar radiation, air vapor pressure deficit, and canopy and air resistances. Manipulation of Eq 3.2 allows the calculation of upper and lower limits that a plant experiences under potentially transpiring and stressed conditions. The actual temperature of the canopy always falls between the temperatures of a potentially evaporating and of a completely dry surface. The upper limit of canopy-air temperature difference is evaluated when \( r_c \) is allowed to increase without bound, meaning that \( r_c \rightarrow \infty \), for a non-transpiring plant, and Eq 3.2 becomes:

\[
T_{ul} - T_a = \frac{r_c Q_v}{\rho v C_v} 
\]

(3.3)
and, the lower limit is obtained when \( r_c \to 0 \) in Eq 3.2, thus the relationship takes the form:

\[
T_{ul} - T_x = \frac{r_c Q_k}{\rho_c C_p} \frac{\gamma}{\gamma + \Delta} - \frac{1}{\gamma + \Delta} (e^*_a - e_x)
\]  

(3.4)

which is the case for a plant transpiring at a potential rate. However, canopy resistance will be larger than zero even when the plants are well-watered because most plants shows some resistance to water flow even though the water is not limiting. Jackson et al. (1988) recommended that \( \gamma \) in Eq 3.4 must be replaced with \( \gamma^* = \gamma (1 + r_m / r_a) \), where \( r_m \) is the minimal canopy resistance of the canopy under potentially transpiring conditions. Then, the proposed CWSI takes the form as:

\[
\text{CWSI} = \frac{T_c - T_{ul}}{T_{ul} - T_{ll}}
\]

(3.5)

where \( T_{ll} \) and \( T_{ul} \) are the theoretically lower and upper limits of canopy temperature, respectively.
Jackson *et al.* (1988) reexamined the theoretical approach and proposed a method for estimating an aerodynamic resistance applicable to a plant canopy. It was indicated that solar radiation and wind speed at the time of the measurement might affect the canopy-air temperature difference. Therefore, they recommended that the upper and the lower limits must be calculated for the same environmental conditions that existed at the time of the measurements. This is a major difference between the empirical and the theoretical method. The upper and lower limits are assumed to be constant in the empirical method.

Theoretical approach and its modifications has also been used to determine plant water stress and was related to plant water potential, soil water depletion, and crop yield. Jackson (1982) used his original CWSI to determine plant water stress and its relation to the fraction of extractable soil water for wheat crop. The study reported that CWSI increased with time, roughly in parallel with the extractable soil water used. However, it was pointed out that CWSI values did not drop to its lowest value immediately after irrigation. This indicated that stressed wheat required some time to recover. It was explained that leaves needed to rehydrate and the roots that were previously in dry soil needed to develop new root hairs.

O'Toole and Hatfield (1983) evaluated the effect of wind speed on determination of upper limits for CWSI. They found that CWSI values measured at low wind speed overestimated the level of water stress, while CWSI measured at high wind speeds underestimated it. Therefore, they recommended that it is necessary to account for environmental factors and their effects on determination of upper and lower baselines and also CWSI.
Saha et al. (1986) conducted field studies on differentially irrigated chickpea to investigate the potential use of remotely sensed canopy temperature using a theoretical CWSI to assess crop water stress. They also evaluated the relationship between canopy temperatures and crop evapotranspiration. The results showed that canopy air temperature differential was inversely related to vapor pressure deficit of air as well as to evapotranspiration in both irrigated and unirrigated plots.

Clawson et al. (1989) used a modified version of the theoretically driven CWSI to evaluate plant water stress under different irrigation regimes. They used a reference well-watered plot to determine the lower limit of canopy temperature rather than calculating it, and included this value in the calculation of CWSI. The results showed that modified CWSI responded to different irrigation regimes and to plant water stress better than the original method. They also pointed out that using a measured lower limit canopy temperature would obviate the need for estimations of minimum canopy resistances.

Stanghellini et al. (1992) proposed a model to calculate canopy internal resistance \( r_i^* \) to replace \( r_c \) in Eq 3.2. The motivation behind their work was to account for both the finite minimal value of internal resistance and its reaction to weather as:

\[
r_i^* = r_{\text{min}} \prod_k \left( \frac{T_i}{P_k} \right)
\]  

(3.6)
In their model, the behavior of the internal resistance was related only to shortwave radiation. They stated that a rectangular hyperbola represented the relation between $r_i$ and shortwave radiation as:

$$ r_i^* = r_{ma} \frac{I_i + C_1}{I_i + C_2} \quad (3.7) $$

The study concluded that detection of water stress by this method provided similar results when compared to those obtained by Jackson’s (1988) crop water stress index.

De Lorenzi et al. (1993) also proposed a model for calculation of minimal canopy resistance to evaluate lower limit of canopy temperature. Based upon an empirical model for stomatal conductance:

$$ g_t = g_e + g_{ma} (1 - e^{-a_t}) \quad (3.8) $$

Hence, the minimal canopy resistance, $r_m$, was determined as:

$$ r_m = \frac{1}{LAIg_t} \quad (3.9) $$

This study evaluated three methods to detect water stress on field grown pepper plants: theoretical methods proposed by Jackson et al. (1981), Clawson et al. (1989), and their proposed index which used a model to predict minimal canopy resistance for well-watered plants as explained above. The study concluded that the theoretical model introduced by Clawson et al. (1989) was the quickest method to detect water stress, some
70 mm potential evapotranspiration having accumulated that is fifteen days after irrigation. In addition, CWSI values calculated both with Jackson et al. (1981) and De Lorenzi et al. (1993) displayed higher values (close to CWSI values of their stressed counterparts) even for well-watered plants. This was explained as the plant’s inability to transport water from the root zone.

Stanghellini and De Lorenzi (1994) compared soil and canopy temperature based methods to detect plant water stress for rye-grass. The study implemented theoretically derived CWSI with a constant canopy resistance \( r_m = 65 \text{ s m}^{-1} \) as canopy-temperature-based method and time domain reflectometry measurements of soil water content as soil-based detection method. Both methods were able to detect water deficit by the time transpiration was reduced to 80% of its potential value. Soil-based index relied on the estimation of root water extraction rate which might be difficult to determine. It was concluded that canopy-temperature-based method was to be preferred to soil-based stress detection method. Stanghellini (1994) stated that in a poorly ventilated ambient, stomatal closure might be lesser a defense against water loss than in the more natural environment. It was also suggested that plant-temperature-based means of stress detection could be more timely in greenhouses than in open field.

Al-Faraj et al. (1999) developed a fuzzy logic crop water stress index for tall fescue turf irrigation decision making. The application of Fuzzy logic eliminated the need to calculate upper and lower temperature baselines which were the necessary calculations in the empirically derived CWSI approach. The study showed promising adjustment and contribution in evaluating CWSI, however the baseline CWSI values to detect water stress were not established.
3.6. Infrared thermometry measurement considerations

Accurate temperature measurement using infrared thermometers requires considerations of field of view and sensor body temperature. Bugbee et al. (1998) discussed the effect of these parameters on error of leaf temperature measurement. They recommended the use of an infrared thermometer with a wider field of view and placement close to the target. It was pointed out that there were several disadvantages to narrow field of view infrared thermometers such as seeing less of the target and more of their body temperature so they have increased sensitivity to sensor body temperature. The output of the infrared sensor is determined by the difference between the temperature of the target and sensor's body temperature. An error in the measurement of the detector body temperature may result in an error in the temperature measurement. Bugbee et al. (1998) minimized this error by adding thermal mass around the detector to prevent rapid temperature changes and to keep all parts of the sensor at the same temperature. This also allows a ±0.2°C accuracy in leaf temperature measurements. The infrared thermometer they developed is now available on the market.

O'Toole and Real (1984) discussed the calculation of target dimension provided that the field of view of a specific infrared sensor is known. It was indicated that the knowledge of target dimensions and special characteristics related to the ellipse center and target point at various distances to and angles above the target surface should decrease error in surface temperature measurements and assist in standardization of canopy temperature measurement techniques with infrared sensors.
Nielsen et al. (1984) investigated the effect of solar azimuth and infrared thermometer view direction on measured soybean canopy temperature. The study showed that the canopy temperature of soybeans observed from a given azimutal position varied from the average as determined from four measurements made from the four different directions. They recommended that an average of temperatures viewed from several directions was the best approximation of true canopy temperature. The temperature averaging process, which occurs in the field of view of the infrared thermometers, may limit the ability of the instrument to detect moderate plant water stress level above which crop productivity is decreased. The sensitivity could be improved when sighting of the infrared thermometer is parallel to the sun rays (Fuchs, 1990).

The presence of reproductive structures such as fruits and flowers, may present a sampling problem on temperature measurements (Hatfield, 1990). It was pointed out that these structures, typically non-transpiring surfaces, could be as much as 3 to 5 °C warmer than transpiring leaves if they are exposed to full sunlight. Thus, modification of the sampling from an oblique to a more vertical angle was attested.

Based on the recommendations obtained from above literature, in the present study, attention will be paid first to use an infrared thermocouple which has a body temperature compensation to reduce measurement errors. Secondly, field of view of the sensor will be determined experimentally to ensure that the sensor measures only plant canopy temperature.
3.7. Machine vision applications for plant biotic movement and detection of plant water stress

Vision allows humans to perceive and comprehend the world around them. Machine vision aims to duplicate the human vision by electronically perceiving and understanding an image. It is a non-invasive, non-contact sensing technology which enables multi-dimensional sensing capabilities. Machine vision can be used to extract various information from a targeted object. These can be morphological (size, shape, texture etc), spectral (color, temperature, moisture), and temporal (growth rate, development, dynamic change of spectral and morphological states). In this section, only those literature using plant morphology changes to determine plant water status and water stress will be presented.

Under adequate water supply, plant leaves are often perpendicular to the incident radiation while under water stress conditions leaves may be oriented parallel to the direct solar radiation. Oosterhuis et al. (1985) studied soybean leaflet movements as an indicator of crop water stress. This study showed a good agreement between leaflet angle and leaf water potentials as well as relationship between leaflet angle and plant available water in the soil. The data revealed that as the plant started experiencing water stress, leaflets became noticeably inverted at a critical leaf water potential of about -1.4 Mpa. The data reported that inversion of soybean leaflets started when 60% of the plant available water was depleted.
Tracking of leaf tips for tomato plant wilt detection using machine vision was evaluated by Seginer et al. (1992). This study evaluated the vertical movement of leaf tips. The data showed that fully expanded leaves of tomato plants had linear vertical motions in response to both water stress level and carbon dioxide assimilation rate. It was pointed out that growing leaves had complex biotic motions which were less useful for monitoring water stress level. This study concluded that machine vision was able to detect the onset of wilt before visual stress detection and triggered irrigation at predetermined leaf tip deflections.

Using single leaf tips to detect water stress requires monitoring of many leaves to represent the whole canopy. Kurata and Yan (1996) used an algorithm to extract lines reflecting inclinations of rachises of tomato plants from the whole canopy image obtained from a fixed position slightly above the canopy. The study showed that average incline of the extracted lines correlated well with plant water potential of tomato plants. The data revealed that water potential of well watered plants did not change much while the average incline of the treatment plants decreased as the water potential decreased. This study did not provide a quantitative baseline as an indicator of plant water stress.

Nyakwende et al. (1996) investigated the use of image boundary moments as a parameter for plant wilt detection. The moments of the edges of an image about its center of gravity were defined as edge moments. The outer boundary of the image was considered to be a special case of an edge. Thus, only outer boundary pixels were considered to compute boundary moments ignoring other line segments in the image. Water stress was induced by withdrawing irrigation from the treatment plants. As the visual wilting occurred, plants were irrigated and the images of plants were evaluated to
determine the image boundary moments as the plant recovered from wilting condition. The data showed that a significant (40-45%) change in boundary moments was seen for the water stress plants as they recovered from stress condition. The authors suggested that leaf motions of fully expanded leaves under water stress gave more accurate measure of wilt since it was those leaves which primarily determine boundary moments. This study did not evaluate the image boundary moments under series of water stress cycles, image boundary moment was evaluated only after the plants were irrigated and recovered from stress condition. No quantitative baseline using image boundary moments for early water stress detection was reported.

Murase et al. (1997) incorporated the change of plant movement into a neural network model to determine the water stress level of tomato plants. The images of the plant were used as inputs on a finite element image processing grid. Movement of the leaves and stem of the wilting plant due to water loss of turgidity of tissues was recorded on a videotape through a CCD color camera. The data revealed that a strong non-linearity existed in the relationship between water content of the leaf and the value of the finite element plant feature.

Revollon et al. (1998) evaluated the possibility of using the change of angle between the horizontal and the line from the axil to the tip of the leaf (ATL) to detect plant water stress of an ornamental shrub (*Forsythia x intermedia*). After stopping irrigation to the plant in the treatment group, ATL of a single leaf was able to show deviations in three days when compared to the values obtained from the plant in control group. The authors suggested that this technique should be tested and validated further using whole plant canopy.
Studies conducted on machine vision applications for non-destructive and non-contact assessment of plant water status and water stress has advantages over other methods. The literature show that there is a good correlation between leaf deflection and water status of the plant. However, it is necessary to establish baselines for early water stress detection with the markers being used. Most of the literature using machine vision applications to detect plant water stress focused on tracking of leaf tip and the change of leaf inclination from a single plant leaf. Another possibility of machine vision based plant water stress detection may be to analyze the image of the whole canopy, rather than that of the individual plant part, to extract information related to the average water status of the plant canopy. With this approach, required hardware will also be simpler, because the camera will be fixed at a position above the plant canopy to obtain a wide view of the canopy and no movable parts will be needed.
CHAPTER 4

MATERIALS AND METHODS

4.1. Physical basis of transpiration and evaporation

Modeling water transport through the soil-plant-atmosphere continuum is a function of potential energy differences. As the potential energy of position increases when a mass is elevated some distance above a datum, a potential for water transport represents a measure of the energy which is available to move water. The forces involved in the movement of water originate from different phenomena such as electrical, mechanical, chemical, etc. Water is involved in processes in green plants and much of the activity is due to either to a phase change or results from the presence or addition of solutes. Therefore, an ideal choice is to approach the potential causing water movement in the soil-plant-atmosphere continuum from a chemical viewpoint (Merva, 1995).

Water is absorbed by the roots and translocated through the plant in the direction of decreasing chemical potential gradient which is driven from a high chemical potential in the soil to a low potential in the atmosphere. Here, the chemical potential is derived from the concept of Gibbs free energy, the energy which is available to move water.
The free energy of a substance can be related to its temperature and pressure. For instance, assuming that the temperature is constant and using the perfect gas law, the free energy of a perfect gas would change with pressure from state 1 to state 2 according to the following relationship:

\[ f_2 - f_1 = nRT \ln \left( \frac{P_2}{P_1} \right) \]  

(4.1)

And, writing this relationship for ideal solutions:

\[ f_2 - f_1 = nRT \ln \left( \frac{c_2}{c_1} \right) \]  

(4.2)

where,

- \( f_{1,2} \) = free energy of substance in state 1 and 2 (J)
- \( n \) = number of moles of substance
- \( R \) = standard gas constant (8.314 J mol\(^{-1}\) K\(^{-1}\))
- \( T \) = temperature (K)
- \( P_{1,2} \) = pressure of substance in state 1 and 2 (Pa)
- \( c_{1,2} \) = concentration of substance in state 1 and 2 (mol kg\(^{-1}\))
Chemical potential is defined as free energy per mole of a substance. From the perfect gas law, the partial molal volume of water can be expressed in terms of vapor pressure:

$$V_w = \frac{n_wRT}{e} \tag{4.3}$$

From which:

$$\frac{dV_w}{dn_w} = \frac{RT}{e} = \overline{V}_w \tag{4.4}$$

And integrating both side of the equation:

$$\int_{\mu_w}^{\mu} d\mu_w = RT \int_{e^o}^{e} \frac{de}{e} \tag{4.5}$$

Equation 4.5 integrates to give the chemical energy of a solution:

$$\mu_w - \mu^o_w = RT \ln\left(\frac{e}{e^o}\right) \tag{4.6}$$

where,

- $V_w$ = volume of water ($m^3$ mol$^{-1}$)
- $\overline{V}_w$ = partial molal volume of the water ($m^3$ mol$^{-1}$)
- $n_w$ = number of moles of water
- $e$ = vapor pressure of water in the system (Pa)
\( \varepsilon^o \) = saturation vapor pressure of water at the same temperature (Pa)

\( \mu \) = chemical potential of water in the system (J mol\(^{-1}\))

\( \mu^o \) = chemical potential of pure water at atmospheric pressure at the same temperature as the system which is under consideration (J mol\(^{-1}\))

The chemical potential has units of energy content (J mol\(^{-1}\)). However, the common practice in plant physiology is to express water status as water potential using pressure units. This can be done by dividing chemical potential of water by the partial molal volume of water as:

\[
\psi = \frac{\mu - \mu^o}{\bar{V}_w}
\]

(4.7)

where,

\( \psi \) = water potential (Pa)

\( \bar{V}_w \) = partial molal volume of the water (18.05 x 10\(^{-6}\) m\(^3\) mol\(^{-1}\) at 20°C)

Water moves from the soil (\( \psi \approx 0 \)) to the leaf (concentration higher and \( \psi \) is negative) and from leaf to the air (\( \psi \) is more negative). The rapid thermal motions of the individual molecules in a fluid lead to random rearrangement of molecular position and, in inhomogeneous fluid, to transfer of mass and heat. This process is called diffusion.
Water evaporates from leaf according to the basic law of mass transfer. And, one-dimensional form of Fick's Law of Diffusion describes diffusion for an entity by following the relationship (Monteith, 1973):

\[
J_i = -D_i \frac{\partial C_i}{\partial x}
\]

(4.8)

where,

\( J_i \) = the flux density or rate of mass transfer (mol m\(^{-2}\) s\(^{-1}\))

\( D_i \) = diffusion coefficient (m\(^2\) s\(^{-1}\))

\( \frac{\partial C}{\partial x} \) = concentration gradient in the x direction (mol m\(^{-3}\) m\(^{-1}\))

Defining resistance as the inverse of conductance (here diffusion coefficient divided by distance) and integrating equation 4.8 between locations 1 and 2 gives:

\[
J_i = \frac{C_{i1} - C_{i2}}{r_i}
\]

(4.9)

where,

\( C_{i1-2} \) = concentration of entity i at locations 1 and 2 (mol m\(^{-3}\))

\( r_i \) = resistance to diffusion of entity i in a medium (s m\(^{-1}\))
Evaporation of water from leaf to air occurs due to concentration gradient, basically due to differences in humidity between the location of water evaporation in the leaf and the air surrounding the leaf. Hence, Fick's Law can be applied here to calculate the evaporation in mass per unit area per unit of time as:

\[ E = \frac{d_{vt} - d_{va}}{r_v} \]  

where,

- \( E \) = evaporation (kg m\(^{-2}\) s\(^{-1}\))
- \( d_{vt} \) = absolute humidity at leaf liquid-vapor phase transition (kg\(_{\text{vapor}}\) m\(^{-3}\) air)
- \( d_{va} \) = absolute humidity of air surrounding the leaf (kg\(_{\text{vapor}}\) m\(^{-3}\) air)
- \( r_v \) = resistance to vapor diffusion (s m\(^{-1}\))

Expanding equation 4.10 further enables the calculation of evapotranspiration for plant canopies.
4.2. Plant canopy energy balance and evapotranspiration

The first law of thermodynamics states that energy can not be created or destroyed, however it only changes from one form to another. This law can be applied to the energy balance of plant canopy with no exception and can be expressed as (Figure 4.1a):

\[ E_S = E_I - E_O \]  \hspace{1cm} (4.11)

where,

- \( E_S \) = Stored energy in leaf resulting from photosynthesis and metabolic activity (W m\(^{-2}\))
- \( E_I \) = Radiation from both long wave and short wave (W m\(^{-2}\))
- \( E_O \) = Heat loss by convection, conduction, latent heat (W m\(^{-2}\))

The first law of thermodynamics can be re-written to define the energy balance for a unit area of a plant canopy (Figure 4.1b):

\[ Q_{RAD} - Q_G - Q_E - Q_H = 0 \]  \hspace{1cm} (4.12)

where,

- \( Q_{RAD} \) = irradiance absorption rate by plant canopy (W m\(^{-2}\))
- \( Q_G \) = the energy stored or released by the biochemical reactions (W m\(^{-2}\))
- \( Q_E \) = evapotranspiration latent heat flux (W m\(^{-2}\))
- \( Q_H \) = convective heat flux between plant leaves and the air (W m\(^{-2}\))
Figure 4.1: The application of first law of thermodynamics: a) to plant canopy b) to leaf
The heat lost through latent heat flux must balance the portion of the heat gain from radiation which is not removed by convective sensible heat flux or stored in the canopy. The latent heat flux is the energy used in the evaporation process, and can be calculated from Equation 4.10:

$$Q_e = \lambda E = \lambda \frac{d_x - d_u}{r_v}$$  \hspace{1cm} (4.13)

where,

$\lambda$ = latent heat of vaporization (\(J\ kg^{-1}\))

Equation 4.13 can be rearranged to determine latent heat flux in terms of vapor pressure with the assumption that the leaf temperature defines the temperature at the interface between liquid and vapor in the leaf. Hence, the latent heat flux is determined as:

$$Q_e = \lambda \frac{\rho \varepsilon [e^*(T_c) - e(T_a)]}{P}$$  \hspace{1cm} (4.14)

where,

$\rho$ = density of air (\(kg\ m^{-3}\))

$P$ = atmospheric pressure (Pa)

$\varepsilon$ = molar conversion from water to dry air [(kg mol\(^{-1}\))\(_w\) (kg mol\(^{-1}\))\(_a\)^{-1}]

$e^*(T_c)$ = saturation vapor pressure at leaf temperature (Pa)

$e(T_a)$ = vapor pressure at air temperature (Pa)
Using psychrometer constant one can combine some of the physical constants in Equation 4.14. The resistance to vapor diffusion in evapotranspiration can be separated into two different functional terms: one mass transfer term which describes the resistance offered by the air and one physiological term which accounts for the variable resistance offered by the leaf due to stomatal opening. The sensible heat flux from a surface can be derived from Fick's Law (Equation 4.8) in a similar manner to the latent heat flux, with the temperature as the driving force. Thus, latent and sensible heat fluxes can be written for a fully developed boundary layer as:

\[
Q_e = \frac{2LAI \rho C_p \left[ e^*(T_c) - e(T_a) \right]}{\gamma (r_{av} + r_s)} \tag{4.15}
\]

\[
Q_{ht} = \frac{2LAI \rho C_p (T_c - T_a)}{r_{ah}} \tag{4.16}
\]

where,

- \(C_p\) = specific heat of the air at constant pressure (J kg\(^{-1}\) °C\(^{-1}\))
- \(\gamma\) = thermodynamic psychrometric constant (Pa °C\(^{-1}\))
- \(r_{av}\) = resistance to water vapor transfer due to air around leaf (s m\(^{-1}\))
- \(r_s\) = resistance to water vapor transfer due to the leaf or stomates (s m\(^{-1}\))
- \(r_{ah}\) = air resistance for heat diffusion (s m\(^{-1}\))
- \(LAI\) = leaf area index (dimensionless)
Based on Penman's transformation, the saturation vapor pressure at leaf temperature can be approximated using a basic relation between saturation vapor pressure deficit and a corresponding temperature difference (Figure 4.2) (Oke, 1992).

Hence, the saturation vapor pressure at the leaf temperature equals:

$$e^*(T_c) = e^*(T_a) + \delta(T_c - T_a)$$  \hspace{1cm} (4.17)

where,

- $e^*(T_a) =$ saturation vapor pressure at air temperature (Pa)
- $\delta =$ slope of the saturation vapor pressure vs. temperature curve (Pa °C$^{-1}$) $\left(\delta = \frac{de^*}{dT}\right)$. 

Figure 4.2: Saturation vapor pressure vs. temperature diagram showing Penman's transformation.
Substituting Equation 4.17 into 4.15 yields to:

\[
Q_E = \frac{2LAIpC_p \left[ e^*(T_a) - e(T_a) + \delta(T_e - T_a) \right]}{\gamma(r_{av} - r_s)} \tag{4.18}
\]

Introducing vapor pressure deficit of air as \(VPD_a = e^*(T_a) - e(T_a)\), Equation 4.18 can be rearranged as:

\[
Q_E = \frac{2LAIpC_p \left[ VPD_a + \delta(T_e - T_a) \right]}{\gamma(r_{av} - r_s)} \tag{4.19}
\]

The resistance to water vapor transfer, \(r_{av}\), can be related to air resistance for heat diffusion, \(r_{ah}\) (Yang, 1988):

\[
\frac{r_{av}}{r_{ah}} = \left( \frac{\alpha_h}{\alpha_w} \right)^{1.333} = 0.81 \times r_{ah} \tag{4.20}
\]

where \(\alpha_h\) and \(\alpha_w\) are heat and water vapor diffusion coefficients, respectively. Using the relationship in Equation 4.20, and plugging Equation 4.16 and 4.19 into Equation 4.12 to solve for \(Q_E\) (assuming that energy storage in the plant due to biochemical reaction \(Q_G\) is negligible) yields:

\[
Q_E = \frac{2LAIpC_p VPD_a + \delta Q_{RAD}}{\delta + \gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)} \tag{4.21}
\]
Then, dividing latent heat flux, Equation 4.21, by latent heat of vaporization, $\lambda$, provides final equation to calculate evapotranspiration rate from a plant as:

$$
ET = \frac{2LAI\rho CP_{VPD} + \delta Q_{RAD}}{\delta + \gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)} \times \frac{1}{\lambda}
$$

(4.22)

where,

$ET = $ evapotranspiration rate (kg m$^{-2}$ s$^{-1}$)

4.3. Defining plant water stress

Transpiration furnishes the driving force for water movement through the plant and is the evaporative cooling mechanism of the plants. Because large quantities of energy are required in the change of phase from liquid to vapor, evaporation from the leaf surface provides a very efficient mechanism for heat dissipation. Plants must withdraw enough water to meet demand for transpiration.

The principle concept of plant-temperature-based stress detection is that transpiration from plants not adequately supplied with water will be less than well-watered plants, and therefore the temperature of stressed plants will be warmer than their well-watered counterparts.
A well-watered healthy plant transpires at a potential rate. When water supply becomes a limiting factor, the actual evapotranspiration rate will be lower than the potential evapotranspiration rate. The onset of incipient plant water stress occurs when the evaporative demand (transpiration) exceeds supply (the available water to the plants) for that demand. For a plant system, the potential evapotranspiration rate can be considered as the evaporative demand because it is the maximal rate of water loss from the plant under non-water limiting condition. Therefore, deviation of actual evapotranspiration rate from its potential rate can be used to determine the water stress level in the plant. Thus, the ratio of actual evapotranspiration to potential evapotranspiration can be used as a stress index, namely crop water stress index (CWSI), to define the water status of the plant as

\[
CWSI = 1 - \frac{ET}{ET_p} \tag{4.23}
\]

where,

- \(CWSI\) = crop water stress index (dimensionless)
- \(ET\) = actual evapotranspiration rate (kg m\(^{-2}\) s\(^{-1}\))
- \(ET_p\) = potential evapotranspiration rate (kg m\(^{-2}\) s\(^{-1}\))
Equation 4.22 can be used to calculate the potential evapotranspiration rate by replacing psychrometric constant $\gamma$ with:

$$
\gamma^* = \gamma \left[ 0.81 + \left( \frac{r_s}{r_{ah}} \right)_p \right]
$$

(4.24)

where, the ratio of $r_s$ to $r_{ah}$ is calculated with the measurements obtained from well-watered plants. Then, using the relationship in Equation 4.24 and plugging Equation 4.22 into Equation 4.23 to calculate CWSI results in:

$$
\text{CWSI} = \frac{\gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right) - \gamma^*}{\delta + \gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)}
$$

(4.25)

The temperature of the plant can be predicted theoretically by using the energy balance of the plant in a similar manner which was used to calculate the evapotranspiration rate of the plant canopy. Substituting Equation 4.19 into 4.12 and solving for $T_c - T_a$ enables the prediction of leaf-air temperature differential as:

$$
T_c - T_a = \frac{Q_{RAD} r_{ah}}{2\text{LAI}\rho C_p} \frac{\gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)}{\delta + \gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)} - \frac{\text{VPD}_a}{\delta + \gamma \left( 0.81 + \frac{r_s}{r_{ah}} \right)}
$$

(4.26)
The ratio of $r_s$ to $r_{ah}$ is required in the calculation of CWSI. The ratio can be obtained by solving Equation 4.26 for $r_s r_{ah}^{-1}$ and this yields:

$$
\frac{r_s}{r_{ah}} = \frac{0.81 Q_{RAD} r_{ah} \gamma}{2 LAI \rho C_p} \left[ (T_c - T_a)(0.81 \gamma + \delta) \right] - VPD_a
$$

(4.27)
4.4. Growth chamber description

This study was carried out in a walk-in sized growth chamber installed in a laboratory of the Department of Food, Agricultural, and Biological Engineering at The Ohio State University, Wooster, Ohio (Figure 4.3). The growth chamber internal dimensions were 2.4 x 3.6 x 2.4 m in width, length, and height, respectively. A 32 mm double acrylic glazing material served as a barrier between the lamps and the growing area to reduce the thermal load from the lamps onto the plants. The walls were 1 mm thick sheet steel, painted semi-gloss latex white, enclosing 0.15 m thick rigid polystyrene foam insulation. The floor was white vinyl sheet on a wooden plywood frame.

The lighting system included eight 400 W high pressure sodium (HPS) and seven 400 W metal halide (MH) lamps mounted above the acrylic barrier on a 0.38 x 0.45 m spacing (Figure 4.3). A 370 W, 0.61 m diameter axial fan drew air in through two filtered air inlets above the acrylic panels, and across the lamps to remove the thermal load caused by the lamps.

The recirculating air inlet was a slot extending the entire width of one end wall at a height 0.2 m below the acrylic barrier. The air outlet was a 0.5 x 0.5 m in size and was located 0.1 m from both the floor and the side wall of the chamber on the bottom corner of the opposite wall from the inlet. The air, leaving the growth chamber, passed a cool coil and a hot coil heat exchanger before reentering the chamber in a closed system. Water was the agent used in the cooling system. The water was heated and chilled respectively by a 10.5 kW chilled water refrigeration unit (Model PC-300, Heat-X, Inc.,
Brewster, NY). The air inlet in the chamber had an adjustable slot to control the amount of air coming into the chamber and onto the plant canopy.

Four experiments were conducted in this study. The experiments were repeated three times under low humidity conditions. The fourth experiment was run under higher humidity conditions to further evaluate the effectiveness of plant water stress detection techniques. The test plants were subjected to several stress-recover cycles of water stress during the experiments. The only environmental control in the growth chamber during the first three experiments was temperature control which was achieved using a Honeywell temperature controller (UDC 3300, Honeywell, Fort Washington, PA). The humidity level in the chamber during the fourth experiment was increased using a steam generating type of humidifier (AutoFlo, Model WSU-14, EWC Controls Inc., Englishtown, NJ). Measured environmental conditions and cultural info during the experiments are shown in Table 4.1.

<table>
<thead>
<tr>
<th>Environmental conditions</th>
<th>Experiment</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>First</td>
</tr>
<tr>
<td>Light level (W m⁻²)</td>
<td>141.1±5.00</td>
</tr>
<tr>
<td>Relative humidity (%)</td>
<td>21.0±1.30</td>
</tr>
<tr>
<td>Air temperature (°C)</td>
<td>22.1±0.45</td>
</tr>
<tr>
<td>Air velocity (m s⁻¹)</td>
<td>0.25±0.01</td>
</tr>
<tr>
<td>Carbon Dioxide (ppm)</td>
<td>450±10</td>
</tr>
<tr>
<td>Nutrient solution (ppm)</td>
<td>200 N-P-K</td>
</tr>
<tr>
<td>Growth medium composition</td>
<td>29% sphagnum peat moss, 20% coarse perlite, 20% grade 2 vermiculate, and 31% composted pine bark</td>
</tr>
</tbody>
</table>

Table 4.1: Measured environmental conditions during the experiments.
Figure 4.3: Growth chamber facility at the Department of Food, Agricultural, and Biological Engineering, Ohio Agricultural Research and Development Center, Wooster, Ohio. Side view of the chamber is presented.
4.5. Light and air velocity characterization of the chamber

The environment is usually not constant with time in growth chambers. Radiative spectral quality and quantity can be affected by factors such as degradation of lamp output, barrier discoloration, and chamber surface temperature etc. That is why it was necessary to have an idea of the radiative characteristics of the growth chamber used in this study.

The radiative environment in this study was characterized using a Li-Cor pyranometer (PY 8017, LI-COR Inc., Lincoln, NE). Measurements were taken over a 2.1 x 3.3 m surface area in the chamber using a 0.3 x 0.3 mesh spacing at the beginning of the study. Figures 4.4 shows the measurement locations and the distribution of the total irradiation iso-line plot, respectively.

Total irradiation iso-line plot shows a peak in the center of the chamber area, and a gradual but axial symmetrical decrease in irradiance toward the edges of the chamber. The graph shows that spatial light distribution is more uniform at the center of the chamber.

The air velocity profile in the growth chamber was characterized using computational fluid dynamics (CFD) simulation software FLUENT 4.5 (Figure 4.5). This growth chamber provided air movement horizontally over the plant canopy. The continuous slot air inlet opening was tapered (from 25 mm to 2 mm) to provide maximal uniformity of air movement across the width of chamber. The air entering from the recirculating slot inlet flows across the ceiling of the chamber under the acrylic barrier and moves downward at the far-end wall of the chamber and leaves the chamber through the air outlet. The air velocities were not uniform in the chamber.
Figure 4.4: Light characterization measurement mesh (a) and spatial distribution of total irradiation (W m\(^{-2}\)) at height of 1.0 m (b).
Figure 4.5: CFD simulated air velocity profile in the growth chamber.
4.6. Plant Material

In this study, varieties of New Guinea Impatiens were used. Antares and Riviera Pink varieties were used in the first and second experiments respectively while Paradise variety was used in the third and the fourth experiments. All the varieties had green foliage and pink flowering plant characteristics. Therefore, they were assumed to have similar physiological characteristics (McMahon, 1999).

New Guinea Impatiens are native to the Australian New Guinea subtropical highlands. Impatiens spp. are annual or perennial succulent herbaceous plants with green stems (Mankin, 1994). New Guinea Impatiens are known to be "low light" and "low transpiring" plants.

The plants were obtained from a local grower at their vegetative stage in 101mm (4 inches) pots. Before the experiments started, the plants were acclimatized in the growth chamber for a week with a lighting scheme of 10 hours of lighting and 14 hours of darkness. The plants were fertilized once prior the first day of the experiment with 200 ppm N-P-K fertilizer mixture while all of the plants were applied with nutrient solution twice during the fourth experiment, one at the beginning of the experiment and one after the second stress cycle.

Six New Guinea Impatiens plants, all uniform in appearance and size, were used in each experiment. Three of these were stressed plants and the other three were control plants. All six plants were kept well watered for two days above 45% volumetric soil moisture content at the beginning of each experiment. While the control plants were maintained ≥ 45% moisture level for the rest of the experiment, the water was withheld
from the stress group until water stress was observed by the operator. When the stress
was observed visually, water was added to the media of the water stressed plants to bring
soil moisture back to $\geq 45\%$. Both a computer controlled automated drip irrigation system
and manual irrigation were used to water the plants.
4.7. Potting medium analysis and soil water release curve

A growth medium is a combination of materials used to furnish support, water retention, aeration, or nutrient retention for plant growth. The growing medium used in this study was a soilless potting mix consisted of peat moss, perlite, vermiculate, and compost bark. The percentages of the potting mixture components by volume were 29% sphagnum peat moss, 20% coarse perlite, 20% grade 2 vermiculite, 31% composted pine bark.

Irrigation management involves controlling the moisture content of the root zone. Once water is placed in the growing medium, the plant needs to be able to extract it. The force that the plants have to overcome to extract water plays a major role in if the plants are able to take up enough adequate amounts. This force is measured as moisture tension (matric tension) and is nonlinearly related to moisture content (Rendig and Taylor, 1989). It is possible to generate a curve for this relationship, called the soil moisture release curve. A soil moisture release curve characterizes the media's ability to retain water under various tensions. This curve shows different trends for different growing media, except that the curve always decreases with increasing tension.

A precise understanding of physical properties and water supply conditions of the growing medium is necessary to make proper evaluations and conclusions on the detection of plant water stress. Thus, the soil water release curve for the growing medium used in this study was obtained by a standard laboratory procedure.
Before the laboratory procedure took place, potting mix had to be prepared for the experiment. For the preparation, the potting mix was placed into cylindrical metal ring blocks and was compacted by adding water for one week. This procedure allows the soil to settle. Hence, dry bulk density of the medium increases as the total volume of the soil decreases. Figure 4.6. illustrates the cylindrical metal ring block. The sample then was placed into tempe cells for the experiment (Figure 4.7).

In the experiment, a tension table (Figure 4.8) and pressure plate (Figure 4.7) were used for <10 kPa (low tension) and <200 kPa tensions (high tension), respectively. A thermocouple psychrometer was used for measurements >200 kPa tensions (Figure 4.9). In the thermocouple psychrometer experiment, NaCl standard solutions were used to obtain psychrometric curves. Obtained standard curves were then used to produce the moisture content of the samples.
Figure 4.6: Cylindrical metal ring block for preparation of the potting mix for soil moisture release curve experiment.

Figure 4.7: Illustration of pressure plate experimental setup and tempe cells.
Figure 4.8: Illustration of tension table experimental setup.

Figure 4.9: Illustration of thermocouple psychrometer experimental setup.
Moisture content of the soil can be determined either by gravimetric or volumetric methods. Gravimetric soil moisture content is defined as:

\[ \theta_G = \frac{M_w - M_S}{M_s} \]  

(4.28)

where:

- \( \theta_G \) = gravimetric soil moisture content \((\text{g g}^{-1})\)
- \( M_w \) = the mass of water in the sample \((\text{g})\)
- \( M_S \) = the total mass of dry sample \((\text{g})\)

Assuming that the specific gravity of the water in the soil is 1.0, then volumetric soil moisture content of the soil was calculated by:

\[ \rho_s = \frac{M_s}{V_s} \]  

(4.29)

\[ \theta_v = \theta_G \times \rho_s \]  

(4.30)

where:

- \( \theta_v \) = volumetric soil moisture content \((\text{m}^3 \text{ m}^{-3})\)
- \( \rho_s \) = dry bulk density of the soil sample \((\text{g m}^{-3})\)
- \( V_s \) = total volume of the soil sample \((\text{m}^3)\)
In the thermocouple psychrometer experiment, thermocouple psychrometer assumes a relationship between vapor pressure of soil air and soil water potential, and it is in equilibrium with the soil water. This relationship is formulated as (Hanks, 1992):

\[ \psi_{swp} = \frac{R \times T}{V_m} \ln \left( \frac{e_{swp}}{e^*} \right) \]  

(4.31)

Where:

\( \psi_{swp} \) = soil water potential (kPa)

\( R \) = universal gas constant

\( T \) = temperature (°K)

\( V_m \) = molar volume of water (m\(^3\) mol\(^{-1}\))

\( e_{swp} \) = vapor pressure of soil air (kPa)

\( e^* \) = vapor pressure of saturated air at soil air temperature (kPa)

The complete experimental procedure for the soil moisture release curve laboratory experiment and experimental results are provided in Appendix A.
4.8. Turn-table design

In this study, a computer controlled turn-table was designed and installed in the growth chamber. Its operation was controlled by a computer system which rotated the table by powering a stepper motor, thus bringing plants individually to a camera and to other stationary sensors. The main purpose of rotating the plants was to provide more uniform environment to each plant, and to eliminate the randomization consideration for the experimental setup. A drawing of the turn-table with structural dimensions illustrates the setup in Figure 4.10.

The turn-table with it's instrumentation was designed and custom-built in the Department of Food, Agricultural, and Biological Engineering, The Ohio State University/OARDC, Wooster. The diameter and the thickness of plywood turn-table were 1.22 m and 2.54 mm. Six load cells were positioned 60° apart from each other at 0.45 m distance from the center of the table (Figure 4.10). The turn-table main board was mounted on a rotary positioning table (RT-12, Arrick Robotics Inc., Hurst, TX) which was powered by a stepper motor (MD2-a, Arrick Robotics Inc., Hurst, TX) through a pulley assembly (PR-23, Arrick Robotics Inc., Hurst, TX), (Figure 4.11). The stepper motor system provided precise positional control of the table. The accuracy of the rotary table used was 0.1 degree. An electronic control switch was installed underneath the turn-table main board in order to initialize the start of each revolution and home positioning of the table. The table was painted flat white color to establish maximum contrast between the target images and the background.
Figure 4.10: Structural dimensions of the turn-table and load cell with weighing platform, designated AA in the figure.
Figure 4.11: Parts of the turn-table motor drive system: a) MD2 Stepper motor b) RT-12 Rotary table, and c) PR-23 Pulley reducer assembly.
4.9. Instrumentation, data acquisition, and data storage

4.9.1. Measurement of variables for water supply

Each plant (total six) pot was mounted on a custom-built lysimeter consisting of a Tedea-Huntleigh load cell (Model 355, Tedea-Huntleigh, CA) and an 11.2 cm diameter weighing platform to monitor the weight change due to evapotranspiration (Figure 4.10). This load cell had a resolution of ±1 gram and a maximum weighing capacity of 5000 grams.

In order to obtain information about the water supply conditions, volumetric soil moisture of the growing medium in each pot was measured using a ThetaProbe soil moisture sensor (Model ML2, Dynamax, Houston, TX) (Figure 4.12a). The working range of the sensor was 0-1 m³m⁻³ with an accuracy of ±0.02 m³m⁻³. The response time of the probes was less than 0.5 seconds. The sensors were inserted into the pots from the side and were placed close to the root level of the plant (Figure 4.12b,c,d). This sensor used time domain reflectometry principle to measure the soil moisture.

*Time Domain Reflectometry*

In time domain reflectometry, soil moisture content is measured by responding to changes in the apparent dielectric constant. The changes are converted into a DC voltage, virtually proportional to soil moisture content.

The ThetaProbe consists of a waterproof housing which contains the electronics, and, attached to it at one end, four stainless steel rods that are inserted into the soil. The cable provides connection to a suitable power supply and an analog output signal. It is an
analog device, continually producing an output signal measuring soil properties in voltage difference terms.

The ThetaProbe measures soil parameters by applying a 100 MHz signal via a specially designed transmission line whose impedance is changed as the impedance of the soil changes. This impedance has two components; the apparent dielectric constant and the ionic conductivity. The signal frequency is chosen to minimize the effect of ionic conductivity, so that changes in transmission line impedance are dependent almost solely on the soil's apparent dielectric constant (ML2 User Manual, 1998).

This sensor requires one time calibration for the growing medium that is being used. Therefore, all the probes were calibrated by a laboratory procedure for the actual growing medium used in this study. The complete calibration procedure and the results of the calibration are provided in Appendix B.
Figure 4.12: The location of soil moisture sensor on the pot: a) soil moisture sensor b) sensor location in the medium c) design of the pot, and d) insertion of the sensor to the pot.
4.9.2. Measurement of variables for water demand

Two IRTC-P Precision Model infrared sensors (Apogee Instruments, Logan, UT) were mounted at a stationary location over the path of the plants to measure the plants' canopy temperatures. The readings from two sensors were then averaged to obtain a representative canopy temperature. The resolution of the infrared thermocouple sensor was ±0.02 °C.

In order to determine the field of view, one of the infrared sensors was positioned at a fixed height vertically and then a small LED point light source, painted to flat black, was moved slowly towards the field of view of the infrared sensor while the sensor continuously read the temperature of a flat white background. The LED light source was moved until a sudden increase in the temperature reading was observed. The sudden increase in the temperature change occurs due to the emissivity of the flat black painted LED point light source and the temperature of the light source. The location, at which the temperature was changed, was marked. This procedure was repeated several times approaching from different directions and the position of the temperature change was marked. Then, a circle was drawn passing through the marked positions. Knowing that the sensor had a conical sensing field, thus the field of view of the sensor was determined by using basic trigonometric relationships. The field of view of these sensors was found to be 3:1 ratio (0.01 m diameter sensed at a distance of 0.03 m). In order to accommodate this field of view and to ensure that the sensors did not shade the leaves, the sensors were located 0.05 m above the plants from the vertical axes of the center of
the plants. A schematic of this procedure and the field of view of the sensor are illustrated in Figure 4.13.
Figure 4.13: Schematic illustration of the procedure to determine field of view of the infrared thermocouple sensor.
The output of the infrared sensor is determined by the difference between the temperature of the target and sensors body temperature. An error in the measurement of the detector body temperature may result in an error in the temperature measurement. Bugbee et al. (1998) minimized this error by adding thermal mass around the detector to prevent rapid temperature changes and to keep all parts of the sensor at the same temperature. The sensor used in this study also had a thermal mass around the detector to prevent rapid temperature changes and its effect on errors in temperature measurements.

Other environmental variables made at the plant canopy level to determine water demand for the plants included air-dry bulb temperature measurement using one type-K thermocouple, ambient air velocity using a hot-wire anemometer (TSI 8455-12, TSI Inc., St. Paul, MN), light intensity using a LI-COR sensor (PY 8017, LI-COR Inc., Lincoln, NE), and a relative humidity sensor (H3V-200, Rotronic Instrument Corp., Huntington, NY). Air temperature measurement was made at a height of 0.3 cm at the center of the turn-table. The thermocouple was shielded to eliminate errors which could be caused by the heat load from light sources. Relative humidity, air velocity, light intensity measurements were made at a height of 1.5 m on a stationary platform which was about plant canopy level.

An electronic leaf area meter was used to determine total leaf area of the plants. In this study, leaf area index was defined as the ratio of the total leaf area to the projected horizontal area of the plant canopy. Total leaf area of three plants, similar in appearance and size as the experimental plants, was measured by removing the leaves to obtain an average total leaf area for the experimental plants at the beginning of the experiments. The total leaf area of the experimental plants were also measured at the end of each
experiment. The leaf area meter was calibrated with a known area of a sample object prior to the measurements. The electronic leaf area meter is illustrated in Figure 4.14.

Figure 4.14: Delta-T electronic leaf area meter.
4.9.3. Data acquisition and storage

In this study, data acquisition and storage were accomplished by using both data loggers and a desktop computer. Two Campbell Scientific data loggers (Model 21X and 23X) were used in the experiment. Model 21X with a multiplexer was mounted at the center of the turn-table. Model 23X was located outside of the chamber. In addition to data storage with the data loggers, the analog channels of the data loggers were also used to send the data to the computer for storage through a data acquisition board (DAQ AT-MIO-16XE, National Instruments, Austin, TX). Load cells required excitation voltage for the measurement, thus data logger was also used as a signal-conditioning unit.

All the sensors except infrared thermocouples were scanned every five seconds and the data was averaged every one minutes. The temperature of the plant canopy was measured by two stationary infrared thermocouples, mounted at the same height, only when the plant was positioned underneath the infrared sensors.

Complete calibration of the data loggers and data acquisition board was performed prior to the experiment.
4.10. Image acquisition and processing

In this study, image acquisition system consisted of two main components; a monochrome CCD camera (Pulnix TM-200, Pulnix America Inc., Sunnyvale, CA) and a 640 x 480 x 8 resolution frame grabber board (Matrox Meteor II Standard, Matrox Electronic Systems Ltd., Quebec, Canada) (Figure 4.15) installed in a personal computer.

The camera was mounted perpendicular to the horizontal plane at a height of 1.0 m above the turn-table. Images of each plant were captured every 15 minutes. The image seen by the camera was the top view of the plant canopy. Only light period images were captured. To reduce measurement errors due to air-movement-induced plant motion and other random electronic noises, four consecutive images of the same plant were acquired, the top projected area of each image was determined and they were averaged during the data analysis to obtain a single top projected canopy area value for that plant.

Figure 4.15: Components of the image acquisition system: a) CCD camera b) Frame grabber board.
Images of the plant top projected area were analyzed using Visilog 5.1 image processing software (Noesis S.A., Velizy, France). Image acquisition system generated 160 images per plant per ten hours of light period and a total of 960 images per ten hours from six plants. Since a very large number of images were to be processed, a macro was written to automate the image processing. The flow diagram of the image processing procedure is shown in Figure 4.16.
Figure 4.16: A flow diagram of the image processing procedure used for top projected canopy area measurement.
Image Processing

Gray level images were processed to remove noise. The noises were unwanted objects such as appearance of datalogger, moisture sensor, and cables in the original image. Thus, certain coordinates of the original image containing the noise were removed using a simple image editing procedure. Then, noise removed gray level images were binarized using a thresholding procedure. Thresholding is the transformation of an input gray level image \( f(x,y) \) to an output binary image \( g(x,y) \). Thus, the definition of thresholding was:

\[
g(x,y) = \begin{cases} 
1 & \text{if } \lambda_1 \leq f(x,y) \leq \lambda_2 \\
0 & \text{if } f(x,y) < \lambda_1 \text{ or } f(x,y) > \lambda_2 
\end{cases}
\] (4.32)

Here, \( g(x,y) = 1 \) for image elements of objects, and \( g(x,y) = 0 \) for image elements of the background or vice versa. \( \lambda_1 \) and \( \lambda_2 \) are the selected threshold gray levels. The schematic of the thresholding is illustrated in Figure 4.17.

![Graphical illustration of thresholding procedure.](image)

Figure 4.17: Graphical illustration of thresholding procedure.
A histogram of an image is the global information about that image. The histogram provides the frequency of the gray levels in the image, and makes operator's job easier to make a decision for a threshold value to differentiate the target object from a background in an image. Therefore, a histogram of a sample gray level image was used to determine the threshold levels for image binarization process. A sample image histogram obtained from an experimental image is illustrated in Figure 4.18. The histogram of the image shows that plant has a gray level of 100 or below while white background had gray levels ≥250. Therefore, the thresholding level can be selected anywhere from 100<λ<250. The thresholding level to binarize images was selected to be λ = 220 in this study. Figure 4.19 and 4.20 show a gray level and binarized image of the experimental plant, respectively.

Figure 4.18: Gray level histogram of a typical plant image of the experiments.
Figure 4.19: A typical gray level image of a plant used in the experiments.

Figure 4.20: Binarized image after noise removal with a threshold level $\lambda = 220$. 
Binarized images were used for blob analysis. In blob analysis, the area of the image was determined by the number of pixels providing the top projected area of the plant. The area of object, \( TPCA(X) \), was calculated by the number of pixels in the image \( (X) \), thus the definition of the area was:

\[
TPCA(X) = \sum_{i,j} g(x_i, y_j)
\]

\[ (4.33) \]

\[
g(x_i, y_j) = \begin{cases} 
1 & \text{if pixel lies within object } X \\
0 & \text{otherwise}
\end{cases}
\]

In the analysis of plant water stress detection in this study, both plant canopy area change due to growth and biotic canopy movement of the plants were used. However, TPCA data obtained from the blob analysis describes both canopy area change and biotic movement of the plant canopy (Li et al., 1997). Therefore, these two components were decoupled to determine percent movement of the plant canopy as:

\[
\text{Movement} = \frac{TPCA_i - TPCA_{\text{ave}}}{TPCA_{\text{ave}}} \times 100
\]

\[ (4.34) \]

where,

- \( \text{Movement} \) = movement of the plant canopy (\%)
- \( TPCA_i \) = top projected canopy area at time \( i \) (pixels)
- \( TPCA_{\text{ave}} \) = average of top projected canopy area (pixels)
Plant canopy expansion was further analyzed by calculating the coefficient of variation of the top projected canopy area (COV of TPCA). The coefficient of variation is expressed as the unit standard deviation as a percentage of the general mean. In this case, calculating coefficient of variation of TPCA provided the stability of the plant canopy movement as:

$$\text{COV}_{\text{TPCA}} = \frac{\sigma_{\text{TPCA}}}{\mu_{\text{TPCA}}} \times 100$$

(4.35)

where,

- $\text{COV}_{\text{TPCA}}$ = coefficient of variation of TPCA (%)
- $\sigma_{\text{TPCA}}$ = standard deviation of top projected canopy area
- $\mu_{\text{TPCA}}$ = mean of top projected canopy area
Figure 4.21 illustrates the complete schematic of the experimental setup used in this study. The whole system was computer controlled and automated. In order to achieve automation of the system, computer programs were written using C++ and C programming languages.
Computer based data acquisition and control system

An overall flow chart of the main program of the system is shown in Figure 4.22. The program for the plant health monitoring system was written in C++ programming language. The main program of the system was designed to achieve four main goals; motion control of the turn-table, image acquisition and storage, sensor readings, and irrigation control.

Two subroutines were called in the main module for motion control: 1) Subroutine "Home.C" and 2) Subroutine "Move1.C". The first subroutine was used to bring the turn-table to its home position using an electronic home switch. The first reason for homing the turn-table was to make sure that the position of the table was zero at the beginning of each revolution for positional accuracy during the revolution of the table. The second reason was not to allow the table to rotate more than 360 degrees due to possible problems which could be caused by sensor cables being twisted. The second subroutine was used to move the table forward using a ramp function.

In the image acquisition subroutine, four sequential images were acquired from each plant when the plant was under the camera. Each acquired image was registered and stored in the hard disk with the time of the measurement and the plant identification number.

The sensor reading subroutine scanned sensors through a data acquisition board. Analog channels of the data loggers were also used to send voltage signals from the sensors to the data acquisition board due to lack of channels in the board. Another reason for using the data loggers was that the excitation voltage signal was required to scan the load cells. In this case, the data loggers served as a signal conditioning unit. This
subroutine read signals from six load cells, six Theta-Probes, two infrared thermocouples, one dry bulb sensor, one humidity sensor, one radiometer, and one hot wire anemometer.

The main program was also designed to perform automatic irrigation. The irrigation control was based upon the readings from the Theta-Probe reading. A soil moisture set point was pre-selected to decide the time of the irrigation. When the actual soil moisture reading was below the threshold, an analog voltage signal was sent to a relay through the data acquisition board to trigger the water pumps. Then, the water was delivered by a drip emitter system.

The actual image of the experimental setup and instrumentation is shown in Figure 4.23. The detailed flow diagrams and source codes of the main and sub programs for the system are provided in Appendix C.
Motion control
1) Forward movement
   - using "ramp" function
2) Homing operation
   - bring the table to zero position

Image acquisition
1) Sequential acquisition
   - 4 images at once
2) Store images to hard disk

Sensor readings
1) Load cells (6)
2) Theta Probes (6)
3) IRTC (2)
4) Dry bulb sensor (1)
5) Humidity sensor (1)
6) Radiometer (1)
7) Hot wire anemometer (1)

Irrigation control
Soil moisture sensor base
irrigation control

Hardware components
- data acquisition board
- relays
- pumps
- drip emitters

4.22: Overall flow chart of the main program of the system.
Figure 4.23: The experimental setup and instrumentation
CHAPTER 5

RESULTS AND DISCUSSIONS

In this study, a non-contact, non-destructive, and plant-response-based water stress detection approach using crop water stress index with infrared thermometry and the top projected canopy area change obtained from plant images with machine vision applications for plants grown under controlled environment conditions was proposed. The experiments were repeated four times, three of which were under low humidity and the last one was conducted under a higher humidity condition.

5.1. Irrigation regime and water stress cycles

Figures 5.1 through 5.4 illustrate the history of irrigation regimes, water stress cycles, and the volumetric soil moisture change for plants in control and treatment group during each experiment. There were six potted New Guinea *Impatiens* plants in each experiment. Three of which were stressed plants and the other three were control plants. All six plants were kept well watered for two days at the beginning of each experiment. While the plants in the control group were well watered for the rest of experiments, the water was withheld from the plants in the treatment group until water stress was visually
observed by the operator. When the stress was observed, water was added to the media of the water stressed plants to bring soil moisture back to well-watered conditions.

Figure 5.1 through 5.4 illustrate irrigation regimes applied during the experiments. The arrows in Figures indicate the time of the human stress detection. The average volumetric soil moistures of control plants were about 0.71, 0.55, 0.50, and 0.68 m³m⁻³ during the first, second, third, and the fourth experiments, respectively (Figures 5.1.b, 5.2.b, 5.3.b, and 5.4.b). There were usually 5-7 days time intervals between stress cycles on the plants in the treatment group during the first three experiments under low humidity conditions while the stress cycle time intervals were 10-14 days between the stress cycles on the plants during the fourth experiment. The main reasons for the time interval differences for stress cycles between the first three and the last experiment were due to different environmental demand conditions, especially VPDair (2.11±0.03, 2.30±0.03, 2.22±0.2, and 1.16±0.12 kPa for the 1st, 2nd, 3rd, and the 4th experiments, respectively), and the amount of water applied to the plants. The breaks on the curves in Figure 5.4.b were a result of unavailable data on those days due to corrupted data files.
Figure 5.1: History of volumetric soil moisture: a) for plants in the treatment group b) for plants in the control group (First experiment).
Figure 5.2: History of volumetric soil moisture: a) for plants in the treatment group b) for plants in the control group (Second experiment)
Figure 5.3: History of volumetric soil moisture: a) for plants in the treatment group b) for plants in the control group (Third experiment)
Figure 5.4: History of volumetric soil moisture: a) for plants in the treatment group b) for plants in the control group (Fourth experiment).
5.2. Leaf temperatures as a response to water stress

The heat balance of a plant can be compared to the water balance of a tank supplied with water from a tap and fitted with an outlet near its base. When water is poured into the tank at a steady state, the level rises until the rate of outflow is precisely equal to the supply. If the supply is increased or if the outflow is restricted, the water level rises until a new equilibrium is reached. In the same manner, the temperature at any point within or on the surface of plant responds to changes in the supply or in the dissipation of heat. Transpiration is a very effective way for plants to dissipate heat. It is the evaporative cooling mechanisms of the plants. For instance, for each 102 g m\(^{-2}\) h\(^{-1}\) of transpiration, the effective radiation load on the leaf is reduced by 60000 cal m\(^{-2}\) h\(^{-1}\) (Gates, 1964).

The leaf to air temperature difference (\(T_c - T_a\)) is a direct result of leaf energy balance. The shortage of water may cause shifting release of excess energy from latent to sensible heat (De Lorenzi et al, 1993). Figures 5.5 shows the correlation between leaf to air temperature differential and measured evapotranspiration rate of the plants obtained from the first experiment. The difference between leaf and surrounding air was found to be inversely and linearly correlated to measured evapotranspiration rates for all the experiments. The same trends were obtained from the other experiments (Figure 5.6 through 5.8). In order to draw a global conclusion from the data and to show the effect of evapotranspiration rate on the leaf to air temperature differential, combined data obtained from all the experiments was plotted (Figure 5.9).
As seen from the Figures 5.5 through 5.9, a negative \((T_c - T_a)\) value indicates that transpiration is evaporatively cooling the leaf at a faster rate than it is being radiatively heated. The results showed that the leaf temperature of the plants been well-watered, in other words transpired at potential rate, were 3 to 4 °C less than the air temperature. Under water limiting conditions as the plants started to experience some level of water stress, the leaf to air temperature difference became closer to zero. The leaf temperatures of the further water-stressed plants were found to be 1 to 3 °C higher than the air temperature.

It was reported in the literature that the leaf temperature of greenhouse grown cucumbers (Yang et al., 1989) and chrysanthemums (Al-Shooshan et al., 1991) were found to be always lower than that of air while Pang (1992) found that the temperature of upper leaves of New Guinea Impatiens were 2 °C higher than the air temperature especially at noon time when the solar radiation load was relatively high. The inconsistency is more likely due to the fact that New Guinea Impatiens are colloquially known to be low light (low transpiration) plants; in contrast, cucumbers and chrysanthemums are high transpiration plants. Therefore, temporary water stress might be expected for New Guinea Impatiens during the periods of high solar radiation even though the plants are well watered. Because, for this plant, the transpiration rate is assumed to be limited by the maximum stomatal opening and the ability of the system to move water through the plant.

In the present study, however, less than maximum radiant load (usually higher than 300 W m\(^{-2}\) according to Pang, 1992) for New Guinea Impatiens was provided for the experiments. Therefore, it may be reasonable to make an argument that the increase in
leaf temperature on the plants in the treatment group was due to reduced evapotranspiration (less cooling effect).

This study revealed that the leaf temperatures of well-watered plants were consistently lower than the air temperature while the leaf temperatures of the water stressed plants were found to be close or above the air temperature as seen in Figures 5.10 through 5.13. The arrows in figures show the time of watering to the plants in the treatment group. The mean ($\mu$) and standard deviations ($\sigma$) of $T_C - T_A$ for well-watered plants were found to be $\mu = -1.76^\circ C$ with $\sigma = 0.43$; $\mu = -0.91^\circ C$ with $\sigma = 0.40$; $\mu = -2.62^\circ C$ with $\sigma = 0.57$, and $\mu = -0.85^\circ C$ with $\sigma = 0.35$ for the first, second, third, and the fourth experiments, respectively. The differences in leaf to air temperature differential of the well-watered plants might be due to the differences in vapor pressure deficit during the experiments ($2.11\pm0.03$, $2.30\pm0.03$, $2.22\pm0.2$, and $1.16\pm0.12$ kPa for the 1st, 2nd, 3rd, and the 4th experiments, respectively). The magnitude of leaf to air temperature difference under high VPD$_{air}$ conditions were more negative compared to those values obtained under low VPD$_{air}$. Differences in cultivars used in the experiments might have also contributed to the differences in leaf to air temperature differential of the well-watered plants.

The temperature of the stressed plants started to increase after a certain volumetric soil moisture reached which was at about 30%. Following the irrigation, the leaf temperatures of the stressed plants were usually decreased to the level of average leaf temperatures of well-watered plants. However, in the third experiment, it was observed that even though the leaf temperatures of the stressed plants were lowered following the irrigation, their temperature were still higher than those of well-watered plants.
Figure 5.5: The correlation between measured leaf to air temperature differential and measured evapotranspiration rate (First experiment).
Figure 5.6: The correlation between measured leaf to air temperature differential and measured evapotranspiration rate (Second experiment).
Figure 5.7: The correlation between measured leaf to air temperature differential and measured evapotranspiration rate (Third experiment).
Figure 5.8: The correlation between measured leaf to air temperature differential and measured evapotranspiration rate (Fourth experiment).

\[ y = -0.0154x + 2.0905 \]

\[ R^2 = 0.65 \]
Figure 5.9: The correlation between measured leaf to air temperature differential and measured evapotranspiration rate (combined data for all experiments).
Figure 5.10: The history of leaf and air temperature differentials of stressed and well-watered plants (First experiment). Arrows in the graph indicate watering events immediately following human identification of the water stress.
Figure 5.11: The history of leaf and air temperature differentials of stressed and well-watered plants (Second experiment).
Figure 5.12: The history of leaf and air temperature differentials of stressed and well-watered plants (Third experiment).
Figure 5.13: The history of leaf and air temperature differentials of stressed and well-watered plants (Fourth experiment).
5.3. Crop water stress index, evapotranspiration rates, and leaf temperatures

The results of crop water stress index (CWSI-modeled) and measured evapotranspiration rates (ETm) obtained from the experiments are presented in Figures 5.14 through 5.21. Figures 5.14, 5.16, 5.18, and 5.20 show CWSI and ETm values for plants in treatment groups while Figures 5.15, 5.17, 5.19, and 5.21 illustrate the data for plants in well-watered group.

The average measured evapotranspiration rates of well-watered plants were 287.6±25.6, 262.44±44.5, 338.4±24.3, and 184.2±32.5 g m\(^{-2}\) h\(^{-1}\) during the experiments, consecutively (Figures 5.15, 5.17, 5.19, and 5.21). The water was withheld from the plants in the treatment group on the second day in each experiment. The average crop water stress index values of well-watered plants were about zero and were unchanged throughout the experiments while the crop water stress index values of the treatment group plants increased from about zero to their highest in response to decreased availability of soil water that led to reduced evapotranspiration rates and increased plant leaf temperatures.

In the first experiment, average ETm rates and CWSI values of the plants in the treatment group were same as those of well-watered plants until the 4\(^{th}\) day (Figure 5.14 and 5.15). The evapotranspiration rate of stressed plants were decreased 60% to 114 g m\(^{-2}\) h\(^{-1}\) when compared to the average ETm rates of the well-watered plants. This resulted in increased leaf to air temperature differentials (see Figure 5.10) and caused CWSI values to increase from about zero to 0.5-0.6 (Figure 5.14). The stress was observed by the operator on the 5\(^{th}\) day and the plants were irrigated to help the plants
recover from the water stress. Following the irrigation, the evapotranspiration rate increased and reached their potential rates. Due to the evaporative cooling of plants through transpiration, the leaf temperatures of the plants were decreased and CWSI was also reduced to zero. The recovery of the plants from stress condition depended on the degree of water stress that the plant was subjected to. It took at least one day for the plants to recover from stress and transpire at the potential rate again. At the same time, CWSI values return to close to zero. Jackson et al. (1981), Howell et al. (1984), Saha et al. (1986), Jones (1991), and Fernandez and Turco (1998) reported similar results with field crops. These trends observed on the plants for the second and third cycles of stress were the same during the experiment. However, the time of the occurrence of the second and third stress cycles differed depending on the amount of water added after previous visual detection of the water stress. Other environmental conditions such as air temperature, vapor pressure deficit, and the light intensity were almost constant during the experiment. The maximum CWSI values of the three plants in the first experiment were found to be 0.57, 0.77, and 0.69. CWSI values started to increase at least one day prior to the time of human stress detection as seen on Figure 5.14.

Figures 5.16 and 5.17 illustrate the history of CWSI and ETm of the plants in the treatment and well-watered groups during the second experiment, respectively. The response of CWSI to changes in evapotranspiration rates was similar to those obtained in the first experiment. As seen in Figure 5.17, there was a reduction on evapotranspiration rates on well-watered plants. This was caused by a power outage which happened during the experiment (shown with arrows on the figure). This did not affect the crop water stress index values of well-watered plants while it resulted in small peaks, designated
with letter "A" in Figure 5.16, on CWSI values of stressed plants depending on the dynamic responses of leaf temperatures of plants to changes in environmental conditions in treatment group compared to those in well-watered group. It is assumed that the leaf temperature of a stressed plant took longer time to drop to a certain value, due to less demand caused by less radiant load and VPD, when compared to a well-watered plant.

The maximum values of crop water stress index were found to be 0.96, 0.61, and 0.68 for the first, second, and the third stressed plant in the second experiment, respectively (Figure 5.16). In addition, crop water stress index as well as the leaf to air temperature differential increased at least one day prior to the time of visual stress detection as can be seen on Figure 5.11 and 5.16.

The history of CWSI and ETm rates of stressed and well-watered plants obtained from the third experiment is shown in Figures 5.18 and 5.19, respectively. The average ETm of well-watered plants was found to be 338.4 g m$^{-2}$ h$^{-1}$ in this experiment. In this experiment, CWSI values of stressed plants started to increase when the ETm rate was reduced 30-45% from the average ETm rate of well-watered plants as seen on the 4$^{th}$, 10$^{th}$, and 14$^{th}$ days. However, CWSI values reached to their maximum when ETm rate was reduced about 60% from those of well-watered plants. The stress was visually detected on the 5$^{th}$, 11$^{th}$, and 15$^{th}$ days for first plant; 5$^{th}$, 11$^{th}$, and 15$^{th}$ days for the second plant; and 5$^{th}$, 11$^{th}$, and 16$^{th}$ days for the third plant for the first, second, and the third stress cycles, respectively. The maximum values of CWSI were found to be 0.53, 0.49, and 0.52 for the first, second, and the third stressed plants. The average CWSI value of well-watered plants was also found to be zero in this experiment (Figure 5.19). Following the irrigation, even though the CWSI (Figure 5.18) and leaf to air temperature
differential of the stress plants (Figure 5.12) were reduced, the values were found to be still higher than those of well-watered plants.

Similar response of crop water stress index to changes in evapotranspiration rates were observed in the fourth experiment (Figures 5.20 and 5.21). However, it took at least one to two days for the plant to recover from water stressed condition. One of the reasons for this can be explained by considering the fact that the humidity level was high during this experiment, thus the driving force was low (\(\text{VPD}_{\text{air}} = 1.16 \, \text{kPa}\)). When the plants experience the stress, the water potential in the leaf is reduced. This results in higher water potential gradient between the soil and the leaf to move water from the soil into the plant. Therefore, the stressed plants should still be able to uptake certain amount of water from the soil to rehydrate (Salisbury and Ross, 1992). However, the rate of uptake may be affected by the level of damage caused by stress to root hairs of the plant, the species, the growth stage, and the environmental demand. Also, low \(\text{VPD}_{\text{air}}\) during this experiment might have reduced New Guinea Impatiens' naturally low transpiring capability. Therefore, this might have caused the water stressed plants to take longer to recover from the stressed conditions in this experiment. The maximum values of CWSI were found to be 0.56, 0.57, and 0.65 for the first, second, and the third stressed plant, respectively. The CWSI values of the plants in the control group remained close to zero throughout the experiment.
Figure 5.14: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of water-stressed plants (First experiment)
Figure 5.15: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of well-watered plants (First experiment).
Figure 5.16: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of water-stressed plants (Second experiment).

*The curve is continuous during the power outage because the curves shown in the graph are obtained from 5hr averaged data.
Figure 5.17: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of well-watered plants (Second experiment).
Figure 5.18: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of water-stressed plants (Third experiment).
Figure 5.19: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of well-watered plants (Third experiment).
Figure 5.20: The history of measured evapotranspiration (ETm) rate and crop water stress index (CWSI) of water-stressed plants (Fourth experiment).
Figure 5.21: The history of measured evapotranspiration rate (ETm) and crop water stress index (CWSI) of well-watered plants (Fourth experiment).
The correlations between modeled crop water stress index and measured evapotranspiration rate are provided in Figures 5.22a, b, c, and 5.23 for each experiment. Crop water stress index was increased when evapotranspiration rate of the plants was reduced. Crop water stress index were found to be inversely and linearly related to measured evapotranspiration rate with $R^2$ values of 0.79, 0.50, 0.78, and 0.64.

The leaf temperature of a water-stressed plant can increase and will be warmer than leaf temperatures of well-watered plants due to reductions in evaporative cooling through latent heat loss. This statement was also supported by the findings of the present study. It was found that crop water stress index increased as leaf to air temperature difference became more positive. They were linearly correlated with $R^2$ values of 0.82, 0.72, 0.83, and 0.87 (Figures 5.24a, b, c, and 5.25).

Figures 5.26 and 5.27 show the correlation between model predicted CWSI and measured CWSI for each experiment. The model predicted CWSI was correlated to measured CWSI with $R^2$ values of 0.83, 0.50, 0.79, and 0.76 for the 1st, 2nd, 3rd, and the 4th experiments, respectively. Figure 5.28 shows the correlation for the combined data for all the experiments.
Figure 5.22: The correlation between crop water stress index and measured evapotranspiration rate for a) First experiment b) Second experiment c) Third experiment.
Figure 5.23: The correlation between crop water stress index and measured evapotranspiration for the Fourth experiment.
Figure 5.24: The correlation between crop water stress index and leaf to air temperature differential ($T_c - T_a$) for a) First experiment b) Second experiment c) Third experiment.
Figure 5.25: The correlation between crop water stress index and leaf to air temperature differential ($T_c - T_a$) for the Fourth experiment.
Figure 5.26: Measured vs. modeled crop water stress index for a) First, b) Second, and c) Third experiments.
Figure 5.27: Measured vs. modeled crop water stress index for the Fourth experiment.

\[ y = 0.6354x + 0.0136 \]

\[ R^2 = 0.76 \]
Figure 5.28: Measured vs. modeled crop water stress index for combined data obtained from all the experiments.
5.4. Plant resistances and water stress

5.4.1. Stomatal functions and water stress

Salisbury and Ross (1982) reported that experimental evidence was not conclusive on stomatal function. Stomatal response has been attributed to carbon dioxide concentration in the leaf. A high CO$_2$ gradient between the leaves and free air causes stomata to open. This gradient could be induced by decreasing the CO$_2$ concentration in the leaf or by increasing the concentration in the free air. It was reported that stomata responded to changes in light levels. Sharkey and Raschke (1981) found that blue light (430 to 460 nm) was more effective than red light (630 to 680 nm) in producing a given stomatal opening. Yang (1988) and Pang (1992) also related stomatal opening to light levels and found good agreement between stomatal resistance and solar radiation.

Stomatal control was related to water status of the leaf (Jones, 1991). Stomatal movement depends on changes in turgor pressure inside the guard cells and in the adjacent epidermal cells. Turgor pressure changes can result either from a change in total water potential of the guard cells as the supply or loss of water changes, or from active changes in osmotic potential. K$^+$ ion enters into the guard cell when CO$_2$ decreases in the guard cells by creating an osmotic gradient for water to enter, this pressurizes guard cells and stomate opens.
There is also evidence that plant growth regulator abscisic acid (ABA) involves in regulating stomatal responses, especially under water stress conditions. Externally applied ABA closes stomata. The level of endogenous ABA increases rapidly when the plant experiences water stress.

Figures 5.29a, b, c, and 5.30 illustrate the relationship between total canopy resistance (sum of $r_z$ and $r_{av}$) and the modeled crop water stress index. Total canopy resistance was determined by solving plant energy balance equation with measured evapotranspiration rate (using equation 4.14). As seen from the figures, crop water stress index was positively and linearly correlated to total canopy resistance. All four experiments revealed that crop water stress index increased as canopy resistance increased. Jones et al. (1997), Jalali-Farahani et al. (1994), Hattendorf et al. (1990), and Nielsen and Anderson (1989) reported similar results. One of the reasons for increasing canopy resistance under water stress might be due to decreases in leaf water status. In general, stomata respond to factors that lower the leaf water potentials in a way to minimize further increases in stress.
Figure 5.29: The correlation between total canopy resistance and crop water stress index for a) First experiment b) Second experiment c) Third experiment.
Figure 5.30: The correlation between total canopy resistance and crop water stress index for the Fourth experiment.
5.4.2. Plant resistance and soil water availability

The relationship between canopy resistance (calculated by equations 4.20 and 4.27) and volumetric moisture content of the soil is shown in Figures 5.31 through 5.34. Canopy resistance of the plants increased as soil water became limited. The results showed that canopy resistance remained almost constant between 85-35% volumetric soil moisture content. Volumetric soil moisture contents less than 30% caused canopy resistance to increase exponentially. The values of canopy resistance under well-watered conditions obtained from this study were similar to those found by Mankin (1994) and Pang (1992) who also studied New Guinea Impatiens under growth chamber and greenhouse conditions.
Figure 5.31: Relationship between canopy resistance and volumetric soil water content obtained from the First experiment.
Figure 5.32: Relationship between canopy resistance and volumetric soil water content obtained from the Second experiment.

\[ y = 1453 + 2.2 \times 10^5 e^{-25.5x} \]

\[ R^2 = 0.43 \]
Figure 5.33: Relationship between canopy resistance and volumetric soil water content obtained from the Third experiment.

\[ y = 749.7 + 4642e^{-9.2x} \]

\[ R^2 = 0.64 \]
Figure 5.34: Relationship between canopy resistance and volumetric soil water content obtained from the Fourth experiment.
5.5. Soil moisture tension and crop water stress index

Soil moisture is usually expressed in one of two ways. Soil moisture content ($Q_v$) is a measure of the actual water content, and is defined as the percentage volume of a moist soil occupied by water. Soil moisture potential (soil water tension) on the other hand is an indirect measure of water content, and it is the energy necessary to extract water from the soil matrix. This concept is of value in estimating the availability of water for plant use. In general, in a given soil the water tension increases as volumetric soil moisture decreases but not in a linear fashion. The relationship between volumetric soil moisture content and soil water tension can be characterized by the soil moisture release curve.

In this study, the soil moisture release curve for the growing medium used in the experiments was obtained by a laboratory test and the resulting curve is illustrated in Figure 5.35. A logarithmic mathematical model explained the relationship between soil water tension and volumetric soil moisture content with an $R^2$ value of 0.98.

In the experiments, the volumetric soil moisture content was measured and soil water tension values were extracted by using the mathematical relationship obtained from the soil moisture release curve. As the soil water tension increases, water becomes less available to the plants. It is relatively easy to extract moisture from a wet soil but as it dries out it becomes increasingly difficult to remove additional units.
Figures 5.36 through 5.39 illustrate the relationship between soil water tension and crop water stress index. The data obtained from the first, third, and fourth experiments showed that crop water stress index increased exponentially as soil water tension increased. However, a linear regression equation better explained the relationship for the data obtained from the second experiment. Crop water stress index and soil water tension data from the fourth experiment were more scattered as shown in Figure 5.39. Possible reasons for this was that it took longer for the plants in the treatment group to recover from water stress under high humidity conditions, thus the crop water stress index values of the plants in the treatment group remained high even though the soil medium was wet and the tension level was low. As seen from the figures, soil water tensions 10 kPa and higher levels caused crop water stress index values to increase.
Figure 5.35: The soil moisture release curve for the growing medium used in the experiments (The growing medium consisted of 29% sphagnum peat moss, 20% coarse perlite, 20% grade 2 vermiculite, and 31% composted pine bark). A, B, and C represent data collected from three independent laboratory experiments.
Figure 5.36: Correlation between soil water tension and crop water stress index obtained from the First experiment.

\[ y = 1.01(1 - 0.98^x) \]

\[ R^2 = 0.82 \]
Figure 5.37: Correlation between soil water tension and crop water stress index obtained from the Second experiment.
Figure 5.38: Correlation between soil water tension and crop water stress index obtained from the Third experiment.
Figure 5.39: Correlation between soil water tension and crop water stress index obtained from the Fourth experiment.
5.6. Plant growth and movement

In the analysis of plant water stress detection using image processing, top projected canopy area (TPCA) images of the plants were obtained. Plant canopy change due to growth and biotic plant movement was analyzed.

5.6.1. Plant canopy expansion and growth

Figures 5.40a, 5.41a, 5.42a, and 5.43a illustrates the history of top projected canopy area (TPCA) of the treatment group plants while Figures 5.40b, 5.41b, 5.42b, and 5.43b show TPCA of control group plants. Only light period images were captured, TPCA extracted, and the data shown in the figures. As the plants in the treatment group experienced water stress, plant growth and cell expansion was inhibited temporarily due to reduced leaf water potentials in plant cells. This was reflected as a decrease in top projected canopy area of the stressed plants. Figure 5.44 illustrates detailed history of top projected canopy area for a water stressed plant. As can be seen from the graph, the slope of the TPCA curve becomes close to zero as stress developed on the plant (pointed by the arrows in the figure). Following the irrigation, the plants continued to grow. The valleys of the TPCA curves for the stressed plants indicate the timing of the human stress detection, shown with arrows in Figures. Following the stress detection, stressed plants were irrigated and allowed to recover from the stress condition.
The graphs for the stressed plants showed that TPCA was not reduced dramatically during the second and third stress cycles when compared to reduction of TPCA in the first stress cycles. This observation was consistent for all the experiments. As the plants experience water stress, the cell wall of the plant is usually thickened, this is called hardening. This might be one of the reasons that less TPCA reduction was seen on the plants during the second and the third cycles. As the plants were hardened after the first stress cycle, lesser number of plant leaves collapsed due to water stress during the second and the third stress cycles.

The change of TPCA was mainly influenced by the motions of fully developed leaves in this study. Therefore, the analysis of motions of young growing leaves was not attempted with the use of TPCA in this study. However, still images of a water-stressed plant taken during one stress cycle were collected and a movie was made by using a commercial software for further motion analysis. The animated plant movie allowed a closer look of the plants dynamic motion under water stress condition. The animated plant images also supported the conclusion that fully developed old leaves collapsed first as the plant started experiencing water stress. Similar conclusions were reported by Nyakwende et al. (1996) for tomato plants, Revollon et al. (1998) for ornamental shrub. Seginer et al. (1992) also reported that fully expanded leaves of tomato plants showed linear vertical motions under water stress while young growing leaves had complex motions and were less useful for monitoring water stress level.

The plants in the well-watered group in the fourth experiment had smaller TPCA gains at day 18 and 19 compared to second and third experiments that had similar plant sizes at the beginning of the experiments (Figures 5.41, 5.42, and 5.43). Therefore, it
may be reasonable to make an argument that the level of humidity and the evapotranspiration rate affected the growth during the fourth experiment.

The TPCA gains of the plants in the treatment groups and the control groups for each experiment are illustrated in Table 5.1. The TPCA gain was defined as the change of TPCA from the beginning of the experiment to the end of the experiment expressed as a percentage. The TPCA gains of the plants in the treatment groups were found to be less than those values for the plants in the control group in all experiments. A two sample T-test was used to evaluate the effect of water stress on plant growth and canopy expansion by comparing TPCA gains of the plants in the treatment group to the TPCA gains of the plants in the control group. The test results suggested that the water stress did not significantly affect plant growth and canopy expansion of the plants in the treatment group for the first experiment at \( \alpha=0.05 \) (P-value=0.87) or second experiment at \( \alpha=0.05 \) (P-value=0.07). However, there was statistical evidence that the TPCA gains of the plants in the treatment groups were significantly different than the TPCA gains of the plants in the control groups for the third and the fourth experiments at \( \alpha=0.05 \) with P-values of 0.03 and 0.04, respectively. The results of the two sample T-test are provided in Appendix E. This preliminary data suggests that stress does reduce TPCA gain but experiments designed to evaluate this need to be designed and conducted to make a more definite statement.
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Table 5.1. TPCA gains of the plants in the treatment groups and the control groups.
Figure 5.40: Top projected canopy area change for plants in a) treatment group b) well-watered group obtained from the First experiment.
Figure 5.41: Top projected canopy area change for plants in a) treatment group b) well-watered group from the Second experiment.
Figure 5.42: Top projected canopy area change for plants in a) treatment group b) well-watered group from the Third experiment.
Figure 5.43: Top projected canopy area change for plants in a) treatment group b) well-watered group from the Fourth experiment.
Figure 5.44: Detailed history of top projected canopy area for a typical water stress plant.
5.6.2. Plant biotic movement under water stress

Both leaf expansion and plant biotic movement affect top projected canopy area (TPCA) measurement. Therefore, it was of interest to decouple plant biotic movement from the canopy expansion for a better understanding of the plant motion under water stress and well-water conditions. Figures 5.45, 5.47, 5.49, and 5.51 illustrate the biotic movement of the treatment group plants while Figures 5.46, 5.48, 5.50, and 5.52 are used for well-watered plants for all the experiments.

The plant movement was defined as the difference between instantaneous TPCA’s and average TPCA’s over five hours, normalized by average TPCA’s over five hours (Equation 4.34). It was found that the movement was mostly equal or less than ±1% when the plants were under well-watered conditions. The plants appeared to be at upright position when the lights were turned on. Then, the leaves showed a gradual downward movement, causing increased TPCA measurements, that is the reason why the values of the movement of the plants under well-watered conditions started with negative values and moved toward positive values during the light period as plant canopies returned to their original state before light period (shown with letter "A" in Figures 5.46a). This was a typical observation and similar trend was also observed on the plants in the treatment group when they were under well-watered conditions. Figure 5.53 provides the movement of typical well-watered experimental plant and better explains this observation.
However, when the evapotranspiration rate started to decrease, plant leaf water potential decreases and the water stressed plant becomes less turgid, thus movement become less dynamic. In this study, as the stress developed on the plants, movement started to decline (more negative values) dramatically due to less turgidity in plant leaves, indicating that actual top projected canopy area of the stressed plant deviated from the average TPCA values.

In the third experiment, the movement of the well-watered plants was much higher than those values obtained from the well-watered plants of the first, and the second experiments, and the magnitude of movement were as close as those of stressed plants. The percent movement values of well-watered plants were also found to be as large as the plants in the treatment group during the fourth experiment (Figures 5.52 a, b, and c). Transpiration demand difference, due to a higher relative humidity during the fourth experiment, may have contributed to the different movement patterns in addition to cultivar differences compared to other experiments.
Figure 5.45: Instantaneous biotic movement of stressed plants of the First experiment: a)Plant1-S b)Plant2-S c)Plant3-S.
Figure 5.46: Instantaneous biotic movement of well-watered plants of the First experiment: a) Plant1-C b) Plant2-C c) Plant3-C.
Figure 5.47: Instantaneous biotic movement of stressed plants of the Second experiment:
a) Plant1-S b) Plant2-S c) Plant3-S.
Figure 5.48: Instantaneous biotic movement of the well-watered plants of the Second experiment: a) Plant1-C b) Plant2-C c) Plant3-C.
Figure 5.49: Instantaneous biotic movement of the stressed plants of the Third experiment: a) Plant1-S b) Plant2-S c) Plant3-S.
Figure 5.50: Instantaneous biotic movement of the well-watered plants of the Third experiment: a) Plant1-C b) Plant2-C c) Plant3-C.
Figure 5.51: Instantaneous biotic movement of stressed plants of the Fourth experiment: a) Plant1-S b) Plant2-S c) Plant3-S.
Figure 5.52: Instantaneous biotic movement of well-watered plants of the Fourth experiment: a) Plant 1-C b) Plant 2-C c) Plant 3-C.
Figure 5.53: A typical detailed movement curve for a well-watered plant during the light period.
The movement (obtained by equation 4.34) presented in the previous section evaluates the instantaneous change of actual TPCA values normalized by the average TPCA's. The movement of the plant canopy was also further evaluated by analyzing the coefficient of variation of the TPCA values (determined by equation 4.35). This was called COV of TPCA. It was defined as the ratio of the standard deviation of TPCA's (over five hours) to the average of TPCA's (over five hours). The COV of the TPCA value evaluates the stability of the plant movement.

Figures 5.54a, 5.55a, 5.56a, and 5.57a illustrate percent TPCA change of the treatment group plants and 5.54b, 5.55b, 5.56b, and 5.57b show the changes for the plants in the well-watered group during the experiments. It was consistent in all the experiments that plants under well-watered conditions exhibited less than 0.7 percent value of COV of TPCA. In the first and second experiments, COV of TPCA of well-watered plants were found lower than 0.5%. The peaks indicated with arrows in the graphs on the COV of TPCA curves coincide with the time of the human stress detection for the stressed plants. The values were found to be higher than 0.5% and reached as high as 7.5%, 6.5%, and 3% at the time of human stress identification during the first, second, and the third experiments, respectively.

The COV of TPCA of well-watered plants was found to be less than 0.3% during the fourth experiment (Figure 5.57b). However, in this experiment, the COV of TPCA for the plants in the treatment group during the stress cycles were not considerably different than those of well-watered plants. The leaves of the stressed plants under the higher humidity condition tended to stay horizontal when compared to those results obtained from other experiments under the lower humidity conditions.
Figures 5.58 and 5.59 show the relationship between COV of TPCA and evapotranspiration rates of the plants in the treatment group for the experiments. When the evapotranspiration rate was high, the COV of TPCA was low. This might be attributed to the higher water potentials and turgor pressures in the leaves. As the evapotranspiration rate further deviated from its potential rate, the COV of TPCA was increased. The trends were similar for the first three experiments conducted under low humidity conditions. Nonetheless, the same trend was not clearly observed in the fourth experiment (Figure 5.59b). The water supply was limited to the plants, yet the driving force for evapotranspiration was also low due to high relative humidity during this experiment. Therefore, even though the plants in the treatment group experienced water stress as evaluated in terms of decreasing water availability, increased leaf to air temperature differential, increased CWSI and reduced evapotranspiration rates (Figures 5.4, 5.13, and 5.20), the COV of TPCA of the plants in the treatment group were found to be similar both at low and high evapotranspiration rates. This might also be caused by the morphological characteristics of the cultivar (Paradise) used as well as high humidity conditions during this particular experiment.
Figure 5.54: The COV of TPCA for the plants in a) the treatment group b) well-watered group obtained from the First experiment.
Figure 5.55: The COV of TPCA for the plants in a) the treatment group b) well-watered group obtained from the Second experiment.
Figure 5.56: The COV of TPCA for the plants in a) the treatment group b) well-watered group obtained from the Third experiment.
Figure 5.57: The COV of TPCA for the plants in a) the treatment group b) well-watered group obtained from the Fourth experiment.
Figure 5.58: Relationship between the coefficient of variation of TPCA (COV of TPCA) and measured evapotranspiration rates of the plants in the treatment group for (a) the First and (b) the Second experiments.
Figure 5.59: Relationship between the coefficient of variation of TPCA (COV of TPCA) and measured evapotranspiration rates of the plants in the treatment group for (a) the Third and (b) the Fourth experiments.
5.7. Establishing baselines for CWSI and TPCA for early stress detection

The main goal of this study was early water stress detection using crop water stress index and top projected canopy area change of the plants. It was of interest to establish the baselines for CWSI and TPCA in order to determine the earliest time at which the techniques were able to identify the occurrence of the water stress. Therefore, baselines were established by using a parametric approach.

Figures 5.60 illustrate a two-dimensional plot of original data points having coordinate values of crop water stress index and COV of TPCA obtained from the experiments. Black dots in the graphs show the data points for the plants in the well-watered group while gray dots were used to represent the data obtained from the plants in the treatment groups. As can be seen from the figures, data points in the treatment group represent plant status both during well-watered and water-stressed periods. Reliable water stress detection may be established providing that water stressed plants can be identified in a timely fashion.

The water status classification of the plants was performed by first examining the distribution of the data points of the well-watered plants. Because the classification approach was parametric and was based on the mean and the standard deviations of normally distributed data sets, it was necessary to examine the normality of the data of the plants in the well-watered group. The normality of the data, both CWSI and COV of TPCA for each experiment, was tested by using two normality tests provided in Minitab statistics software. They were I) Ryan-Joiner normality test (a correlation based test), and II) Kolmogorov-Smirnov normality test (a chi-square based test). The data was assumed
to be normally distributed if the data showed a satisfactory distribution at least in one of these two tests. In the normality tests, the vertical axis had a probability scale while the horizontal axis had a data scale. A least-squares line was fit to the plotted points and drawn on the plot for reference. The line formed an estimate of the cumulative distribution function for the population from which the data were drawn. The resulting P-value for the test was provided to test the null hypothesis which was the normality of the distribution of the data. The data was normally distributed when the resulting P-value was higher than the significance level selected. The significance level was selected to be $\alpha = 0.01$.

In the analysis, the original data (CWSI and COV of TPCA) was first tested for the normality. In some cases, the original data did not satisfy the normal distribution especially due to large values on the tails of the distribution deviating from the least-squares line (Figure 5.61a). Therefore, power transformation ($x^{1/2}$) was applied to the data and the normality was tested again with the transformed data set. The power transformation $x^{1/2}$ helped to shrink the large values of the data set and approximated the data better to a normal distribution as seen in Figure 5.61b (Johnson and Wichern, 1988). The results of the normality tests are provided in Appendix F. All the data were assured normality.
Figure 5.60: Crop water stress index vs. coefficient of variation of TPCA (COV of TPCA) of the plants in the treatment and well-watered groups obtained from the a)First, b)Second, c)Third, and d)Fourth experiments.
Figure 5.61: Data transformation effect on the distribution of the data set: a) normality test result with original data set b) normality test result after data transformation.
After testing the normality of the data set of well-watered group, the baselines for both CWSI and COV of TPCA were determined by using the mean plus three standard deviations ($\mu + 3\sigma$) of the data points of the well-watered plants. Using mean plus three standard deviations, it made possible to define the distribution of the data of the well-watered plants at 99% confidence interval. Thus, those data points of the treatment group which have a value less than $\mu + 3\sigma$ of the well-watered group were considered to be representing well-watered plants; those data points have a value greater than $\mu + 3\sigma$ were classified as plants under water stress.

After the classification, the classification results were re-plotted using original data as shown in Figures 5.62 through 5.65 for the experiments. Figures 5.62a, 5.63a, 5.64a, and 5.65a illustrate the classification of the data for well-watered and stressed groups using only CWSI as a classifier for each experiment. From the results of classification, the CWSI baselines for early water stress detection were found to be 0.14, 0.12, 0.20, and 0.10 for the first, second, third, and the fourth experiments, respectively. Furthermore, the data was classified into well-watered and stressed plants using only COV of TPCA as a classifier (Figures 5.62b, 5.63b, 5.64b, and 5.65b). Using COV of TPCA for classification, the earliest time of the water stress detection was possible when the COV of TPCA baselines were 0.40, 0.55, 0.70, and 0.36 for the first, second, third, and the fourth experiments, respectively.
Figure 5.62: The baselines for early stress detection using a) Only CWSI b) Only COV of TPCA for the First experiment.
Figure 5.63: The baselines for early stress detection using a) Only CWSI b) Only COV of TPCA for the Second experiment.
Figure 5.64: The baselines for early stress detection using a) Only CWSI b) Only COV of TPCA for the Third experiment.
Figure 5.65: The baselines for early stress detection using a) Only CWSI b) Only COV of TPCA for the Fourth experiment.
Tables 5.2 through 5.9 illustrate the timing and the performance of using the two indices for early detection of water stress. Detection times of water stress using only CWSI as an indicator are given in Tables 5.2, 5.4, 5.6, and 5.8 while Tables 5.3, 5.5, 5.7, and 5.9 show time of the detection using COV of TPCA as an indicator. In the tables, the first column shows the order of the stress cycles, the second column identifies the plant, the third column provides the earliest time of stress detection by the sensing technique prior to the time of human stress detection. The values in the parenthesis in the third column show the values of CWSI and COV of TPCA at the time of stress detection by the sensing techniques. In addition, the value of CWSI and COV of TPCA at the time of human stress detection is also provided in the table for comparison. The initials "HD" in the tables stands for stress detection by human.

The results showed that CWSI was able to detect the stress 5 to 29 hours prior to the visual stress detection for the first and second experiments while the time of stress detection by CWSI were found to be 19 to 48 hours and 24 to 115 hours prior to visual detection for the third and fourth experiments, respectively (Tables 5.2, 5.4, 5.6, and 5.8). On the other hand, using the COV of TPCA provided usually 5 hours prior detection of water stress to human detection in the first and second experiments while the timing of the stress detection was found to be the same as the stress detection by human in the third and the fourth experiments (Tables 5.3, 5.5, 5.7, and 5.9). The success rate of CWSI baseline was 89% to identify and detect the time of stress detection by human for the first experiment. Established CWSI baseline was able to identify and detect the time of human stress detection for the 2nd, 3rd, and the 4th experiments. The COV of TCA baselines for the first and the second experiments were also identified the time of stress detection by
human, however the success rate of this baseline was found to be 89% and 73% for the third and the fourth experiments, respectively. These results suggested that early and non-contact detection of plant water stress using CWSI was more successful and was quicker method compared to COV of TPCA obtained by machine vision evaluation of plant images.
### Table 5.2: Timing of water stress detection using CWSI baseline of 0.14 for the First experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>CWSI based detection prior to HD</th>
<th>CWSI at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>19 hr before HD (0.290)</td>
<td>0.569</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>19 hr before HD (0.383)</td>
<td>0.694</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>24 hr before HD (0.140)</td>
<td>0.458</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>5 hr before HD (0.372)</td>
<td>0.556</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>24 hr before HD (0.145)</td>
<td>0.767</td>
</tr>
<tr>
<td>3</td>
<td>2</td>
<td>29 hr before HD (0.149)</td>
<td>0.677</td>
</tr>
</tbody>
</table>

### Table 5.3: Timing of water stress detection using COV of TPCA baseline of 0.40 for the First experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>COV of TPCA based detection prior to HD</th>
<th>COV of TPCA at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>5 hr before HD (3.196)</td>
<td>4.967</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>5 hr before HD (0.539)</td>
<td>1.764</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>29 hr before HD (0.805)</td>
<td>4.239</td>
</tr>
</tbody>
</table>

Table 5.2: Timing of water stress detection using CWSI baseline of 0.14 for the First experiment (HD: stress detection by human).
### Table 5.4: Timing of water stress detection using CWSI baseline of 0.12 for the Second experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>CWSI based detection prior to HD</th>
<th>CWSI at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>19 hr before HD (0.355)</td>
<td>0.966</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>24 hr before HD (0.186)</td>
<td>0.732</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>24 hr before HD (0.233)</td>
<td>0.595</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>29 hr before HD (0.254)</td>
<td>0.605</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>19 hr before HD (0.208)</td>
<td>0.679</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>5 hr before HD (0.555)</td>
<td>0.597</td>
</tr>
</tbody>
</table>

### Table 5.5: Timing of water stress detection using COV of TPCA baseline of 0.55 for the Second experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>COV of TPCA based detection prior to HD</th>
<th>COV of TPCA at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>5 hr before HD (2.297)</td>
<td>4.245</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>-</td>
<td>1.072</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>29 hr before HD (0.991)</td>
<td>6.417</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>5 hr before HD (0.595)</td>
<td>1.585</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>24 hr before HD (1.017)</td>
<td>1.751</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>5 hr before HD (0.670)</td>
<td>2.771</td>
</tr>
</tbody>
</table>

Table 5.4: Timing of water stress detection using CWSI baseline of 0.12 for the Second experiment (HD: stress detection by human).

Table 5.5: Timing of water stress detection using COV of TPCA baseline of 0.55 for the Second experiment (HD: stress detection by human).
### Table 5.6: Timing of water stress detection using CWSI baseline of 0.20 for the Third experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>CWSI based detection prior to HD</th>
<th>CWSI at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>43 hr before HD (0.321)</td>
<td>0.49</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>48 hr before HD (0.239)</td>
<td>0.50</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>43 hr before HD (0.244)</td>
<td>0.53</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>24 hr before HD (0.247)</td>
<td>0.44</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>29 hr before HD (0.228)</td>
<td>0.49</td>
</tr>
<tr>
<td>3</td>
<td>2</td>
<td>19 hr before HD (0.255)</td>
<td>0.33</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>43 hr before HD (0.235)</td>
<td>0.47</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>43 hr before HD (0.233)</td>
<td>0.52</td>
</tr>
<tr>
<td>3</td>
<td>3</td>
<td>24 hr before HD (0.276)</td>
<td>0.39</td>
</tr>
</tbody>
</table>

### Table 5.7: Timing of water stress detection using COV of TPCA baseline of 0.70 for the Third experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>COV of TPCA based detection prior to HD</th>
<th>COV of TPCA at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>-</td>
<td>2.44</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>-</td>
<td>1.13</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>24 hr before HD (0.799)</td>
<td>2.06</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>-</td>
<td>2.16</td>
</tr>
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<td>2</td>
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<td>-</td>
<td>1.51</td>
</tr>
<tr>
<td>3</td>
<td>2</td>
<td>-</td>
<td>0.67</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>-</td>
<td>2.73</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>-</td>
<td>1.48</td>
</tr>
<tr>
<td>3</td>
<td>3</td>
<td>-</td>
<td>1.12</td>
</tr>
</tbody>
</table>

Table 5.6: Timing of water stress detection using CWSI baseline of 0.20 for the Third experiment (HD: stress detection by human).

Table 5.7: Timing of water stress detection using COV of TPCA baseline of 0.70 for the Third experiment (HD: stress detection by human).
Table 5.8: Timing of water stress detection using CWSI baseline of 0.10 for the Fourth experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>CWSI based detection prior to HD</th>
<th>CWSI at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>24 hr before HD (0.111)</td>
<td>0.300</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>53 hr before HD (0.100)</td>
<td>0.434</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>77 hr before HD (0.143)</td>
<td>0.558</td>
</tr>
<tr>
<td>4</td>
<td>1</td>
<td>96 hr before HD (0.245)</td>
<td>0.482</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>48 hr before HD (0.136)</td>
<td>0.294</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>72 hr before HD (0.168)</td>
<td>0.458</td>
</tr>
<tr>
<td>3</td>
<td>2</td>
<td>96 hr before HD (0.114)</td>
<td>0.529</td>
</tr>
<tr>
<td>4</td>
<td>2</td>
<td>91 hr before HD (0.193)</td>
<td>0.568</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>72 hr before HD (0.197)</td>
<td>0.414</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>115 hr before HD (0.104)</td>
<td>0.467</td>
</tr>
<tr>
<td>3</td>
<td>3</td>
<td>96 hr before HD (0.184)</td>
<td>0.650</td>
</tr>
</tbody>
</table>

Table 5.9: Timing of water stress detection using COV of TPCA baseline of 0.36 for the Fourth experiment (HD: stress detection by human).

<table>
<thead>
<tr>
<th>Stress cycle</th>
<th>Plant No</th>
<th>COV of TPCA based detection prior to HD</th>
<th>COV of TPCA at HD</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>1</td>
<td>-</td>
<td>0.240</td>
</tr>
<tr>
<td>2</td>
<td>1</td>
<td>-</td>
<td>0.249</td>
</tr>
<tr>
<td>3</td>
<td>1</td>
<td>-</td>
<td>0.360</td>
</tr>
<tr>
<td>4</td>
<td>1</td>
<td>-</td>
<td>0.351</td>
</tr>
<tr>
<td>1</td>
<td>2</td>
<td>-</td>
<td>0.375</td>
</tr>
<tr>
<td>2</td>
<td>2</td>
<td>-</td>
<td>0.367</td>
</tr>
<tr>
<td>3</td>
<td>2</td>
<td>-</td>
<td>0.688</td>
</tr>
<tr>
<td>4</td>
<td>2</td>
<td>-</td>
<td>0.109</td>
</tr>
<tr>
<td>1</td>
<td>3</td>
<td>-</td>
<td>0.878</td>
</tr>
<tr>
<td>2</td>
<td>3</td>
<td>-</td>
<td>-</td>
</tr>
<tr>
<td>3</td>
<td>3</td>
<td>-</td>
<td>0.461</td>
</tr>
</tbody>
</table>
6.1. Conclusions

The goal of this dissertation was to establish a methodology for early, non-contact, and non-destructive water stress detection for plants grown under controlled environment. The proposed techniques were crop water stress index based on infrared thermometry and top projected canopy area change of the plants based on machine vision. To evaluate the proposed sensing techniques for early detection of water stress, a total of four experiments was conducted using New Guinea Impatiens as a model plant. The experiments were repeated three times under low humidity conditions and one experiment was conducted under a higher humidity condition with a constant radiant load. The results and conclusions are summarized based on the dissertation's objectives.

**Objective 1.** Designed and built an automated computer controlled continuous plant health and growth monitoring system. Conclusions were: The instrumentation of the system was able to measure plant leaf and air temperature, relative humidity, air velocity, light intensity, and plant images; the system provided necessary data evaluate
the proposed techniques; the operation and positional control of the turn-table was accurate.

**Objective 2.** Developed and tested a crop water stress index (CWSI) model for plants grown under controlled environment conditions using basic thermodynamics principles and the energy balance of the plant. Conclusions were: Model predicted CWSI values were correlated to measured CWSI values with $R^2$ values of 0.83, 0.50, 0.79, and 0.76.

**Conclusions drawn from experimental results:**

1. Leaf to air temperature differential was found to be inversely correlated with the measured ET rates. The leaf temperatures of well-watered plants were consistently lower than air temperature during the experiments. The leaf temperatures of the stressed plants were found to be 1-3 °C higher than the air temperature.

2. Crop water stress index was inversely and linearly correlated with measured evapotranspiration rates.

3. Average maximum crop water stress index values of three stressed plants were found to be 0.68, 0.75, 0.52, and 0.59 at the time of water stress detection by human for the first, second, third, and the fourth experiments, respectively. The CWSI values close to zero indicated that the plants were well-watered.

4. Water became less available to the plants at 30-35% volumetric soil moisture levels resulting in reduced evapotranspiration rates, increased leaf temperatures, and increased total canopy resistances. The soil water tension levels higher than 10 kPa caused crop water stress index to increase.
**Objective 3.** Determined top projected canopy area (TPCA) of the plants from plant images using machine vision and image processing applications. Instantaneous plant movement and the coefficient of variation of TPCA (COV of TPCA) were determined and analyzed.

**Conclusions drawn from the experimental results:**

1. Water stress temporarily inhibited TPCA expansion of the plants in the treatment group. The TPCA expansion resumed as the plants recovered from water stress condition following the irrigation.
2. The TPCA gains of the plants in the treatment group were found to be less than the TPCA gains of the plants in the control group due to the effect of water stress.
3. Visual observations showed that the motions of fully developed leaves mainly affected the change of TPCA.
4. The COV of TPCA was found to be more suitable parameter to use in TPCA based water stress detection compared to instantaneous movement (defined by equation 4.34), because it shows the stability of plant's movement.

**Objective 4.** Established baselines for early detection of the water stress with CWSI and COV of TPCA of the plants. The CWSI baselines were 0.14, 0.12, 0.20, and 0.10 while the COV of TPCA baselines were 0.40, 0.55, 0.70, and 0.36 for early water stress detection.
**Objective 5.** The effectiveness of the sensing techniques was evaluated by the time of detection prior to the human stress detection. The results suggested that CWSI based method was more successful and provided earlier detection of water stress compared to TPCA based method.
6.2. Recommendations for future research

1. The effectiveness of the sensing techniques under high humidity conditions must be further evaluated with repeated experiments with the same plants used in this experiment.

2. Dynamic responses of the plants, especially leaf temperature, to varying environmental conditions should be evaluated and the stability and the effectiveness early water stress detection using CWSI must be tested under greenhouse conditions with a broader range of environmental conditions.

3. The sensing techniques should be evaluated with high transpiration plants before generalizations are made to other plants. This study focused on a single crop, New Guinea Impatiens, with specific physiological characteristics of low transpirational capacity and high capacity for water storage.

4. Future research should study the effect of the night period on water stress development and the detection of water stress. This study evaluated only light time period data on the detection of water stress.

5. Destructive and contact measurements such as leaf conductance, sap flow measurements are recommended for more detailed comparison between the sensing techniques used in this study and the other methods of detecting plant water stress.

6. Automatic irrigation scheduling applications using CWSI with a feedback control system and its comparison to other scheduling methods is recommended.
7. Evaluations of the sensing techniques under wider ranges of environmental conditions and with different plants should be considered. Transfer of the water stress detection technology to commercial plant production settings should also be considered.

8. There is strong evidence in the literature that secondary metabolism of the plant is promoted as the plant experiences water stress. Therefore, it would be valuable to develop a water management method using CWSI to promote secondary metabolism in the plants for phytoceutical production.
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APPENDIX A

Soil moisture release curve experimental procedures and results
I. Procedure for water release curve experiment for pressures ≤ 0.5 bar (McCoy, 1999)

1. Place exco porous plastic in the bottom of the tempe-cell and replace the tempe-cell sleeve into the tempe-cell.

2. Weigh the empty tempe-cell including the lid and wing nuts. This gives the tare weight.

3. Take tempe-cell containing soil medium and place tempe-cell into its container.

4. Place the container (including tempe-cell) in a cup and saturate the medium with degassed water until the water level is at the top of the tempe-cell bottom.

5. Let the soil sample stay in the water overnight.

6. Attach plastic tubing to the tempe-cell and adjust manometer to 10 cm of water tension.

7. Wait for 3-4 days until soil sample equilibrate. Decision on the time of equilibrium is made when the weight change of the sample is equal or less than 0.05 grams per 8 hours.

8. Record the weight of the sample at 10 cm water tension, also record time and the date.

9. Repeat the steps #11-14 at 20cm, 40 cm, 60, and 80 cm on manometer setup; also at 100 cm, 200 cm, 300, and 500 cm on the pressure manifold setup.

10. After the steps #8 through #12 completed, remove the tempe-cell lid.

11. Dry the samples for 48 hours at 105 °C in the oven.

12. Weigh the dry sample including lid and wing nuts, then record the weigh and also time and the date.
II. Procedure for water release curve experiment for 2 bar pressure (McCoy, 1999)

1. Place two sheets of filter paper on the bottom of a copper sleeve with 2 layers of cheese cloth and a plastic rubber band.
2. Weight the empty sleeve including filter paper, cheese cloth, and the plastic rubber band. This gives the tare weight.
3. Fill the sleeve with soil and soak the samples in degassed water overnight.
4. Place the samples in the pressure plate extractors and place a moist paper towel on top of the samples
5. Screw-down the lid of the extractor firmly
6. Adjust the pressure to 2 bars.
7. Six to seven days later, dry the samples 48 hours at 105 °C and weigh each sleeve.
III. Psychrometer analysis for permanent wilting point (McCoy, 1999)

A. Preparation of standard solutions

1. Moisten the experimental unit box and turn-on the nanovoltmeter and let it warm-up

2. Prepare calibration samples
   - Make sure the soil cups are clean and dry
   - Weigh cups on the analytical balance.

3. Zero the nanovoltmeter.

4. Add calibration solutions to cups from 100, 500, 900, 1100, and 1500 mOsm/kg H₂O.
   Make sure that enough solution is put to wet the strip of the thermometer.

5. Add distilled water to the well, so meniscus is even with the top of the well.

6. Place cups in changer apparatus

7. Let the cups and changer equilibrate for 30 minutes

8. At equilibrium, start reading the microvolt and temperatures reading from the solutions.

9. Remove all cups except water well and 500 mOsm/Kg H₂O solution, it is used as a reference check throughout the experiment.
B. Preparation of soil samples

1. Take soil from 2 bar pressure chamber and make sure that the sample is kept in a petri dish to reduce evaporative water loss from the soil.

2. Place soil samples into the cups with a small spatula. The cups must be filled only half-full.

3. Place the cups into psychrometer sample changer.

4. Place the soil cups under the thermocouple and let the sample equilibrate and read the microvolt and temperature, also read from reference solution.

5. After all the soil samples are measured, take the soil cups and weigh them using the analytical balance.

6. Dry the sample quickly by damping the soil onto a wax paper. Drying time depend on the soil type.

7. Place the samples back to psychrometer and let them equilibrate for 30 min.

8. Repeat the procedure with soil samples and reference solution and record microvolts and temperature readings. The readings are repeated until a big number is reached from the micrometer such as 40 µm or up.

9. After psychrometer unit measurements are completed, oven dry the soil samples for 48 hours at 105 °C.

10. Perform the calculations to obtain soil moisture release curve for the experimental soil.
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<th>Sample ID</th>
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Table A.1: Tension and volumetric soil moisture content calculation results from soil moisture release curve experiment.
APPENDIX B

Theta probe calibration procedure and the results
I. Soil-specific calibration

The relationship between ThetaProbe output, Volts, and square root of dielectric constant, $\varepsilon^{1/2}$, is given by a 3$\textsuperscript{rd}$ order polynomial relationship as (ThetaProbe User Manual, 1998):

$$\sqrt{\varepsilon} = 1.07 + 6.4V - 6.4V^2 + 4.7V^3, R^2 = 0.99 \quad (C.1)$$

where, $V$ is the volt reading from the sensor in volts. It was provided that there is a simple linear relationship between square root of dielectric constant and volumetric soil moisture as:

$$\sqrt{\varepsilon} = a_o + a_1 \theta V \quad (C.2)$$

where, $a_o$ and $a_1$ two necessary coefficients of soil sample. In order to determine the relationship between sensors voltage reading and volumetric soil moisture following protocol was followed:

1. Collect samples of damp soil, distributing it as little as possible so that it is at the same density as in situ. For this, three sets of soil were prepared using the experimental soil medium. Three pots of soil were used at three different moisture level. Before the measurements, the soil was watered for one week in order to let the soil settle in the pot until the bulk density of the soil remained unchanged.
2. Insert the theta probe and measure the probe output as $V_w$. Then, use Eq. C.2. to calculate $\varepsilon_w^{1/2}$. Weigh the damp sample to record $W_w$ and measure its volume.

3. Oven dry the sample, insert the theta probe into dry soil ($\theta_v=0$), and measure the probe output as $V_0$. Weigh the dry sample to record $W_0$. Use Eq. C.2. to calculate $\varepsilon_o^{1/2}$. This equals $a_o$.

4. Calculate the volumetric water content $\theta_w$ of the original sample as:

$$\theta_w = \frac{W_w - W_o}{L} \quad \text{(C.3.)}$$

5. Then calculate $a_1$ using the following equation:

$$a_1 = \frac{\sqrt{\varepsilon_w} - \sqrt{\varepsilon_o}}{\theta_w} \quad \text{(C.4.)}$$

6. Then, the water content was determined from a calibrated theta probe as:

$$\theta_v = \frac{(1.1 + 4.44V) - a_o}{a_1} \quad \text{(C.5)}$$

In this calibration experiment, seven theta probes were calibrated and six of them was used for the experiments in this study. The results of the probe calibration are provided in the following tables.
### PROBE READINGS FROM MOIST SOIL

<table>
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<tr>
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<th>Reading 4</th>
<th>Reading 5</th>
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Wet Weight of Media and Container (A) 1054.89
Container Weight (A) 57.08
Wet Weight of Media (A) 997.81
Dry Weight of Media and Bag (A) 239.32
Bag Weight 9.49
Dry Weight of Media (A) 229.83

Soil Media Volume (m³): 0.0013

Table B.1: Theta probe readings from moist and dry soil sample A and weight measurements
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Wet Weight of Media and Container (AB)  571.85

Container Weight (B)  55.49

Wet Weight of Media (B)  516.36

Dry Weight of Media and Bag (B)  233.10

Bag Weight  9.55

Dry Weight of Media (B)  223.55

Table B.2: Theta probe readings from moist and dry soil sample B and weight measurements
### Table B.3: Theta probe readings from moist and dry soil sample C and weight measurements

#### PROBE READINGS FROM MOIST SOIL

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<tr>
<td>probe 5</td>
<td>0.216</td>
<td>0.230</td>
<td>0.245</td>
<td>0.296</td>
<td>0.195</td>
<td>0.236</td>
</tr>
<tr>
<td>probe 6</td>
<td>0.218</td>
<td>0.230</td>
<td>0.237</td>
<td>0.256</td>
<td>0.262</td>
<td>0.241</td>
</tr>
<tr>
<td>probe 7</td>
<td>0.209</td>
<td>0.200</td>
<td>0.242</td>
<td>0.202</td>
<td>0.172</td>
<td>0.205</td>
</tr>
</tbody>
</table>

#### PROBE READINGS FROM DRY SOIL

<table>
<thead>
<tr>
<th>Probe ID</th>
<th>Reading 1</th>
<th>Reading 2</th>
<th>Reading 3</th>
<th>Reading 4</th>
<th>Reading 5</th>
<th>Average</th>
</tr>
</thead>
<tbody>
<tr>
<td>probe 1</td>
<td>0.013</td>
<td>0.016</td>
<td>0.015</td>
<td>0.014</td>
<td>0.016</td>
<td>0.015</td>
</tr>
<tr>
<td>probe 2</td>
<td>0.017</td>
<td>0.019</td>
<td>0.018</td>
<td>0.016</td>
<td>0.018</td>
<td>0.018</td>
</tr>
<tr>
<td>probe 3</td>
<td>0.014</td>
<td>0.017</td>
<td>0.015</td>
<td>0.015</td>
<td>0.017</td>
<td>0.016</td>
</tr>
<tr>
<td>probe 4</td>
<td>0.017</td>
<td>0.019</td>
<td>0.018</td>
<td>0.017</td>
<td>0.019</td>
<td>0.018</td>
</tr>
<tr>
<td>probe 5</td>
<td>0.017</td>
<td>0.019</td>
<td>0.018</td>
<td>0.017</td>
<td>0.019</td>
<td>0.018</td>
</tr>
<tr>
<td>probe 6</td>
<td>0.016</td>
<td>0.019</td>
<td>0.017</td>
<td>0.016</td>
<td>0.018</td>
<td>0.017</td>
</tr>
<tr>
<td>probe 7</td>
<td>0.017</td>
<td>0.020</td>
<td>0.019</td>
<td>0.017</td>
<td>0.020</td>
<td>0.019</td>
</tr>
</tbody>
</table>

**Wet Weight of Media and Container (C)**: 571.85

**Container Weight (C)**: 55.49

**Wet Weight of Media (C)**: 516.36

**Dry Weight of Media and Bag (C)**: 233.10

**Bag Weight**: 9.55

**Dry Weight of Media (C)**: **223.55**
Table B.4: Calculated soil coefficients for theta probe calibration
APPENDIX C

Source codes and flow diagrams for continuous plant health monitoring system

A1.a. Flow chart for main module "System.CPP"
A1.b. Module "System.CPP" (Code was written in C and C++)

A2.a. Flow chart for sub-module "Move1.C"
A2.b. Module "Move1.C" (Code was written in C)

A3.a. Flow chart for sub-module "Home.C"
A3.b. Module "Home.C" (Code was written in C)
Start

Initialize variables

\( \text{NB} \_\text{GRAB} = 5 \)
\( i = 1 \)
\( k = 0 \)
\( \text{QVset} = 0.60 \)

Irrigation

while \( i = 1 \)

\( j = 0 \)

\( j = j + 1 \)

Call to subroutine "Home.c" to home the table

\( n = 0 \)

Grab image

Delay

\( n = n + 1 \)

is \( n < \text{NB} \_\text{GRAB} \)

A

Figure A1.a: Flow chart for main module "System.CPP." (continued)
Figure A1.a (continued).

Read sensors
Leaf temperature (IRTc)
Air temperature
Light intensity
Humidity

Display process on the monitor

Delay

if \( j = 1 \) Y:
save images into "plant1"

if \( j = 2 \) Y:
save images into "plant2"

if \( j = 3 \) Y:
save images into "plant3"

if \( j = 4 \) Y:
save images into "plant4"

if \( j = 5 \) Y:
save images into "plant5"

if \( j = 6 \) Y:
save images into "plant6"

(continued)
Figure A1.a (continued).

Show soil moisture reading on the monitor

(continued).
Call to subroutine "Move.c" to move the table to the next plant

Average soil moisture readings:
\[ A = \frac{\sum A_n}{6}, B = \frac{\sum B_n}{6} \]
\[ C = \frac{\sum C_n}{6}, D = \frac{\sum D_n}{6} \]
\[ E = \frac{\sum E_n}{6}, F = \frac{\sum F_n}{6} \]

Save average soil moisture readings into file

if \( j < 6 \)

Irrigation pump OFF

Irrigation pump ON

Delay pump for \( t \) (seconds) to be ON

Reset the variables:
\[ A = B = C = D = E = F = 0, A1 = B1 = C1 = D1 = E1 = F1 = 0 \]
\[ A2 = B2 = C2 + D2 = E2 = F2 = 0, A3 = B3 = C3 = D3 + E3 = F3 = 0 \]
\[ A4 = B4 = C4 = D4 = E4 = F4 = 0, A5 = B5 = C5 = D5 = E5 = F5 = 0 \]
\[ A6 = B6 = C6 = D6 = E6 = F6 = 0, k = j = \text{Irrigation} = 0 \]
A1.b Source Code for main module "System.CPP"

Module : SYSTEM.CPP
Purpose : MAIN MODULE FOR THE EXPERIMENT
(3 CONTROL, 3 STRESS PLANTS)
  1) Operate turn-table
  2) Data acquisition
     a) Acquire plant images and store it
     b) Read theta probe (soil moisture)
     c) Read IR/Tc, Radiation, Humidity

Programmer : Murat Kacira
Last update : 04/06/1999

*******************************************************************************/
#pragma hdrstop
#include <process.h>
#include <math.h>
#include <stdio.h>
#include <stdlib.h>
#include <conio.h>
#include <errno.h>
#include <dos.h>
#include <time.h>
#include <condefs.h>
#include "c:\ni-daq\include\nidaqex.h"
#include "c:\matrox\mil\include\mil.h"

/* Number of buffers in the sequence. */
#define NB_GRAB 5
/* Image scale. */
#define IMAGE_SCALE 0.5

/////////////////////////////////////////////////////////////////////////
/*This section provides necessary library files which includes definitions for functions
used to operate frame grabber board*/
USELIB("..\NI-DAQ\Lib\nidx32b.lib");
USELIB("..\NI-DAQ\Lib\nidaq32b.lib");
USELIB("..\matrox\mil\library\winnt\bc\dll\mil.lib");
USELIB("..\matrox\mil\library\winnt\bc\dll\milmet2.lib");
/////////////////////////////////////////////////////////////////////////

230
#pragma argsused
void main(int argc, char *argv[])
{
    FILE *stream;
    MIL_ID MilApplication;
    MIL_ID MilSystem;
    MIL_ID MilDigitizer;
    MIL_ID MilDisplay;
    MIL_ID MillImage[NB_GRAB];
    MIL_ID MillImageDisp;

    double Time = 0.0;
    double TimeWait = 0.0;
    long n;

    i16 iStatus = 0;
    i16 iRetVal = 0;
    i16 iDevice = 1;
    i16 iChanA = 0, iChanB = 1, iChanC = 2, iChanD = 3, iChanE = 4,
              iChanF = 5, iChanG = 6, iChanL = 15, iChanH = 14, iChanK = 7;
    i16 iChan1 = 1;
    i16 iInputModeA = 0, iInputModeB = 0, iInputModeC = 0, iInputModeD = 0,
              iInputModeE = 0, iInputModeF = 0, iInputModeG = 1, iInputModeH = 1,
              iInputModeK = 1, iInputModeL = 1;
    i16 iInputRangeA = 10, iInputRangeB = 10, iInputRangeC = 10,
              iInputRangeD = 10, iInputRangeE = 10, iInputRangeF = 10,
              iInputRangeG = 10, iInputRangeH = 10, iInputRangeK = 10, iInputRangeL = 10;
    i16 iPolarityA = 1, iPolarityB = 1, iPolarityC = 1, iPolarityD = 1,
              iPolarityE = 1, iPolarityF = 1, iPolarityG = 1, iPolarityH = 1,
              iPolarityK = 1, iPolarityL = 1;
    i16 iDriveAISA = 0, iDriveAISB = 0, iDriveAISC = 0, iDriveAISD = 0,
                        iDriveAISE = 0, iDriveAISF = 0, iDriveAISG = 0, iDriveAISH = 0,
                        iDriveAISK = 0, iDriveAISL = 0;
    f64 dVoltageA = 0.0, dVoltageB = 0.0, dVoltageC = 0.0, dVoltageD = 0.0,
               dVoltageE = 0.0, dVoltageF = 0.0, dVoltageG = 0.0, dVoltageH = 0.0,
               dVoltageK = 0.0, dVoltageL = 0.0;
    f64 dVoltage1 = 0.0;
    f64 dVoltage2 = 5.0;
    i16 iGainA = 10, iGainB = 10, iGainC = 10, iGainD = 10, iGainE = 10,
                iGainF = 10, iGainG = 100, iGainH = 20, iGainK = 100, iGainL = 2;
    i16 iIgnoreWarning = 0;
float QVsensorA, QVsetA, A, A1, A2, A3, A4, A5, A6,
QVsensorB, B, B1, B2, B3, B4, B5, B6,
QVsensorC, C, C1, C2, C3, C4, C5, C6,
QVsensorD, D, D1, D2, D3, D4, D5, D6,
QVsensorE, E, E1, E2, E3, E4, E5, E6,
QVsensorF, F, F1, F2, F3, F4, F5, F6,
lrg, IRTC, AIR, HUMID, RAD; /* Reading the devices*/

struct time t;
char f_name[10];

int i, j, k, m, Flow; /* "j" and "k" are the PLANT IDENTIFIERS*/

i=1;
QVsetA=0.70;
k=0;
Flow=0;

while (i= =1) {

printf("BRINGING THE TABLE TO HOME POSITION\n");
/*Home the table. "Home.EXE" is the child program*/
spawnv(P_WAIT, "Home.EXE", argv);

sleep(5);

for (j=1; j<7; j++) {

/*IMAGE ACQUISITION AND SAVE FOUR CONSECUTIVE IMAGES
IMAGES TO DIRECTORY*/
/*Allocations.*/
Mapp Alloc(M_DEFAULT, &MilApplication);
Msys Alloc(M_DEF_SYSTEM_TYPE, M_DEF_SYSTEM_NUM, M_SETUP, &MilSystem);
Mdig Alloc(MilSystem, M_DEFAULT, M_DEF_DIGITIZER_FORMAT, M_DEFAULT, &MilDigitizer);
Mdisp Alloc(MilSystem, M_DEFAULT, M_DEF_DISPLAY_FORMAT, M_DEFAULT, &MilDisplay);

for (n=0; n<NB_GRAB; n++) {
    Mbuf Alloc2d(MilSystem,(long)(Mdig Inquire(MilDigitizer, M_SIZE_X, M_NULL)*IMAGE_SCALE),(long)(Mdig Inquire(MilDigitizer, M_SIZE_Y, M_NULL)*IMAGE_SCALE), 8L+M_UNSIGNED, M_IMAGE+M_GRAB, &MilImage[n]);
}
MbufAlloc2d(MiISystem, (long)(MdigInquire(MilDigitizer, M_SIZE_X, M_NULL)*IMAGE_SCALE),
(long)(MdigInquire(MilDigitizer, M_SIZE_Y, M_NULL)*IMAGE_SCALE),
8L+M_UNSIGNED,
M_IMAGE+M_GRAB+M_PROC+M_DISP, &MillImageDisp);

/* Grab continuous on display at the specified scale. */
MdigControl(MilDigitizer, M_GRAB_SCALE, IMAGE_SCALE);
MdigGrabContinuous(MilDigitizer, MillImageDisp);

/* Halt continuous grab and put digitizer in asynchronous mode. */
MdigHalt(MilDigitizer);
MdigControl(MilDigitizer, M_GRAB_MODE, M_ASYNCHRONOUS);

/* Synchronize the timer and start the grab. */
MdigGrab(MilDigitizer, MillImage[0]);
MdigGrab(MilDigitizer, MillImage[0]);
MappTimer(M_TIMER_RESET, M_NULL);

/* Grab the sequence. */

for (n=0; n<NB_GRAB; n++){

gmtime(&t);

if (j==1) {
    sprintf(f_name, "c:\murat\cprog\plant1\Im%2d%2d%2d%d.bmp",
            t.ti_hour, t.ti_min, t.ti_sec, j);
}
if (j==2) {
    sprintf(f_name, "c:\murat\cprog\plant2\Im%2d%2d%2d%d.bmp",
            t.ti_hour, t.ti_min, t.ti_sec, j);
}
if (j==3) {
    sprintf(f_name, "c:\murat\cprog\plant3\Im%2d%2d%2d%d.bmp",
            t.ti_hour, t.ti_min, t.ti_sec, j);
}
if (j==4) {
    sprintf(f_name, "c:\murat\cprog\plant4\Im%2d%2d%2d%d.bmp",
            t.ti_hour, t.ti_min, t.ti_sec, j);
}
if (j == 5) {
    sprintf(f_name, "c:\murat\cprog\plant5\Im%2d%2d%2d%d.bmp",
            t.ti_hour,t.ti_min,t.ti_sec, j);
}
if (j == 6) {
    sprintf(f_name, "c:\murat\cprog\plant6\Im%2d%2d%2d%d.bmp",
            t.ti_hour,t.ti_min,t.ti_sec, j);
};

/* Grab one buffer at a time. */
MdigGrab(MilDigitizer, MilImage[n]);

if (n>0) {
    /*Save Image to file*/
    MbufExport(f_name, M_BMP, MilImage[n]);
}
sleep(2); /*There must be 2seconds of delay to write the image into folder.*/
}

/* Wait last grab end. */
MdigGrabWait(MilDigitizer, M_GRAB_END);
MappTimer(M_TIMER_READ, &Time);

/* Free allocations. */
MbufFree(MilImageDisp);
for (n=0; n<NB_GRAB; n++)
    { MbufFree(MilImage[n]);
    }
MdispFree(MilDisplay);
MdigFree(MilDigitizer);
MsysFree(MilSystem);
MappFree(MilApplication);
sleep(5);

/*READ THE THETA PROBE OUTPUT AND INFRARED SENSORS*/
iStatus = AI_Configure(iDevice, iChanA, iInputModeA, iInputRangeA, iPolarityA, iDriveAISA);
iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", ignoreWarning);
iStatus = AI_VRead(iDevice, iChanA, iGainA, &dVoltageA);
iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", ignoreWarning);
`iStatus = AI_Configure(iDevice, iChanB, iInputModeB, iInputRangeB, iPolarityB, iDriveAISB);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanB, iGainB, &dVoltageB);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanC, iInputModeC, iInputRangeC, iPolarityC, iDriveAISC);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanC, iGainC, &dVoltageC);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanD, iInputModeD, iInputRangeD, iPolarityD, iDriveAISD);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanD, iGainD, &dVoltageD);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanE, iInputModeE, iInputRangeE, iPolarityE, iDriveAISE);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanE, iGainE, &dVoltageE);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanF, iInputModeF, iInputRangeF, iPolarityF, iDriveAISF);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanF, iGainF, &dVoltageF);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanG, iInputModeG, iInputRangeG, iPolarityG, iDriveAISG);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanG, iGainG, &dVoltageG);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`

`iStatus = AI_Configure(iDevice, iChanH, iInputModeH, iInputRangeH, iPolarityH, iDriveAISH);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);`

`iStatus = AI_VRead(iDevice, iChanH, iGainH, &dVoltageH);`
`iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);`
iStatus = AI_Configure(iDevice, iChanK, iInputModeK, iInputRangeK, iPolarityK,
iDriveAISK);
iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);
iStatus = AI_VRead(iDevice, iChanK, iGainK, &dVoltageK);
iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);

iStatus = AI_Configure(iDevice, iChanL, iInputModeL, iInputRangeL, iPolarityL,
iDriveAISL);
iRetVal = NIDAQErrorHandler(iStatus, "AI_Configure", iIgnoreWarning);
iStatus = AI_VRead(iDevice, iChanL, iGainL, &dVoltageL);
iRetVal = NIDAQErrorHandler(iStatus, "AI_VRead", iIgnoreWarning);

printf("SAVING PLANT AND ENVIRONMENTAL DATA INTO THE

COMPUTER\n");
printf("REGISTERING PLANT ID# WITH ACQUIRED DATA\n");

IRTC = (dVoltageK*1000); /*Plant temperature*/
AIR = (dVoltageG*1000); /*Air temperature*/
HUMID = (dVoltageL*1000); /*Humidity*/
RAD = (dVoltageH*1000); /*Radiation*/

printf("IRTC: %0.4lf\n", IRTC);
printf("HUMID: %0.4lf\n", HUMID);
printf("AIR: %0.4lf\n", AIR);
printf("RAD: %0.4lf\n", RAD);

/*SAVE IRTC, AIR, HUMID, RAD to the file*/

gmtime(&t);

if (j==1) {
    k=6;
}
if (j==2) {
    k=j-1;
}
if (j==3) {
    k=j-1;
}
if (j==4) {
    k=j-1;
}
if (j == 5) {
    k = j - 1;
}
if (j == 6) {
    k = j - 1;
}

if (k == 6) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant6.FIL", "a+");
    fprintf(stream, "%.41f,%.41f,%.41f,%.41f,%s,%d\n",
            IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}
if (k == 1) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant1.FIL", "a+");
    fprintf(stream, "%.4lf,%.4lf,%.4lf,%.4lf,%s,%d\n",
            IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}
if (k == 2) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant2.FIL", "a+");
    fprintf(stream, "%.4lf,%.4lf,%.4lf,%.4lf,%s,%d\n",
            IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}
if (k == 3) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant3.FIL", "a+");
    fprintf(stream, "%.4lf,%.4lf,%.4lf,%.4lf,%s,%d\n",
            IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}
if (k == 4) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant4.FIL", "a+");
    fprintf(stream, "%.4lf,%.4lf,%.4lf,%.4lf,%s,%d\n",
            IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}
if (k == 5) {
    sprintf(f_name, "%2d%2d%2d", t.ti_hour, t.ti_min, t.ti_sec);
    stream = fopen("Plant5.FIL", "a+");
    fprintf(stream, "%.4lf,%.4lf,%.4lf,%.4lf,%s,%d
", 
        IRTC, AIR, HUMID, RAD, f_name, k);
    fclose(stream);
}

printf("READING SOIL MOISTURE AND SAVING THE DATA\n");
if (j == 1) {
    QVsensorA = (0.80*dVoltageA - 0.013);
    A1 = QVsensorA;
    QVsensorB = (0.83*dVoltageB - 0.013);
    B1 = QVsensorB;
    QVsensorC = (0.84*dVoltageC - 0.011);
    C1 = QVsensorC;
    QVsensorD = (0.85*dVoltageD - 0.013);
    D1 = QVsensorD;
    QVsensorE = (0.80*dVoltageE - 0.012);
    E1 = QVsensorE;
    QVsensorF = (0.79*dVoltageF - 0.012);
    F1 = QVsensorF;
    printf("The moisture is %.4lf\n", A1);
    printf("The moisture is %.4lf\n", B1);
    printf("The moisture is %.4lf\n", C1);
    printf("The moisture is %.4lf\n", D1);
    printf("The moisture is %.4lf\n", E1);
    printf("The moisture is %.4lf\n", F1);
}
if (j == 2) {
    QVsensorA = (0.80*dVoltageA - 0.013);
    A2 = QVsensorA;
    QVsensorB = (0.83*dVoltageB - 0.013);
    B2 = QVsensorB;
    QVsensorC = (0.84*dVoltageC - 0.011);
    C2 = QVsensorC;
    QVsensorD = (0.85*dVoltageD - 0.013);
    D2 = QVsensorD;
    QVsensorE = (0.80*dVoltageE - 0.012);
    E2 = QVsensorE;
    QVsensorF = (0.79*dVoltageF - 0.012);
    F2 = QVsensorF;
printf("The moisture is %.41f\n", Al);
printf("The moisture is %.41f\n", Bl);
printf("The moisture is %.41f\n", Cl);
printf("The moisture is %.41f\n", Dl);
printf("The moisture is %.41f\n", El);
printf("The moisture is %.41f\n", Fl);

if (j = =3) {
    QVsensorA = (0.80*dVoltageA - 0.013);
    A3=QVsensorA;
    QVsensorB = (0.83*dVoltageB - 0.013);
    B3=QVsensorB;
    QVsensorC = (0.84*dVoltageC - 0.011);
    C3=QVsensorC;
    QVsensorD = (0.85*dVoltageD - 0.013);
    D3=QVsensorD;
    QVsensorE = (0.80*dVoltageE - 0.012);
    E3=QVsensorE;
    QVsensorF = (0.79*dVoltageF - 0.012);
    F3=QVsensorF;

    printf("The moisture is %.41f\n", A1);
    printf("The moisture is %.41f\n", B1);
    printf("The moisture is %.41f\n", C1);
    printf("The moisture is %.41f\n", D1);
    printf("The moisture is %.41f\n", E1);
    printf("The moisture is %.41f\n", F1);

} if (j = =4) {
    QVsensorA = (0.80*dVoltageA - 0.013);
    A4=QVsensorA;
    QVsensorB = (0.83*dVoltageB - 0.013);
    B4=QVsensorB;
    QVsensorC = (0.84*dVoltageC - 0.011);
    C4=QVsensorC;
    QVsensorD = (0.85*dVoltageD - 0.013);
    D4=QVsensorD;
    QVsensorE = (0.80*dVoltageE - 0.012);
    E4=QVsensorE;
    QVsensorF = (0.79*dVoltageF - 0.012);
    F4=QVsensorF;

    printf("The moisture is %.41f\n", A1);
    printf("The moisture is %.41f\n", B1);
    printf("The moisture is %.41f\n", C1);
\begin{verbatim}
printf("The moisture is %.4lf\n", D1);
printf("The moisture is %.4lf\n", E1);
printf("The moisture is %.4lf\n", F1);

if (j = 5) {
    QVsensornA = (0.80*dVoltageA - 0.013);
    A5=QVsensornA;
    QVsensornB = (0.83*dVoltageB - 0.013);
    B5=QVsensornB;
    QVsensornC = (0.84*dVoltageC - 0.011);
    C5=QVsensornC;
    QVsensornD = (0.85*dVoltageD - 0.013);
    D5=QVsensornD;
    QVsensornE = (0.80*dVoltageE - 0.012);
    E5=QVsensornE;
    QVsensornF = (0.79*dVoltageF - 0.012);
    F5=QVsensornF;

    printf("The moisture is %.4lf\n", A1);
    printf("The moisture is %.4lf\n", B1);
    printf("The moisture is %.4lf\n", C1);
    printf("The moisture is %.4lf\n", D1);
    printf("The moisture is %.4lf\n", E1);
    printf("The moisture is %.4lf\n", F1);
}

if (j = 6) {
    QVsensornA = (0.80*dVoltageA - 0.013);
    A6=QVsensornA;
    QVsensornB = (0.83*dVoltageB - 0.013);
    B6=QVsensornB;
    QVsensornC = (0.84*dVoltageC - 0.011);
    C6=QVsensornC;
    QVsensornD = (0.85*dVoltageD - 0.013);
    D6=QVsensornD;
    QVsensornE = (0.80*dVoltageE - 0.012);
    E6=QVsensornE;
    QVsensornF = (0.79*dVoltageF - 0.012);
    F6=QVsensornF;

    printf("The moisture is %.4lf\n", A1);
    printf("The moisture is %.4lf\n", B1);
    printf("The moisture is %.4lf\n", C1);
    printf("The moisture is %.4lf\n", D1);
    printf("The moisture is %.4lf\n", E1);
    printf("The moisture is %.4lf\n", F1);
\end{verbatim}
if (j < 6) {
    /*Go to the jth plant (Module: Move1.c)*/
    spawnv(P_WAIT, "MoveLEXE", argv);
    sleep(5);
}
else;

/*AVERAGE THE READINGS*/
A = (A1+A2+A3+A4+A5+A6)/6;
B = (B1+B2+B3+B4+B5+B6)/6;
C = (C1+C2+C3+C4+C5+C6)/6;
D = (D1+D2+D3+D4+D5+D6)/6;
E = (E1+E2+E3+E4+E5+E6)/6;
F = (F1+F2+F3+F4+F5+F6)/6;
Irg = ((B+D+F)/3);

/*SAVE ***-MIN-AVERAGED THETA PROBE READING TO A FILE*/

gettime(&t);
sprintf(f_name, "%2d%2d%2d", t.ti_hour,t.ti_min,t.ti_sec);
stream = fopen("Testing2.FIL", "a+");

fprintf(stream, ".%.4lf,%.4lf,%.4lf,%.4lf,%.4lf,%.4lf,%s\n", A, B, C, D, E, F, Irg, 
f_name);
fclose(stream);

/*IRRIGATION BASED ON THETA PROBE READING*/

if ( Irg < QVsetA ) {
    /*iStatus = AO_VWrite(iDevice, iChan1, dVoltage2);
    iRetVal = NIDAQErrorHandler(iStatus, "AO_VWrite", ilgnoreWaming);*/

    printf("Pump should be 'ON' \n\n ");
    Flow=Flow+1;

    gettime(&t);
    sprintf(f_name, "%2d%2d%2d", t.ti_hour,t.ti_min,t.ti_sec);
    stream = fopen("FlowMet.FIL", "a+");
    fprintf(stream, "%d,%s\n", Flow, f_name);
    fclose(stream);

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/* Delay time for the pump to be ON */

/*RetVal = NIDAQDelay(480.0);
 iStatus = AO_VWrite(iDevice, iChan1, dVoltage1);
 sleep(10); */
}

else;

/* Second output voltage (dVoltage1)*/
if ( Irg >= QVsetA ) {
    /*RetVal = NIDAQErrorHandler(iStatus, "AO_VWrite", iIgnoreWarning);*/
    printf("Pump should be 'OFF' \n\n");
}
else;

/**********************************************************/
A=0, A1=0, A2=0, A3=0, A4=0, A5=0, A6=0;
B=0, B1=0, B2=0, B3=0, B4=0, B5=0, B6=0;
C=0, C1=0, C2=0, C3=0, C4=0, C5=0, C6=0;
D=0, D1=0, D2=0, D3=0, D4=0, D5=0, D6=0;
E=0, E1=0, E2=0, E3=0, E4=0, E5=0, E6=0;
F=0, F1=0, F2=0, F3=0, F4=0, F5=0, F6=0;
}
}
Define
Relative distance \( dX = 152.6 \)
Ratio = 7.9

Call calibration file

Load parameter file

Motor ON

Input
Target distance = \( dX \times \text{Ratio} \), Move type = 'R'
Step type = 'H', backlash = 0.005
Max. speed = 0.015, Min. speed = 0.02
Slope = 100

Move the table

Motor OFF

Stop

Figure A2.a: Flow chart for sub-module "Move1.C"
A2.b. Source code for sub-module "Move1.CPP"

Module : Move1.c
Purpoe : Module program to operate a stepper motor 60 degree forward.
Programmer : Murat Kacira
Last update : 04/06/1999

#include "c:\md2\include\md2qc2s.h"
#include "c:\murat\mc\include\stdio.h"
#include "c:\murat\mc\include\math.h"
#include "c:\murat\mc\include\stdlib.h"
#include "c:\murat\mc\include\time.h"
#include "c:\murat\mc\include\stddef.h"

#define RatioX 7.90 /* for stepping motor */

main()
{
    float dX;
    dX=152.6;

    /* Initialize variables */
    strcpy(MD2CalFile, "md2.cal");
    MD2Setup();

    /* Load parameters */
    strcpy(MD2ParFile, "md2.par");
    MD2ParLoad();

/* Turn on MD-2 systems and move the motor*/
MD2Motor = 3;
MD2On();

MD2Target[3]=dX*RatioX;
MD2MoveType = 'R';
MD2StepType = 'H';
MD2Backlash[3]=0.005;
    MD2MaxSpeed[3]=0.15;
    MD2MinSpeed[3]=0.02;
    MD2Slope[3]=100;
    
    MD2MinSpeed[3]=0.008;
    */

MD2MoveO();
/*Turn off MD-2 system*/
MD2Off();

}
Figure A3.a: Flow chart for sub-module "Home.C"
### A3.b. Source code for sub-module "Home.CPP"

```c
#include "c:\md2\include\md2qc2s.h"

main( )
{
    /* Initialize variables */
    strcpy(MD2CalFile, "md2.cal");
    MD2Setup();

    /* Load parameters */
    trcpsy(MD2ParFile, "md2.par");
    MD2ParLoad();

    /* Turn-ON MD-2 system */
    MD2Motor = 3;
    MD2On();
    MD2Backlash[3]=0.005;
    MD2MinSpeed[3]=0.2;

    /* Home the table */
    MD2Home();
    /* Turn-OFF MD-2 system */
    MD2Off();
}
```
APPENDIX D

Source code for processing plant images to obtain TPCA

(Source code was written in Visual Basic and C)
```c
#include "Camera.h"

int main() {
    // Open the camera
    Camera camera;
    if (!camera.open()) {
        // Handle error
        return -1;
    }

    // Process each frame
    for (int i = 0; i < size; i++) {
        // Get frame data
        Frame frame = camera.getFrame(i);

        // Process the frame
        processFrame(frame);
    }

    // Close the camera
    camera.close();
}

int processFrame(Frame frame) {
    // Process each pixel
    for (int i = 0; i < frame.width; i++) {
        for (int j = 0; j < frame.height; j++) {
            // Apply pixel operation
            frame.data[i * frame.height + j] = processPixel(frame.data[i * frame.height + j], frame, i, j);
        }
    }

    // Return the processed frame
    return frame;
}
```

APPENDIX E

Two sample T-test for evaluation of water stress effect on TPCA gains of the plants

(Minitab version 12 was used)
### Experiment 1

**Two Sample T-Test and Confidence Interval**

Two sample T for T1 vs C1

<table>
<thead>
<tr>
<th></th>
<th>N</th>
<th>Mean</th>
<th>StDev</th>
<th>SE Mean</th>
</tr>
</thead>
<tbody>
<tr>
<td>T1</td>
<td>3</td>
<td>8.70</td>
<td>2.76</td>
<td>1.6</td>
</tr>
<tr>
<td>C1</td>
<td>3</td>
<td>9.24</td>
<td>4.68</td>
<td>2.7</td>
</tr>
</tbody>
</table>

95% CI for μ T1 - μ C1: ( -10.5,  9.4)

T-Test μ T1 = μ C1 (vs not =): $T = -0.17$  $P = 0.87$  DF = 3

### Experiment 2

**Two Sample T-Test and Confidence Interval**

Two sample T for T2 vs C2

<table>
<thead>
<tr>
<th></th>
<th>N</th>
<th>Mean</th>
<th>StDev</th>
<th>SE Mean</th>
</tr>
</thead>
<tbody>
<tr>
<td>T2</td>
<td>3</td>
<td>18.66</td>
<td>3.65</td>
<td>2.1</td>
</tr>
<tr>
<td>C2</td>
<td>3</td>
<td>29.87</td>
<td>5.99</td>
<td>3.5</td>
</tr>
</tbody>
</table>

95% CI for μ T2 - μ C2: ( -24.1,  1.7)

T-Test μ T2 = μ C2 (vs not =): $T = -2.77$  $P = 0.070$  DF = 3

### Experiment 3

**Two Sample T-Test and Confidence Interval**

Two sample T for T3 vs C3

<table>
<thead>
<tr>
<th></th>
<th>N</th>
<th>Mean</th>
<th>StDev</th>
<th>SE Mean</th>
</tr>
</thead>
<tbody>
<tr>
<td>T3</td>
<td>3</td>
<td>34.82</td>
<td>1.07</td>
<td>0.62</td>
</tr>
<tr>
<td>C3</td>
<td>3</td>
<td>42.63</td>
<td>2.26</td>
<td>1.3</td>
</tr>
</tbody>
</table>

95% CI for μ T3 - μ C3: ( -14.02,  -1.6)

T-Test μ T3 = μ C3 (vs not =): $T = -5.40$  $P = 0.033$  DF = 2

### Experiment 4

**Two Sample T-Test and Confidence Interval**

Two sample T for T4 vs C4

<table>
<thead>
<tr>
<th></th>
<th>N</th>
<th>Mean</th>
<th>StDev</th>
<th>SE Mean</th>
</tr>
</thead>
<tbody>
<tr>
<td>T4</td>
<td>3</td>
<td>19.80</td>
<td>1.47</td>
<td>0.85</td>
</tr>
<tr>
<td>C4</td>
<td>3</td>
<td>41.62</td>
<td>7.89</td>
<td>4.6</td>
</tr>
</tbody>
</table>

95% CI for μ T4 - μ C4: ( -41.75,  -1.9)

T-Test μ T4 = μ C4 (vs not =): $T = -4.71$  $P = 0.042$  DF = 2
APPENDIX F

Normality test results for crop water stress index (CWSI) and coefficient of variation of TPCA (COV of TPCA)

1) Minitab version 12.0 was used.
2) Ryan-Joiner (correlation based) and Kolmogorov-Smirnov (chi-square based) normality tests were used.
Figure F.1: Normality test results for a) crop water stress index (CWSI) and b) coefficient of variation of TPCA (COV of TPCA) of well-watered plants for the First experiment.
Figure F.2: Normality test results for a) crop water stress index (CWSI) and b) coefficient of variation of TPCA (COV of TPCA) of well-watered plants for the Second experiment.
Figure F.3: Normality test results for a) crop water stress index (CWSI) and b) coefficient of variation of TPCA (COV of TPCA) of well-watered plants for the Third experiment.
Figure F.4: Normality test results for a) crop water stress index (CWSI) and b) coefficient of variation of TPCA (COV of TPCA) of well-watered plants for the Fourth experiment.