The Effect of Peeling and Boiling Process on Nutrition and Health-Promoting Compounds of Root Vegetables Harvested at Mjindi area Located in Jozini, KwaZulu Natal, South Africa

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Received Date: 16 December, 2020; Accepted Date: 5 January, 2021; Published Date: 11 January, 2021

Abstract

The effect of peeling and boiling process was investigated on nutrients and health-promoting compounds of amadumbe (Colocasia esculenta), carrots (Daucus carota), sweet potatoes (Ipomoea batatas) and potatoes (Solanum tuberosum). The results indicated that peeling and boiling led to significant reduction of nutrients (total soluble starch, total soluble protein, crude fibre) and health-promoting compounds (flavonols, anthocyanin and phenolic acid). Peeling effects on nutrient content did not yield similar outcome (increase and decrease). Carrots showed insignificant loss of total soluble starch after peeling and major loss was observed after boiling. Amadumbe shows substantial loss of fibre after peeling, peels yielding 42.38% and edible part yielding 8.31% compared to all of the analysed vegetables. Health-promoting compounds were reduced significantly peeling and boiling process. The nutrient and phytochemical compounds in the peels of the various vegetables depends on method of peeling. Fourier-Transform Infrared Spectroscopy (FT-IR) bands evidenced the presence of nutrients and health-promoting compounds in studied vegetables.

Keywords: Boiling; Flavonols; Health promoting compounds; Nutrients; Peeling; Phenolic acid

Introduction

South Africa possesses a huge diversity of root vegetables crops which are grown under various weather conditions (Allemann et al. 2004) [1]. They are underground plant parts consumed by humans as food. Amadumbe are tuber crops produced as underground corm, potatoes are produced as stem tubers, sweet potatoes and carrots are produced as storage roots (Chandrasekara and Josheph Kumar 2016) [2]. They provide affordable sources of nutritional energy in the form of carbohydrates, fibre and protein (Chandrasekara and Josheph Kumar 2016) [2] and natural pigments with pharmacological relevance and therapeutic properties capable of reducing chronic diseases incidence known as phytochemicals (Petropoulos et al. 2019) [3]. Phytochemicals are bioactive compounds that exert health beneficial effects because they combat oxidative stress in the body by maintaining a balance between oxidants and antioxidants. Their biological activity occurs through the donation of electrons or hydrogen atoms from its hydroxyl to free radicals (Sreeramulu and Raghnath 2010) [4].

The root vegetables production within rural farming communities is on small scale and is mainly for subsistence purposes. These crops undergo domestic processing before consumption, including: washing, peeling, chopping and boiling (Tiwari and Cummins 2013) [5]. Understanding the effect of peeling and boiling on the nutrients and health-promoting compounds will assist the consumer to elude the losses and thus retaining the quality of vegetables (Cavagnaro and Galmarini 2012; dos Santos Siqueira et al. 2013; dos Reis et al. 2015) [6, 7, 8]. If nutrition content and health-promoting compounds intakes are calculated based on raw crops, the intake content may not be accurate after peeling and boiling process. Boiling procedure was conducted in this study based on the dietary habit in African society. Generally, vegetables are prepared at home on the basis of convenience and taste. Therefore, the aim of the present study was to assess the nutrition and health-promoting compounds content in raw root vegetables and assess losses after peeling and boiling.

Materials and Methods

Chemicals and Materials

All chemicals and solvents used were of analytical grade. Deionised water was used for preparing the solutions and
The alpha-amylase and amyloglucosidase were purchased from Sigma-Aldrich (St. Louis, MO). Polyethylene bags were purchased from Prestige (KwaZulu Natal, South Africa).

**Methods**

**Sample collection and pre-treatment**

Root vegetables samples amadumbe, carrots, sweet potatoes were harvested in Makhathini research station located in Mjindi situated in Jozini in KwaZulu Natal, South Africa. Based on Global Positioning System (GPS), the exact geographical location is represented by co-ordinates (27° 24' 37S; 32° 9' 2E). Samples were stored in polyethylene bags for transport. The boiled and raw of peels and edible part were prepared. In order to prepare the boiled samples, the peels and edible part of vegetables were soaked in water at 1:1.5 sample mass per water volume ratio and boiled for 15 minutes. Both raw and boiled samples were dried using a freeze dryer with the ice condenser set at -80°C and vacuum set at 1 mbar for 3 days and kept at 4°C until analysis were conducted.

**Nutrition composition**

**Total soluble protein determination**

The LECO Truspec Nitrogen Analyser (LECO Corporation, Michigan, USA) was employed to measure the content of protein in the samples using Official Analytical Chemists (AOAC) official method 990.03, (Chemists 2005) [9]. Triplicate samples were measured and placed into a combustion chamber at 950°C with an autosampler and the percentage of protein was calculated using equation (1).

\[
% \text{crude protein} = \% N \times 6.25 \quad (1/0.16 = 6.25)
\]  

**Total soluble starch determination**

The starch content was determined by weighing 1 g (d.b) of the sample into a test tube. Thereafter, 5 mL of 80% ethanol was added and the dispersion was vortexed and then incubated at 80°C for until all the ethanol completely evaporated. A 10 mL of acetate buffer was added into the test tube followed by 200 μL of alpha-amylase enzyme. The dispersion was vortexed and incubated for 30 minutes at 90°C, and brought to room temperature. After cooling, 200 μL of glucoamylase enzyme was added to the dispersion in the test tube and gently shaken. The test tube with the mixture was incubated at 60°C for 8 hours. The sample was made up to 200 mL volumetric flask using deionized water and filtered through Whatman filter paper no 1. A 5 mL of copper reagent was added into 3 mL of the filtrate in the test tube followed by addition of the arsénomolybdate reagent (5 mL). Copper reagent was prepared by dissolving 45 g sodium sulphate (anhydrous) and 5 g copper (II) sulphate 5-hydrate in deionized water and made up to 250 mL volumetric flask. Arsénomolybdate reagent was prepared by dissolving 10 g ammonium molybdate 4-hydrate, 1.2 g sodium arsenate 7-hydrate and carefully added concentrated 8.4 mL sulphuric acid in deionized water and made up to 200 mL volumetric flask. The test tube was shaken and allowed to stand for 90 minutes. The starch content of the sample was determined by UV absorption at 750 nm wavelength and the starch was calculated using equation (2) (Rasmussen et al. 1990) [10].

\[
% \text{Starch} = \frac{0.4555 \times \text{Absorbance of sample} \times 0.9}{\text{sample mass} \times \text{Absorbance of glucose standard}} \quad (2)
\]

**Crude fibre determination**

The fibre was determined as Neutral Detergent Fibre (NDF) using the Van Soest method (Van Soest et al. 1991) [11]. The sample (0.5 g d.b) was added into a 100 mL scintilled glass crucible. The marble/buffer beads and 50 mL of neutral detergent solution (NDS) (50 mL) were added to the glass crucible holder. The NDS was prepared with 124 g ethylene diamine tetra-acetic acid, 45.3 g disodium tetraborate, 200 g sodium lauryl sulphate, 67 mL 2-ethoxy ethanol and 30.4 g disodium hydrogen phosphate. The NDS was adjusted to pH 7 using 1 N HCl or 1 N NaOH. The crucible containing the sample was placed into the glass crucible holder which was thereafter placed into a digestive block set at 110°C. A 1 mL of alpha-amylase enzyme was then added and covered with stoppers for 70 minutes. After the glass crucible was removed and placed on a draining rack. The filtration unit connected to the vacuum system was used for drying the samples and washed three times with boiling water. The sample and sides of the crucible were then rinsed with acetone and the samples were placed in a drying oven at 105°C for 4 hours. The samples were then cooled in a desiccator, the crucible was weighed and the NDF of the sample was calculated using equation (3).

\[
% \text{NDF} = \frac{\text{(crude+dry residue)} - \text{(crude+ash)}}{\text{sample mass}} \times 100 \quad (3)
\]

**Crude pigment extraction**

A sample was mixed with (95% v/v) ethanol in a 1:1 ratio and placed in a water bath at 55°C for 30 minutes. The resulting ethanolic extract was filtered directly into glass Petri plates using Whatman filter paper no 1. The petri plates were placed in an incubation chamber to allow ethanol evaporation at room temperature. After drying, the crude pigments extract were collected into a dark-brown glass vial, and kept at 4°C until used (Favaro et al. 2018) [12].

**Antioxidants extraction**

About 30 mg (d.b) of samples was weighed and transferred into a 15 mL falcon tube and 400 μL distilled water was added. The samples were boiled in a water bath at 100°C for 30 minutes. 2 mL of extraction buffer (pH 7.0) was added to each sample. The 100 mL buffer was made by mixing 2 mL distilled water, 94.8 mL of 95% ethanol and 3.2 mL of 12 N HCl (pH 7). The sample solutions were vortexed and left overnight in agitation. The samples were centrifuged at 20800 × g for 15 minutes and the supernatants were collected. The 1 mL extraction buffer was added to each sample pellet; the samples were vortexed and left in agitation for 2 hours. The samples were centrifuged at 20800 × g for 15 minutes and the supernatant was collected and mixed with the first one. The whole amount of supernatant collected from each sample was centrifuged again at 20800 × g for 30 minutes before reading. The absorbance was measured spectrophotometrically (Cary 50, Germany) at 530 nm, at 350 nm and 280 nm respectively for anthocyanins, flavonols and...
phenolic acids, using the extraction buffer as blank. The anthocyanin content was calculated as cyanidin 3-glucoside equivalents [molar extinction coefficient (ε 26,900 Lm⁻¹ mol⁻¹, MW 484.82), the amounts of flavonols and phenolic acids were calculated as quercetin 3-glucoside (ε 21,877 Lm⁻¹ mol⁻¹, M.W. 464.38) and ferulic acid (ε 14,700 Lm⁻¹ mol⁻¹, MW 194.18) equivalents (Lago et al. 2015) [13].

**FT-IR analysis**

Structural analysis of the functional groups was performed using a Perkin Elmer 537 FT-IR spectrophotometer. The crude pigment extract was placed on the sample holder and scanned from 4000-400 cm⁻¹ with 2 cm⁻¹ of resolution by 32 average scans. The spectra were recorded using an attenuated total reflectance (ATR) disc and absorption frequencies were expressed as cm⁻¹.

**Statistical analysis**

The Statistical Package for Social Science (SPSS version 25.0 SPSS Inc, Chicago, IL, USA) was used for the analysis of nutrition data. The mean values and standard deviations of the four root vegetables samples were calculated for all replicate measurements. The significant differences in nutritional composition across root vegetables varieties were determined using Kruskal Wallis non-parametric test. Upon the identification of the significant difference, The Mann-Whitney U test was employed to determine the specific differences. Significance was measured at the 5% level throughout.

**Results and discussion**

**Raw samples analysis**

Nutrition and non-nutrition composition in all vegetables were not significantly different. However, vegetable peels showed highest content in most parameters compared to the edible part except protein on carrots and starch in amadumbe, potatoes and sweet potatoes (Table 1). All vegetables showed high amounts of starch except carrots where peels yielded 0.73% and edible part yielded 0.59% due to its non-starchy properties (Hongu, Suzuki and Wilson 2015) [14]. Potato peels had slightly high content of protein compared to other vegetables. Amadumbe peels have high fibre content of 42.38% due to its fibrous rough brown skin nature (Mokehe 2018) [15]. However, peeling process is required to decrease useless fibrous of vegetables (Thane and Reddy 1997) [16]. Anthocyanin content was the lowest in amadumbe peels (8.56 ppm) and highest in carrot peels (17.21 mg/g). Also, edible part of carrots (9.55 mg/g) had high anthocyanin content, while sweet potatoes had low (4.04 mg/g) content, however, there was no significant difference in all anthocyanin content (P < 0.05) in all vegetables. This could be due to anthocyanins being one of the major antioxidant pigments found in carrots (da Silva Dias 2014; Yoo et al. 2020) [17, 18]. Flavonols content for carrots peels (383.06 mg/g) was highest while amadumbe peels lowest content (117.83 mg/g). However, edible part of potatoes (161.85 mg/g) showed high flavonols content while sweet potatoes had low content (48.31 mg/g), there was no significant difference in all flavonols content (P < 0.05) in all vegetables. Lee et al. (2016) [19], reported 161.33 mg/g flavonols on raw potatoes edible part and this is in agreement with the findings of this study.

The highest amounts of phenolic acid compounds were found in potatoe peels and edible part 387.0 and 330.60 mg/g, respectively. There was no significant difference in all phenolic acid content (P < 0.05) in all vegetables. This is due to genetic ability of this crop to accumulate phenolic acids (Gugala et al. 2017; Keutgen et al. 2019) [20, 21]. Tarwadi and Agte (2005) [22] reported an average of 251.4±107.3 mg/g phenolic acid content of uncooked root vegetable 251.4±107.3 mg/g.

**Quality control**

Maize flour CRM (FCNC21-AFE16) form Fera Science proficiency testing Ltd was analysed for quality assurance. The certified value was compared with the obtained value (Table 1). Certified value compared well with the measured values. This indicates that the methods used to measure these parameters is valid.

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Maize flour CRM</th>
<th>Carrots</th>
<th>Amadumbe</th>
<th>Sweet potatoes</th>
<th>Potatoes</th>
<th>p-value</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>Measure d value</td>
<td>CRM value</td>
<td>peels</td>
<td>edible</td>
<td>peels</td>
<td>edible</td>
</tr>
<tr>
<td>Starch (%)</td>
<td>60.32±0.52</td>
<td>61.42</td>
<td>0.73±0.72</td>
<td>0.59±0.08</td>
<td>55.75±0.91</td>
<td>63.13±0.62</td>
</tr>
<tr>
<td>Protein (%)</td>
<td>8.82±0.06</td>
<td>8.42</td>
<td>8.89±0.23</td>
<td>10.63±0.23</td>
<td>10.32±0.65</td>
<td>8.83±0.32</td>
</tr>
<tr>
<td>Fibre (%)</td>
<td>25.27±0.61</td>
<td>25.72</td>
<td>32.12±0.53</td>
<td>21.65±0.18</td>
<td>42.38±0.54</td>
<td>8.31±0.03</td>
</tr>
<tr>
<td>Anthocyanin (mg/g)</td>
<td>4.24±0.32</td>
<td>3.0</td>
<td>17.21±0.22</td>
<td>9.55±0.76</td>
<td>8.56±0.09</td>
<td>5.32±0.08</td>
</tr>
</tbody>
</table>
Flavonol (mg/g) 63.15±0.12 66.0 383.06±0.78 135.27±0.89 117.83±0.19 76.35±0.19 153.25±0.13 48.3±0.31 275.56±0.63 161.85±0.76 0.021

Phenolic acid (mg/g) 112.41±0.65 113.0 285.30±0.5 295.4±0.53 315.99±0.32 209.0±0.34 289.14±0.94 132.4±0.41 387.0±0.63 330.60±0.34 0.041

Table 1: Analysis of raw skin and edible parts of amadumbe, sweet potatoes, carrots and potatoes

*mean±standard deviation. Kruskal Wallis test; Values in bold indicate P < 0.05

Boiled samples analysis

For improvement of palatability and digestibility vegetable require processing, however, domestic processing changes texture, flavor, appearance, nutrition and health-promoting compounds effect (Azarnia et al. 2011) [23]. Boiling process degraded vegetables quality due to high temperature application that damages the cell structure of the crops (Fabbrini and Crosby 2016) [24]. Starch was not detected in carrots peels and edible parts due to boiling and most of starch loss was observed in vegetables peels other than edible parts (Table 2). This is a positive finding since the peels are not usually consumed by humans. Boiling had minimal effect on the potatoes peels with protein content of 10.92% while carrots were significantly reduced to 6.11%, there was no significant difference in all protein content (P < 0.005). Anthocyanin content after boiling was the lowest in potatoes peels (2.28 mg/g) and highest in carrot peels (16.42 mg/g). Also, edible part of carrots (8.45 mg/g) had high anthocyanin content, while sweet potatoes had lowest (3.45 mg/g) content, there was no significant difference in all anthocyanin content (P < 0.05) in all vegetables. It was observed that carrots have high anthocyanin content on raw and boiled samples when compared to other vegetables in this study. Flavonol content for carrots peels (277.52 mg/g) was highest while amadumbe peels lowest content (58.79 mg/g). However, edible part of carrots (102.43 mg/g) showed high flavonol content while amadumbe had low content (27.76 mg/g), there was no significant difference in all flavonol content (P < 0.05) in all vegetables. Phenolic acid content after boiling was the lowest in potatoes peels (97.57 mg/g) and highest in carrot peels (209.01 mg/g). Also, edible part of potatoes (286.75 mg/g) had high phenolic acid content, while amadumbe had lowest (97.57 mg/g) content, there was no significant difference in all phenolic acid content (P < 0.05) in all vegetables. Nutrients and health promoting compounds instability during cooking and water soluble characteristic caused leaching into the boiling water (Alvi et al. 2003) [25]. (Biezanowska-Kopeć et al. 2016) [26] reported up to 60% loss of polyphenols in carrots due to peeling and boiling. Boiling promoted profound chemical composition change in vegetables, affecting bioavailability and content of chemopreventive compounds (antioxidants) (Yuan et al. 2009) [27]. Boiling procedure was conducted in this study based on the dietary habit in African society. Generally, vegetables are prepared at home on the basis of convenience and taste. (Yuan et al. 2009) [27].

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Carrots</th>
<th>Amadumbe</th>
<th>Sweet potatoes</th>
<th>Potatoes</th>
<th>P-value</th>
</tr>
</thead>
<tbody>
<tr>
<td>skin</td>
<td>27.78±0.27</td>
<td>43.85±0.52</td>
<td>28.84±0.41</td>
<td>45.05±0.63</td>
<td>52.07±0.25</td>
</tr>
<tr>
<td>edible</td>
<td>6.11±0.05</td>
<td>5.43±0.23</td>
<td>6.85±0.09</td>
<td>7.62±0.12</td>
<td>6.74±0.21</td>
</tr>
<tr>
<td>skin</td>
<td>6.11±0.05</td>
<td>5.43±0.23</td>
<td>6.85±0.09</td>
<td>7.62±0.12</td>
<td>6.74±0.21</td>
</tr>
<tr>
<td>edible</td>
<td>6.11±0.05</td>
<td>5.43±0.23</td>
<td>6.85±0.09</td>
<td>7.62±0.12</td>
<td>6.74±0.21</td>
</tr>
<tr>
<td>skin</td>
<td>16.42±0.45</td>
<td>8.45±0.76</td>
<td>5.33±0.56</td>
<td>3.60±0.92</td>
<td>11.42±0.11</td>
</tr>
<tr>
<td>edible</td>
<td>16.42±0.45</td>
<td>8.45±0.76</td>
<td>5.33±0.56</td>
<td>3.60±0.92</td>
<td>11.42±0.11</td>
</tr>
<tr>
<td>skin</td>
<td>277.52±0.75</td>
<td>102.43±0.25</td>
<td>58.79±0.43</td>
<td>27.76±0.56</td>
<td>94.61±0.43</td>
</tr>
<tr>
<td>edible</td>
<td>277.52±0.75</td>
<td>102.43±0.25</td>
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<td>94.61±0.43</td>
</tr>
<tr>
<td>skin</td>
<td>209.01±0.98</td>
<td>153.26±0.67</td>
<td>184.89±0.84</td>
<td>97.57±0.72</td>
<td>194.31±0.45</td>
</tr>
<tr>
<td>edible</td>
<td>209.01±0.98</td>
<td>153.26±0.67</td>
<td>184.89±0.84</td>
<td>97.57±0.72</td>
<td>194.31±0.45</td>
</tr>
</tbody>
</table>

Table 2: Analysis of boiled peels and edible parts of amadumbe, sweet potatoes, carrots and potatoes

*mean±standard deviation. Kruskal Wallis test; Values in bold indicate P < 0.05

FT-IR scans

FT-IR scans of the raw and boiled vegetables were studied. A decrease in the intensity of the boiled vegetables was observed in this study (data not shown). The bands in the fingerprint region between 1800 and 750 cm⁻¹ indicate the chemical composition of polyphenol, fats, proteins, polysaccharides and carbohydrates (Figure 1). The peak at 516 cm⁻¹ is due to CH ring deformation (Andrus 2006; Movasaghi et al. 2008) [28, 29], which indicate information about the polyphenols structure constituted of CH ring in the

Citation: Zondo SG, Qwabe F, Mahlambi P (2020) The Effect of Peeling and Boiling Process on Nutrition and Health-Promoting Compounds of Root Vegetables Harvested at Mjindi area Located in Jozini, KwaZulu Natal, South Africa. Jou Food Scien & Nutri: JFSN-114.DOI: 10.46715/jfsn2020.01.1000114
molecules (Schulz and Baranska 2007) [30]. The absorption bands in the 985–1028 cm\(^{-1}\) region indicate the vibration frequency of \(\text{CH}_2\text{OH}\) groups of carbohydrates: the C–O stretching vibration coupled with C–O bending of the C–OH groups, C–O, C–C stretching, and C–O–H deformation motions (Andrus 2006; Schulz and Baranska 2007) [28, 30]. The band at 1273 cm\(^{-1}\) was allocated to the stretching in-plane C–O vibration combined with the ring structure of phenyl, while the wavenumber of 1380 cm\(^{-1}\) assigned to the stretching C–H and deformation C–O (Schulz and Baranska 2007; Lu et al. 2011) [30, 31]. The distinctive band at 1625 cm\(^{-1}\) was indicate the ring C–C stretch of phenyl. Lu et al. (2011) [19] reported this distinctive absorption peak in the garlic qualitative study of polyphenolic components. The absorption peak at 2937 shows CH\(_2\) antisymmetric stretch of methyl groups mainly from lipids (Naumann 2001; Movasaghi, Rehman and ur Rehman 2008) [29, 32]. The distinctive band at 3260 cm\(^{-1}\) indicate O–H stretching of carbohydrates as well as the vibrational frequency in aromatic ring of alcohol and phenol (Andrus 2006; Movasaghi et al. 2008) [28, 29].

![Figure1](image_url)

**Figure1:** FT-IR scans of crude pigments extract from amadumbe, sweet potatoes, carrots and potatoes.

**Conclusion**

This study showed that vegetables peels prevent high absorption rate of the edible part. The loss of nutrients and health-promoting compounds was observed in this study and this was observed in literature as well. These observations will educate rural people on the effect of these inevitable treatments of vegetables and their consequence on nutrients and health promoting compounds heavy losses. Consumption of washed unpeeled raw vegetables is highly recommended however, steaming process may be employed for quick digestion and palatability purposes. For future studies, boiling, steaming and microwave process effects on vegetables quality will be studied. FT-IR bands showed the availability of nutrients and health promoting compounds in raw and boiled vegetables.

**References**


