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A Preliminary and Comparative Study on the Valorization of Marang (*Artocarpus odoratissimus* Blanco) Fruit Waste Through Enzyme-Assisted Pectin Extraction

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ABSTRACT

Agro-industrial fruit by-products are a potential source of functional ingredients that are currently underexploited, yet sustainable. Peel and core residues are commonly generated in large amounts by tropical fruits, such as the Marang (*Artocarpus odoratissimus* Blanco), which is common in the Philippines, and can be utilized rather than discarded as sources of beneficial polysaccharides, such as pectin. The primary objective of this research was the valorization of marang fruit waste through enzyme-assisted extraction (EAE), followed by the characterization of marang by-products pectin. Three commercial enzymes (cellulase, xylanase, and α-amylase) were used to extract pectin from the peel and core. The extracted samples were tested for yield (%), moisture content (MC %), ash content (AC%), equivalent weight (EW), methoxyl content (MeO), anhydrouronic acid (AUA), and degree of esterification (DE). Functional properties, including water-holding capacity (WHC), oil-holding capacity (OHC), swelling capacity (SC), emulsion capacity (EC), and emulsion stability (ES), were determined. Solubility tests and multivariate analyses (PCA and Pearson correlation) were used to establish relationships among structural and functional attributes. Pectin % yield ranged from 3.06% to 7.05%, with core-cellulase (CC) extraction producing the highest yield. All samples were classified as LM (low-methoxy) pectin (MeO) < 7%; DE < 50%), which can be used in the formation of gels induced by calcium. The core-derived pectin showed high WHC and SC, indicating strong hydration properties, whereas peel-derived pectin extracted with xylanase showed high EC and ES, suggesting potential emulsifying properties. Extraction with α-amylase increased pectin purity by hydrolyzing starch impurities, resulting in high EW and AUA values. Marang peel and core are also promising alternative sources of LM pectin, with various functional properties applicable to food, nutraceutical, and industrial applications. Such a valorization strategy will help reduce waste, advance the circular economy, and develop sustainable functional ingredients.

1 | Introduction

Agricultural, food, and industrial waste are generated at tremendous rates daily, posing significant challenges for their management and disposal. The fruit and vegetable (F&V) industry is among the food sectors that generate the most waste. This covers a large proportion of inedible ingredients, including

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skins, seeds, cores, peels, and pomace, which are usually discarded during production and processing (Ajila et al. 2009). Crop residues from rice, corn, coconuts, and sugarcane are significant in the Philippine agricultural sector, and rice straw alone yields about 17.6 million tons per year. Fruit peels contribute 15%–60% of all processing waste (Nirmal et al. 2023), including significant portions of mangoes, oranges, pineapples, and bananas, from 20% to 50% of the total processing waste per annum. The total amount of fruit waste is estimated at 25–57 million tons annually (Leong and Chang 2022). If unmanaged, this highly biodegradable waste may cause environmental problems, including soil and water contamination, eutrophication, greenhouse gas emissions, and other health-related concerns (De Medeiros et al. 2020).

Waste valorization is the process of converting waste materials into more valuable products, such as chemicals, materials, and fuels (Arancon et al. 2013). This approach plays a crucial role in the development of the circular economy by transforming waste products into valuable products such as organic fertilizers, animal food, and energy, as it will have a beneficial impact on environmental health, economic development, and societal well-being (Sindhu et al. 2019; Garcia-Garcia et al. 2018). Valorization of marang fruit waste has become a promising source for sustainable resource management. In this context, valorizing waste from underutilized tropical fruits represents a sustainable and promising approach to maximizing resource use and reducing waste.

Marang fruit or *Artocarpus odoratissimus* Blanco is a tropical fruit that grows mainly in the Philippines, especially in the uplands and highlands of Mindanao, and is not widespread in Luzon and Visayas. The fruit comes from the genus *Artocarpus*, which includes approximately 50 species of evergreen tropical trees native to South and Southeast Asia. The Marang fruit is usually huge and weighs an average of 1–2 kg. It is oblong in shape, rounded, and bristling with short and brittle spines, which are covered with greenish-yellow. Marang fruit consists of four components: the husk or peel, the flesh, the seeds, and the core (Alvarado 2023). The flesh accounts for 50%–60% of the fruit's weight; the remaining 40%–50% is pulp, peel, and the seed, which are typically discarded. According to a study by Abu Bakar and Abu Bakar (2018), Marang harvesting occurs in Luzon, Philippines, between May and July, and in Mindanao between August and December. The fruit weighs approximately 5 tons per acre and has a short shelf life, making it best consumed immediately. Moreover, the research shows that Marang is classified as an underutilized fruit tree partly because many people are unaware of its existence. Moreover, the Marang fruit is highly valuable for its high nutrient and bioactive content, including antioxidants in its flesh, seeds, and peel, making it a valuable source of innovation in the food, supplement, and health industries.

Pectins are structural polysaccharides within the components of the plant cell walls and are mostly made up of galacturonic acid units, and are highly variable in terms of composition, structure, and molecular weight. This polysaccharide is strongly linked to other cell wall components, such as cellulose, hemicellulose, and lignin (Harholt et al. 2010). In the food industry, pectin is a multifunctional substance used as a gelling agent, thickener, stabilizer, and emulsifier (Rascón-Chu et al. 2009). Commercial pectin is derived from apple and citrus peels, and although it shares similar features, its structure may differ depending on origin and extraction method. To this effect, it should be noted that pectin is vulnerable to physical, chemical, and/or enzymatic alteration. The high number of functional groups within the pectin structure can confer various functions, and some modifications enable pectin to have a wide range of applications (Freitas et al. 2021). In the majority, partly because pectin is a safe, non-toxic product with a low-cost production system, and is highly available.

Despite improvements in waste valorization, a research gap remains in the use of by-products from local, underutilized fruits. Although mainstream crops and generally accessible fruit by-products have been investigated most, numerous region-specific fruits with unique characteristics remain underexplored as sources of value-added food ingredients. Overlooking these locally available resources underscores the importance of targeted research, which can yield not only economic benefits but also environmental advantages through the reasonable use of the resources at hand. Thus, this study aims to use the underutilized marang fruit as an alternative source of pectin using enzyme-assisted extraction (EAE). In particular, the study aims to assess the physicochemical properties and functional characteristics of the pectin extracted to determine its potential use as a value-added food ingredient.

2 | Materials and Methods

2.1 | Sample Collection and Preparation

The sample was prepared according to the study of K. Castillo-Israel et al. (2014) with slight modifications. Ripe and overripe (or overmature) marang (*A. odoratissimus* Blanco) were collected from the local market in Cagayan de Oro City, Misamis Oriental, Philippines. After washing, the fruit peels, cores, and seeds were separated from the edible part of the fruit (flesh) and soaked in a 0.5% sodium metabisulfite ($Na_2S_2O_5$) solution in water for 1 h to prevent browning before drying. It was allowed to dry in a convection drying oven (UF-160, Memmert Oven) for 24 h at 55°C. The dried sample was ground using a fabricated milling machine and passed through 100-mesh (0.15 μm) and 200-mesh (0.075 μm) sieves. The powdered fruit sample was packed into an LDPE bag, sealed, and stored until extraction.

2.2 | Chemicals and Reagents

The chemicals and reagents used in this research were hydrochloric acid (HCl) (RCI Labscan), sodium hydroxide (NaOH) (Scharlau), sodium chloride (NaCl) (Sinopharm AR), phenol red, and ethanol (95.0% purity). All the reagents were ACS and analytical grade. The sodium metabisulfite, cellulase (10,000 U/g), xylanase (20,000 U/g), and α-amylase (40,000 U/g) are all food-grade.

2.3 | EAE of Pectin

Pectin extraction was prepared according to the study of Castillo-Israel et al. (2019), with slight modifications. In this process, cellulase, xylanase, and α-amylase were diluted to 55.5 U/mg and then added in equal portions to the marang powder (peel and core), which was incubated at 45°C ± 5°C with continuous stirring at pH 4.4. The extraction parameters (temperature, pH, and

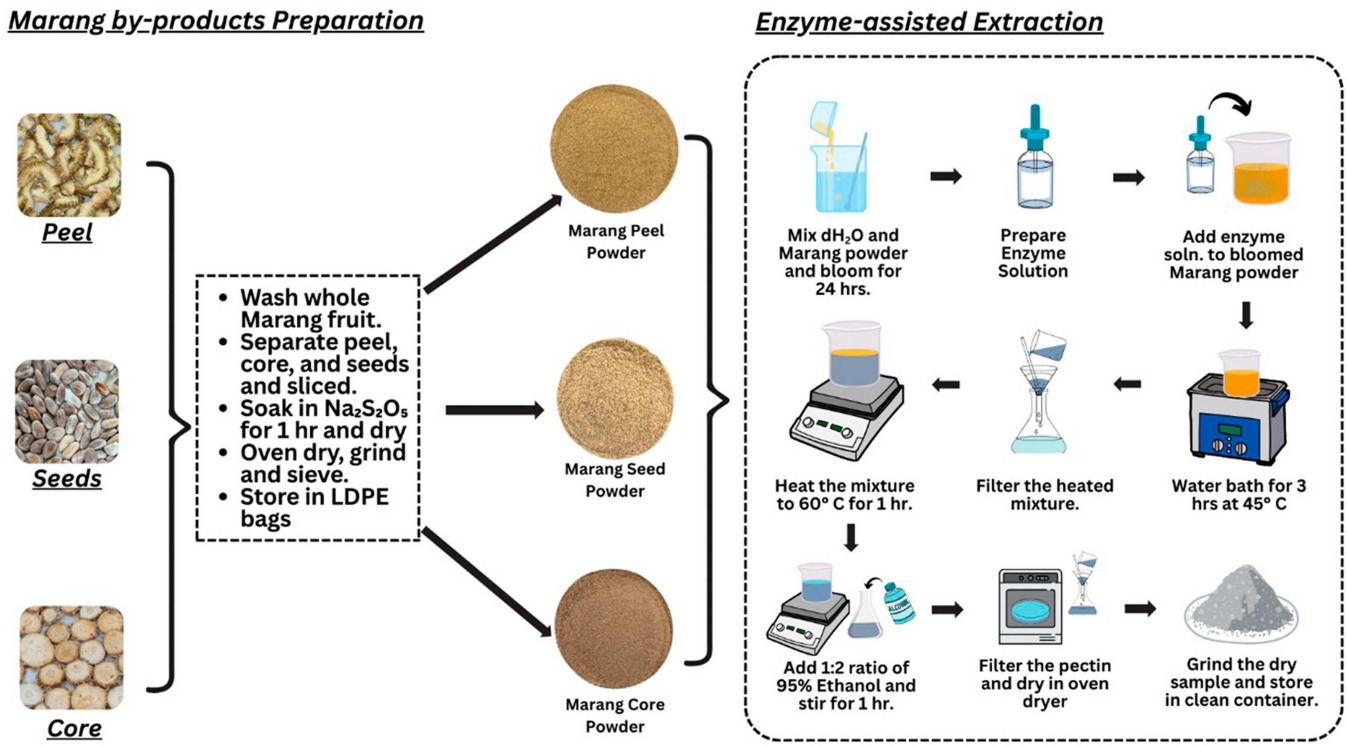


FIGURE 1 | Process flow for marang by-products preparation and enzyme-assisted pectin extraction.

enzyme concentration) were selected based on the optimal conditions for each enzyme and verified in preliminary experiments using marang fruit waste. The final conditions were selected to maximize pectin yield while maintaining polysaccharide structure, ensuring high extraction efficiency and feasibility. After the enzymatic treatment, the mixture was filtered through cheesecloth, and the filtrate was heated at 55°C for 1 h to inactivate the enzymes. Ethanol was used to precipitate pectin by adding twice the volume of the filtrate and stirring for 1 h. After this, the mixture was filtered using previously weighed Miracloth. Finally, the extracted pectin was air-dried in a convection drying oven (UF-160, Memmert Oven) for 5 h at 50°C. The dried sample was ground using a fabricated milling machine and passed through a 200-mesh (0.075 μm) sieve. The pectin powder was packed into an LDPE bag, sealed, and stored until further analysis. The process flow for the extraction of marang pectin is presented in Figure 1.

2.4 | Physico-Chemical Characterization of Marang Pectin

2.4.1 | *Pectin Yield (%)*

Pectin yield refers to the amount of pectin recovered and purified from a given amount of raw material. It is measured as the ratio of the weight of the dried pectin to the weight of the original powder of the raw powder in the extraction process (Kamal et al. 2020). The pectin yield was determined using the equation:

$$Y_{\text{pectin}}(\%) = \frac{P}{W_s} \times 100, \tag{1}$$

where Y_{pectin} (%) denotes pectin percent yield, P denotes the weight of pectin, and W_s denotes the initial amount of dried sample.

2.4.2 | *Determination of Moisture and Ash Content*

The moisture and ash content of the extracted pectin were determined according to AOAC (2005) protocols. The moisture content was given in terms of the percentage of weight loss with respect to the original weight of the sample, while the ash content was the percentage of inorganic residue against the sample weight. Measures were conducted in triplicate and reported as mean ± standard deviation. The moisture and ash content were determined using the equation, respectively:

$$\text{MC}(\%) = \frac{\text{weight of water}}{\text{weight of pectin sample}} \times 100, \tag{2}$$

$$\text{AC}(\%) = \frac{\text{weight of inorganic residue}}{\text{weight of pectin sample}} \times 100. \tag{3}$$

2.4.3 | *Determination of Equivalent Weight (EW)*

Pectin EW was calculated based on the procedure provided by Roy et al. (2017). A 0.5 g pectin sample was placed in a 250 mL conical flask and moistened with 5 mL of ethanol to facilitate EW measurement. The addition of 1 g of sodium chloride was used to sharpen the endpoint of the titration. Then, 100 mL of carbon-free distilled water and six drops of phenol red indicator were added. Gradual titration with a 0.1 N standardized solution of NaOH was performed until the indicator turned pink (pH 7.5), and the mixture was held for at least 30 s. The following equation will be used to compute the EW:

$$\text{EW (mg/mole)} = \frac{\text{Wt of pectin} \times 1000(\text{g})}{\text{mL of alkali} \times \text{normality of alkali}}. \quad (4)$$

2.4.4 | *Determination of Methoxyl Content (MeO)*

The MeO content was determined according to the procedure of Roy et al. (2017). The neutralized solution from the titration of EW was used. In the solution, 25 mL of 0.25 N NaOH was added to the solution and left to stand at room temperature for 30 min. This was followed by the addition of 25 mL of 0.025 N HCl, and titration was carried out with 0.1 N NaOH until the pink color persisted for 30 s. The MeO was measured using the equation:

$$\text{MeO (\%)} = \frac{\text{normality of NaOH (N)} \times \text{vol of NaOH (mL)} \times 3.1}{\text{Wt of sample}} \times 100. \quad (5)$$

2.4.5 | *Determination of Anhydrouronic Acid (AUA)*

The AUA value of pectin was estimated using EW and methoxyl content measurements. K. Castillo-Israel et al. (2014) reported the following formula for calculating AUA content:

$$\text{AUA (\%)} = \frac{176 \times 0.1 \times 100}{w \times 1000} + (z + y). \quad (6)$$

In this study, w is the sample weight, z the mL of NaOH from the EW measurement, and y is the mL of NaOH from the MeO determination. AUA's molecular unit (1 unit) is equivalent to 176 g.

2.4.6 | *Determination of Degree of Esterification (DE)*

The DE of the extracted pectin was calculated using Equation (7) based on MeO and AUA contents:

$$\text{DE (\%)} = \frac{176 \times \text{MeO content (\%)}}{31 \times \text{AUA (\%)}} \times 100. \quad (7)$$

2.5 | Appearance and Color Visualization of Pectin

The appearance was captured using an iPhone 7 smartphone with a 12-megapixel wide-angle lens (f/1.8 aperture) and telephoto lens (f/2.8 aperture). The shutter speed was adjusted to 1/24 s according to the conditions of light. The color was evaluated using a Minolta colorimeter (CR-410, Konica Minolta, Tokyo, Japan). The pectin samples were put on the standard white plate ($L^* = 89.16$, $a^* = 2.31$, and $b^* = 4.67$), and the degree of lightness (L^*), redness or greenness (a^*), and yellowness or blueness (b^*) of the pectin were measured. Meanwhile, the whiteness index relates to visual assessments for certain white and near-white surfaces. The color difference and whiteness index were calculated using the following formula:

$$\Delta E = [(L_{\text{pectin}} - L_{\text{std}}]^2 + (a_{\text{pectin}} - a_{\text{std}})^2, + (b_{\text{pectin}} - b_{\text{std}})^2]^{1/2} \quad (8)$$

$$\text{WI} = [100 - \sqrt{[100 - L)^2 + a^2 + b^2)}]. \quad (9)$$

2.6 | Functional Properties

2.6.1 | *Water-Holding Capacity (WHC)*

The determination of WHC was performed according to the assessment method described by Mohamad Zahari et al. (2016) with slight modifications. A 100 mg of pectin powder extract was combined with 10 mL of distilled water in a 10 mL centrifuge tube and allowed to mix overnight at 5°C. Then, the mixture was centrifuged at 1401 g for 20 min. The slurry was dried at room temperature for 2 h after the removal of the supernatant. After this, the wet residue was weighed and then dried at 120°C for a period of 2 h and again weighed to receive the dry weight. The WHC was calculated as

$$\text{WHC } (g/g) = \frac{\text{g of water}}{\text{g of pectin}}. \quad (10)$$

2.6.2 | *Oil-Holding Capacity (OHC)*

The OHC was determined following the procedure outlined by Mohamad Zahari et al. (2016) with minor modifications. A 100 mg pectin sample was combined with 10 mL of soya oil in a centrifuge tube, mixed thoroughly using a vortex mixer for 1 min at max speed, and then centrifuged at 1401 g for 30 min. The supernatant was weighed, and the OHC was expressed:

$$\text{OHC (g/g)} = \frac{\text{g of precipitate after centrifuge}}{\text{g of pectin}}. \quad (11)$$

2.6.3 | *Emulsion Stability (ES) and Capacity (EC)*

EC and ES were evaluated based on the procedure described by S. W. Cui and Chang (2014), with minor modifications. A 0.01 g sample of pectin powder was first suspended in 5 mL of distilled water. Then 5 mL of soya oil was put into the suspension. This was followed by mixing the mixture in a vortex mixer at the highest speed for 3 min to create an emulsion and centrifuging at a speed of 1401 g for 5 min. The EC was expressed as the % ratio of the emulsion layer height to the total fluid height, and calculated using the formula:

$$\text{EC (\%)} = \frac{\text{height of emulsion layer}}{\text{total height of fluid}} \times 100. \quad (11)$$

The stability of the emulsion (ES) was determined by heating the emulsion to 80°C for 30 min and then cooling it in running tap water for 15 min. This emulsion was again centrifuged after being cooled at 1401 g for 5 min. The ES was determined by the formula:

$$\text{ES (\%)} = \frac{\text{height of remaining emulsion layer}}{\text{total heigh of fluid}} \times 100. \quad (13)$$

2.6.4 | *Swelling Capacity (SC)*

The determination of SC was conducted based on Mohamad Zahari et al. (2016). A sample of 100 mg of pectin was put in a cone-shaped plastic tube, stirred with 5 mL of distilled water, and allowed to stand overnight. The SC was determined using:

$$\text{SC}\ (\%) = \frac{\text{mL of swollen material}}{\text{g of pectin}} \times 100. \quad (14)$$

2.7 | Solubility of Pectin

2.7.1 | *Solubility of Dry Pectin in Cold and Hot Water*

Pectin can be soluble, a significant determinant of the texture and feel of formulated products. Pectin that dissolves too quickly will not have the desired gelling or thickening properties, and pectin that is too insoluble can result in unpleasant textural results. To determine solubility, 0.15 g of extracted pectin was placed in a conical flask. It was then dissolved in a solution of 25 mL of distilled water and 5 mL of 95% ethanol, after which the solubility was first noted at room temperature. It was then heated to 90°C for 15 min to further assess solubility, as described by Rahman et al. (2023) with slight modifications.

2.7.2 | *Solubility of Dry Pectin in Cold and Hot Alkali Solutions*

The solubility of pectin in acid and alkali solutions was based on the method of Rahman et al. (2023) with modifications. The pectin sample (0.3 g) was weighed and placed in a conical flask. Fifty milliliters of distilled water and 10 mL of 99% ethanol were added to it. Out of this solution, 5 mL of pectin solution was collected, and 1 mL of 0.1 N NaOH was introduced. The solubility of pectin was observed before and after the heating of the mixture at 90°C for 15 min.

2.8 | Statistical Analysis

All measurements made in the experiment were taken in triplicate, with the results reported as mean ± standard deviation. The data were also checked for Normality using the Shapiro–Wilk test, and all the data sets were proven to be normally distributed ($p > 0.05$). The statistical analyses were performed using SPSS software (IBM Corp., version 23, Armonk, NY, USA). The treatment means differences were compared with the one-way analysis of variance (ANOVA) at a significance level of $p \leq 0.05$. A multiple-range Tukey honestly significant test was used to examine differences between the mean values of the measured properties with a significance level of 0.05. Additionally, principal component analysis (PCA) and Pearson correlation analysis were conducted to examine the connections between the physicochemical and functional characteristics of the extracted pectin using JMP Student Version 2025 (SAS Institute Inc., Cary, NC, USA).

3 | Results and Discussions

3.1 | Physico-Chemical Properties

3.1.1 | *Pectin Yield*

The pectin yield of the marang by-products (peel and core) was different with respect to the enzyme employed in extracting the pectin. The cellulase-assisted extraction yielded the highest pectin content in the core (7.05% ± 0.29%), which was much higher than that of any other treatment. This is because cellulose is hydrolyzed by cellulase, which breaks it down within the cell wall matrix, thereby releasing pectic material that is firmly attached to the lignocellulosic structure (Jayasekara and Ratnayake 2019). For the peel, xylanase extraction yielded the highest (5.32% ± 1.74%), followed by cellulase (3.68% ± 0.12%) and xylanase (3.06% ± 0.45%). This implies that the linkages between hemicellulosic polysaccharides in the peel matrix and pectic polymers could be stronger, and that xylanase activity effectively cleaves these linkages, enabling higher pectin yield (Liu et al. 2022). The greater variability in xylanase activity might be due to the heterogeneity in the distribution of hemicellulose within the peel tissue. Conversely, the lowest yields were obtained with α-amylase in peel (3.06 ± 0.45) and core (4.59 ± 0.82), suggesting that starch hydrolysis was insufficient to release a large amount of pectin (Bai and Gilbert 2022). However, its moderate pectin yield in the core suggests that starch–pectin interactions may be present in this tissue, which are partially disrupted by amylase activity. Moreover, a preliminary experiment was also conducted on marang seeds, which revealed 0% pectin yield in all the enzymes tested. This shows that the enzymatic treatments used in this research were ineffective at liberating pectin in seed tissues. As a result, marang seeds were not included in the main experimental results and further analyses.

In a study by K.A.T. Castillo-Israel et al. (2019) on the extraction of pectin from different fruit peel waste, the results of EAE could be explained by the high specificity of enzymes during hydrolysis. The researchers have observed that those enzymes bind only a few components of the polysaccharide, which are bound to the pectin molecule, thus yielding less pectin. In this study, marang pectin yields a lower % recovery, consistent with the study of Maric et al. (2018), which compared various techniques for extracting pectin from golden kiwi pomace. The results showed that enzymatic extraction of kiwi fruit pomace yielded the highest pectin content (4.5%). However, Wikiera et al. (2015) also reported the highest pectin yields with EAE using Celluclast and Viscoferm, at 18.95% and 17.86%, respectively. However, the enzyme extraction method is highly effective, saving time and reducing the amount of solvent required. However, the high cost of enzymes has made their widespread use in industry difficult. Enzymes also exhibit specificity and selectivity, so choosing the proper enzymes for each raw material depends on the pectin % yield extracted (Table 1).

3.1.2 | *Moisture Content and Ash Content*

The moisture and ash content of pectin samples obtained as a result of marang peel and core by-products are given in Table 2. There were considerable differences between the samples, depending on the by-product origin (peel or core) and the enzyme type used in the extraction process. The pectin samples had a range of % MC of 12.49% (CC) to 17.92% (PA). The PA had the highest % MC of 17.92% ± 1.54%, which was significantly higher ($p < 0.05$) than those of PC and PX. In contrast, core-derived pectin had a lower value, with CC having the lowest % MC. This implies that lower moisture content confers greater stability and resistance to microbial growth and degradation during storage (Rodulfo et al. 2024; Rahman et al. 2023). According to the IPPA (2014), the moisture content for commercial pectin should not exceed 12%. The results of this study are higher than the standards, except for the core-derived pectin

TABLE 1 | Pectin yield (%) from marang by-products (peel and core) using different microbial enzymes.

Extracting enzymes	Marang by-products Y_{pectin} (%)			
	Abbr.	Peel	Abbr.	Core
Cellulase	PC	3.68 ± 0.12^{a}	CC	7.05 ± 0.29^{a}
Xylanase	PX	5.32 ± 1.74^{a}	CX	3.52 ± 1.04^{b}
α-Amylase	PA	3.06 ± 0.45^{a}	CA	4.59 ± 0.82^{b}

Note: abbr. denotes the abbreviation of pectin samples. Data are presented as mean ± standard deviation (SD). Different superscripts within a column indicate significant differences among marang by-products (peel and core)($p < 0.05$) based on Tukey's HSD test.
Abbreviations: CA, core extracted from α-amylase; CC, core extracted from cellulase; CX, peel extracted from xylanase; PA, peel extracted from α-amylase; PC, peel extracted from cellulase; PX, peel extracted from xylanase.

TABLE 2 | Moisture and ash content of pectin extracted from marang peel and core by-products using different enzymes.

	Pectin sample	Moisture content (%)	Ash content (%)
Peel	PC	15.49 ± 0.60^{b}	15.69 ± 1.20^{a}
	PX	16.30 ± 0.55^{b}	17.05 ± 0.40^{a}
	PA	17.92 ± 1.54^{a}	18.80 ± 0.64^{a}
Core	CC	12.49 ± 0.37^{c}	10.11 ± 0.46^{a}
	CX	13.36 ± 2.67^{b}	13.66 ± 0.61^{a}
	CA	16.46 ± 0.58^{a}	15.10 ± 0.50^{a}

Note: Data are presented as mean ± standard deviation (SD). Using the Tukey honestly significant difference test, mean values with different letters in a column for each by-product (peel and core) indicate a significant difference ($p \leq 0.05$).

from the cellulase sample. Several studies highlight differences in pectin moisture content across various factors. According to Akhster et al. (2024), these differences may be explained by the fruit's inherent moisture content, the methods used to extract it, and laboratory factors. Panchev et al. (2010) also noted that the difference may depend on pH, the solvent's chemical composition, and possible interactions between pectin and the extraction medium. Similarly, Rahman et al. (2023) explained that the chemical structure, fiber composition, water-retention capacity, and drying process of pectin might explain why they exhibit varying moisture content. However, a lower moisture content is preferred, as it can be stored longer and is more applicable to industrial use (Muhmadzadeh et al. 2010).

The ash content of the marang pectin, which reflects the mineral composition, is presented in Table 2. The values ranged from 10% to 15% for both peel and core. The sample of peel-derived pectin is higher compared to core-derived pectin. This observation indicates that the marang peel contains more mineral-associated polysaccharides than the core, possibly because it is more exposed to the environment and is accumulating mineral constituents in the outer fruit tissues. However, the result of this study exceeds the standards set by the IPPA (2014), which states that ash content should be below the maximum allowable value of 10%. Higher ash content implies higher impurities, and ash levels below 10% in pectin are better for good-quality gel formation (Rahman et al. 2023). Moreover, fruit type, pre-treatment, and extraction process can affect variations in ash content (Mamiru and Gonfa 2023). The difference in the variation of ash content between marang pectin samples could be attributed to the difference in the part of the fruit (peel or core), enzymatic treatment, as well as the interplay of polysaccharides with mineral elements. These results indicate that pectin properties should be carefully selected based on the raw material and extraction method to achieve the desired pectin specifications, whether for functionality as an ingredient or for high purity and conformity to industry standards in applications.

3.1.3 | *EW of Pectin*

The EW of pectin extracted using three different commercial enzymes from the peel and core of the marang fruit is presented in Figure 2a. The EW is a significant parameter for characterizing and assessing the quality of isolated pectin. It is the average molecular weight of a free galacturonic acid unit (not esterified). The high EW pectin is directly linked to its high functional performance, which is correlated with a higher content of free galacturonic acid in its molecular chains, resulting in high viscosity and water-binding properties (Baraiya et al. 2023). For marang peel in this study, the mean EW values were approximately 575 mg/mole with cellulase-extracted pectin, 605 mg/mole with xylanase-extracted pectin, and 705 mg/mole with α-amylase-extracted pectin. The EW in the amylase was significantly higher ($p < 0.05$) compared with both cellulase and xylanase.

The marang peel extracted by α-amylase is significantly higher ($p < 0.05$) compared to cellulase and xylanase. The result is unexpected on a conventional pectin extraction basis, since α-D-(1,4)-glucan linkages on starch and other polysaccharides are not the target of amylase but rather catalyze the hydrolysis of galacturonic acid that comprises pectin (Hrabovska et al. 2018). The high efficiency in α-amylase extraction should then be explained by an indirect, yet highly efficient mechanism. It has been shown that pectin and starch can coexist and interact within the plant cell wall, forming complex gel-like structures or physical entanglements through hydrogen bonds (Bai and Gilbert 2022). The high EW in α-amylase-extracted pectin indicates that the molecules of pectin that were extracted were not depolymerized. This implies that the α-amylase was not involved in the direct degradation of the pectin polymer; instead, it acted as a substantial starch-based barrier that was removed. The enzyme hydrolyzed starch fibers, which were cross-linked with or physically trapped pectin, and, in the process, declogged the matrix, enabling high-quality pectin extraction in a non-destructive manner.

On the other hand, the derived pectin from the marang core has a mean value of 610, 695, and 640 mg/mole for cellulase,

TABLE 3 | Color parameters (L^*, a^*, and b^*), color difference (ΔE), and whiteness index (WI) of pectin extracted from marang.

Marang by-products	Pectin sample	L	a^*	b^*	Color	ΔE	WI
Peel	PC	57.21 ± 3.26^{a}	12.01 ± 1.90^{a}	36.23 ± 0.57^{a}		38.19 ± 0.96^{c}	42.63 ± 3.06^{c}
	PX	66.54 ± 0.51^{a}	8.25 ± 0.58^{b}	34.37 ± 1.08^{a}		35.35 ± 1.16^{b}	51.33 ± 1.12^{a}
	PA	67.72 ± 0.81^{b}	10.32 ± 0.49^{b}	22.69 ± 1.10^{b}		24.93 ± 1.21^{a}	59.21 ± 1.38^{b}
Core	CC	67.24 ± 7.36^{a}	10.54 ± 3.17^{a}	31.45 ± 2.03^{a}		33.22 ± 3.18^{a}	53.29 ± 7.47^{a}
	CX	72.16 ± 5.27^{a}	8.55 ± 2.06^{a}	30.5 ± 2.26^{a}		31.71 ± 2.28^{a}	57.70 ± 4.45^{a}
	CA	57.21 ± 3.26^{a}	12.01 ± 1.90^{a}	36.23 ± 0.57^{b}		38.19 ± 0.96^{a}	42.63 ± 3.06^{a}

Note: Values are presented as mean ± standard deviation. Using the Tukey honestly significant difference test, mean values with different letters in a column for each by-product (peel and core) indicate a significant difference ($p \leq 0.05$).

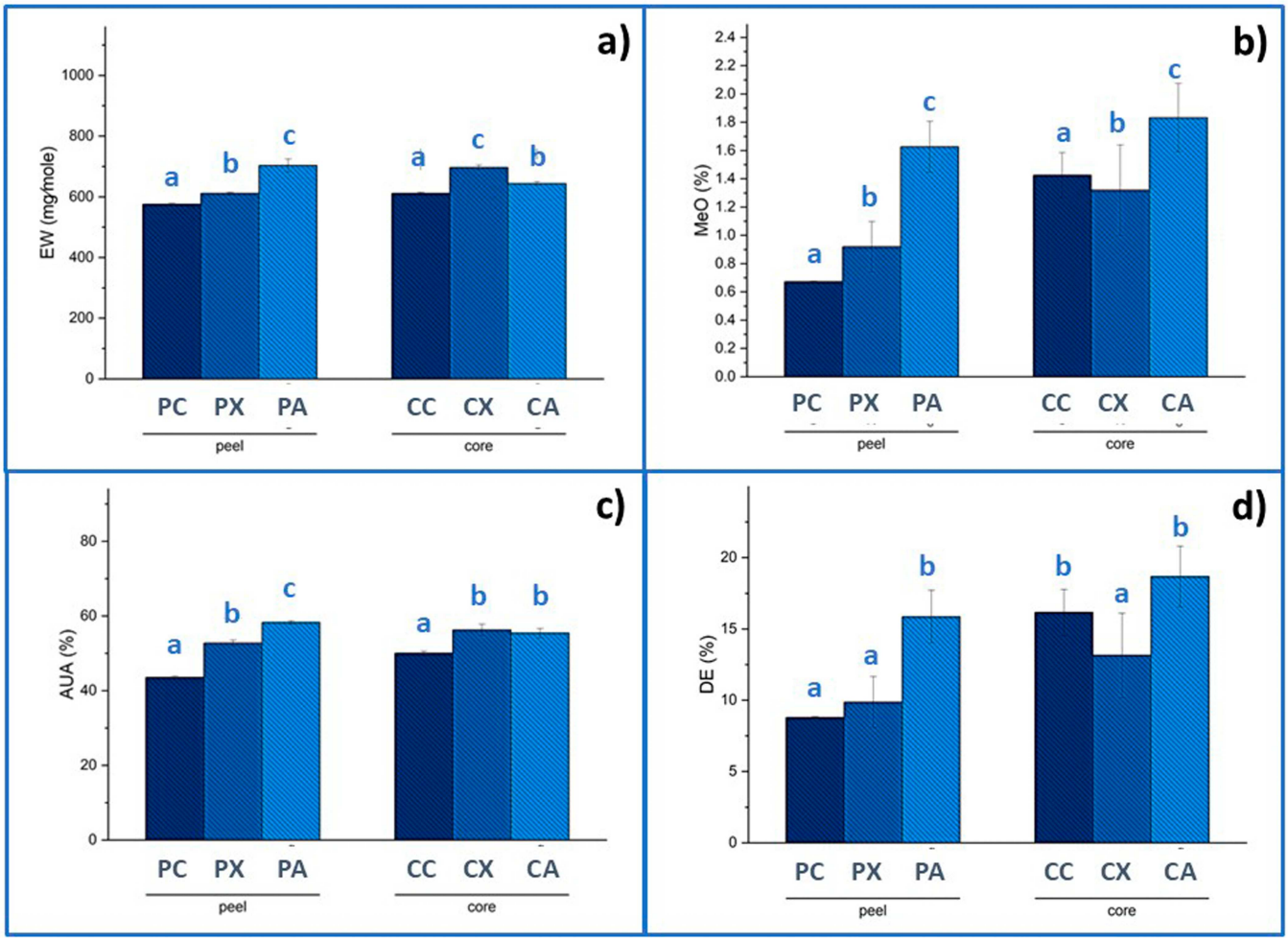


FIGURE 2 | Comparative analysis of the physico-chemical properties of pectin from marang peel and core using different microbial enzymes. (a) Equivalent weight (EW); (b) methoxyl content (MeO %); (c) anhydrouronic acid content (AUA %); and (d) degree of esterification (DE %). Significant differences ($p \leq 0.05$) between treatments are indicated by different letters. Error bars represent standard deviations.

xylanase, and α-amylase, respectively. The pectin extracted from xylanase is significantly higher ($p < 0.05$) compared to cellulase and α-amylase. The results indicate that xylan (hemicellulose) is the major structural component of the cell wall matrix of the core (Santos et al. 2014), making xylanase the preferred EAE method for targeted degradation and the subsequent release of pectin polymers. Cellulase and xylanase are common enzymes used in EAE because their main activity is breaking the physical walls around pectin, not degrading the pectin molecule.

The values of EW of pectin in Marang pectin, which are produced in this study, are competitive with the pectin extracted from other studies. For instance, in the study by Islam et al. (2023) on pectin derived from jackfruit by-products, the EW was determined to be 555.56 ± 1.23 mg/mole (tandem), 476.19 ± 0.81 mg/mole (peel), and 454.54 ± 0.50 mg/mole (core). The EW of marang pectin is higher than that of the jackfruit and also exceeds that of the study of Panwar et al. (2022) on *Citrus limetta* peel, resulting in 477.89 ± 0.43 mg/mole, and the study of Ghoshal and Negi (2020) for kinnow peel resulted in an EW of 652.14 ± 0.21 mg/mole.

However, the results of this study are contrary to those of K.A.T. Castillo-Israel et al. (2019) for fruit peel pectins extracted with different enzymes. Passion fruit exhibited the best EW in combination with xylanase (2781.3 g/mole) and saba banana with cellulase (3201.5 g/mole). The highest EW (3230.1 g/mole) was observed for α-amylase in mango peel. Remarkably, Guyabano peel had the highest EW (8409.7 g/mole) with α-amylase, far than any other treatment and fruit. This study also compares the EW results for commercial pectin (control) with those for apple and citrus pectins, yielding 2809.57 and 4016.12 mg/mole, respectively. The relatively lower EW values of marang pectin compared to other fruit sources, such as passion fruit, saba banana, mango, and guyabano, and commercial varieties, may be due to various factors. According to J. Cui et al. (2021), the cell wall composition of different fruits is important.

In comparison to these other fruit peels, the proportion of tightly bound pectin or portions of galacturonic acid chains that can be readily hydrolyzed may be less in marang peel and core, causing shorter chains of polymer to form after extraction. Another factor to be considered is the extraction process: although enzyme treatments improve preservation better than acid hydrolysis, variation in enzyme activity, concentration, incubation time, and temperature may influence levels of depolymerization (Sharma et al. 2016). In this study, milder enzyme conditions are likely to have released pectin molecules of smaller molecular weight than those observed in other fruits under optimized EAE conditions.

However, both values of the marang by-products (peel and core) pectin extracted using different microbial enzymes were higher than the International Pectin Producer Association's minimum suggested EW of 400 mg/mole (IPPA 2014). The high EW values indicate that the isolated pectin can be used in high-end applications where gelling, thickening, and stabilizing properties are most important (Pattarapisitporn and Noma 2025).

3.1.4 | *Methoxyl Content of Pectin*

One of the parameters that determines the functionality of pectin is its methoxyl content, which determines the ability of pectin to form gels in high-solid, low pH conditions by forming hydrophobic interactions and hydrogen bonds, as well as its ability to form gels in an ionic form by binding divalent cations such as calcium (Schmelter 2002).

The methoxyl content (MeO%) of marang pectin extracted using cellulase, xylanase, and α-amylase was $0.67\% \pm 0.05\%$, $0.89\% \pm 0.20\%$, and $1.63\% \pm 0.18\%$, respectively, for peel. Meanwhile, the pectin extracted from the core resulted in $1.43\% \pm 0.15\%$, $1.34\% \pm 0.28\%$, and $1.83\% \pm 0.29\%$ for cellulase, xylanase, and α-amylase, respectively. The enzymes, including cellulase and xylanase, which break down the cell wall, can also be co-extracted with starch and pectin. Because starch lacks methoxyl groups, its presence in the end product serves as a diluent. Failure to co-extract starch will cause the co-extracted starch to precipitate with pectin during the final purification step of alcohol precipitation, resulting in a final product composed of a mixture of pectin and starch (Hrabovska et al. 2018). The only difference is a clear, consistent pattern: better α-amylase performance ($p < 0.05$). The most important role of α-amylase in this regard is purification. Hydrolyzing the interfering starch into smaller, soluble sugars prevents the latter from co-precipitating with the pectin. The increased MeO% %, therefore, does not result in the enzyme altering the structure of the pectin, but the result was the creation of a more concentrated and purer extract containing pectin. This purification step, performed by amylase, is targeted and yields a better representation of the actual methoxyl content of pectin (Hrabovska et al. 2018).

Another observation is the difference in the source of material; the pectin extracted from the marang core consistently shows higher MeO than the peel, regardless of the enzymes employed. However, all measured MeO % values are below 2%. The results suggest an inherent difference in the chemical structure of pectic polysaccharides in these two parts of the fruits. The marang core can be either more esterified or in a simpler, less fibrous matrix and so yield a purer extract. The peel of marang has a very complex matrix rich in crude fiber and other phytochemicals, which present a difficulty to selectively extract high-quality pectin (Estudillo and Tolentino 2024). The higher complexity and diversification of cellular structure in the peel may be the cause of the non-pectin material or less-esterified fractions of pectin being extracted together and giving a lower measured MeO %.

According to Elsyana and Alvita (2022), pectin is defined as high when the % MeO in its composition is equal to or greater than 7 (HM). In case the MeO % content was lower than 7, the content would be considered low in methoxyl (LM). Based on the extremely low MeO % values in this study, the extracted marang pectin would be classified as a low-methoxyl pectin or LM. The low methoxyl content of the marang pectin will then be capable of creating a gel when the polyvalent cations, like calcium, are added. Thus, in this case, low methoxyl pectin will be profitable since it can be manufactured without the demethylation process of high methoxyl pectin to low methoxyl pectin.

The low MeO % of marang pectin is very low, and this fact would not allow any generalization that EAE is always the best one to use to produce high-quality pectin. The high deviation of the result of the study could be attributed to several factors. It is even likely that the low values represent a natural characteristic of the pectin of this particular species of marang. The properties of pectin are considered to have a great variation depending on the plant variety, genetic type, and stage of maturity (Obodo-Ovie et al. 2024). Another is that the conditions of extraction were not necessarily optimized with marang. Operational parameters like temperature, pH, and time of extraction are highly sensitive to the quality and yield of pectin (Peighambardoust et al. 2021). It is quite probable that the conditions used in the experiment, although suitable for the general activity of enzymes, were less than ideal with this particular raw material, resulting in a certain level of de-esterification or nonselective co-extraction that reduced the result in MeO %.

3.1.5 | *AUA of Pectin*

Determination of AUA content is done through the quantity of uronic acids in relation to the overall mass of the extracted product. Pectin can be described by the AUA and the DE. The purity of the pectin is a measure of the AUA content (characterizing the GalA) (Kheowmung 2024). The higher the AUA value, the purer the pectin. The higher the purity, the higher the gel strength. The pectin extracted with cellulase, xylanase, and α-amylase from

marang peel and marang core shows a significant variation in the AUA content obtained. As shown in Figure 2c, the data indicate differences in the efficacy of each enzyme in producing high-quality pectin. The highest AUA content was obtained with α-amylase extraction at 58.3%, which was significantly higher ($p < 0.05$) than with xylanase ($p < 0.05$) and cellulase ($p < 0.05$), resulting in 53.8% and 43.2%, respectively. A similar trend was observed in the marang core pectin, where α-amylase yielded the highest AUA content, resulting in 56.4%. Pectin that had been extracted using xylanase and cellulase resulted in 56.0% and 50.1%, respectively. The AUA content of α-amylase is significantly higher ($p < 0.05$) than that of cellulose, but no significant difference ($p > 0.05$) in the AUA content of xylanase.

The AUA results for both the marang peel and core using α-amylase were higher than those with other enzymes. This is contrary to expectations regarding the primary role of pectin-liberating enzymes, and there is a need to understand the composition of marang fruit better. Although enzymes such as cellulase and xylanase are designed to degrade the cell wall matrix, thereby physically releasing pectin, the primary activity of α-amylase is the hydrolysis of starch into small sugar units (Jiang et al. 2024).

The higher AUA content obtained using the α-amylase enzyme can be attributed to the purification process rather than to direct extraction. Marang, being a known source of starch, is an *Artocarpus* species that includes the edible pulp of the type and the central fibrous core (Alvarado 2023). During extraction, other polysaccharides, such as starch and other non-pectin glucans, co-extract with pectin (Noorfarahzilah et al. 2019). In this regard, α-amylase could target contaminating starch molecules for degradation. Hydrolysis of these non-pectin carbohydrates by α-amylase increases the proportion of the relative concentration of the uronic acid-rich pectin in the final product. This would be reflected directly in a higher AUA percentage, as observed in the results. This implies that, for starchy biomass, the enzyme used for pectin extraction should not only be able to handle the cell wall structure but also account for and counteract the effects of co-extracted contaminants. The AUA results for both the marang peel and core using α-amylase were higher than those with other enzymes. This is contrary to expectations regarding the primary role of pectin-liberating enzymes, and there is a need to understand the composition of marang fruit better. Although enzymes such as cellulase and xylanase are designed to degrade the cell wall matrix, thereby physically releasing pectin, the primary activity of α-amylase is the hydrolysis of starch into small sugar units (Jiang et al. 2024).

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The comparative performance of cellulase and xylanase in the present study is consistent with the study by Wikiera et al. (2016) on pectin extraction from apple pomace, which indicated that xylanase (or hemicellulase) is better than cellulase at breaking down the cell wall network by hydrolyzing pectin. The improved efficiency of xylanase indicates that the cross-linking of xylanase with pectin with the hemicellulose, specifically xylans, in the marang cell wall may pose a greater barrier to the liberation of pectin than the cellulose network. Hence, with xylanase, it would be more efficient to aim at these xylan-rich chains to release the pectin polymer. This observation indicates that EAE protocols are not universal and must be specifically designed to suit the unique polysaccharide structure of the target biomass to achieve optimal outcomes.

The amount of AUA in the pectin obtained after extraction of the marang peel was always greater than that of the core, irrespective of the enzyme. This observation suggests the presence of fundamental compositional differences between the two sections of the fruit. The outer protective coat, known as the peel, is a recognized source of dietary fiber, including pectin (Liang et al. 2022), and the fibrous core is the fruit's axis, located near the starchy, edible pulp. The reduced AUA values of the core indicate that the non-pectin compounds extracted are more likely to be co-extracted and primarily starch, which would dilute the overall purity of the sample. This is consistent with the hypothesis that α-amylase is so effective because it can degrade these specific contaminants. This increased base AUA in the peel indicates that it is a better, cleaner source of high-purity pectin extraction and that it requires less purification of co-extracted materials than the core.

As per the Food and Agriculture Organization (FAO) and Food Chemical Codex (FCC), the recommended value of AUA should not be less than 65% in pectin as a food additive or as a pharmaceutical agent on an ash and moisture-free basis (FCC 2004). The AUA values of marang pectin in the current study, although showing significant differences between treatments, are all below the 65% purity level commonly reported in the literature. This implies that, despite the use of particular enzymes, high levels of co-extracted non-pectin material remain. A complete characterization of the extracted marang pectin would require further testing of these other physicochemical characteristics to establish its applicability for specific industrial uses, such as a gelling agent in jam or a stabilizer in drinks (Liang et al. 2022).

3.1.6 | *Degree of Esterification*

The relationship of the DE, MeO, and AUA relates to the properties of pectin. The content of methoxyl and AUAs is used to determine the degree of esterification, where AUA is the percentage of the pectin molecule composed of galacturonic acid. The degree of esterification is directly proportional to the MeO content and inversely proportional to the AUA content (Sayah et al. 2016). The level of esterification (DE) is a valuable measure of the structural properties and potential pectin gelling properties (Dixit et al. 2025). The DE values in this study varied with enzyme treatment and the origin of the raw material (peel vs. core). In the peel, the treatment with α-amylase yielded the

highest DE ($p < 0.05$), followed by xylanase and cellulase ($p > 0.05$). The same trend was observed in the marang core pectin, where α-amylase has the highest DE, followed by cellulase and xylanase. These findings indicate that α-amylase helped to retain or increase esterified galacturonic acid residues in pectin, which was probably due to its capacity to degrade the associated starches and expose more esterified areas of homogalacturonan. Conversely, cellulase and xylanase, which are predominant in the action on the cell wall polysaccharides, might have liberated the pectin fractions with relatively lower esterification.

The differences in DE show a strong correlation with MeO, as DE indicates the extent of carboxyl group esterification. The study's results align with the trends in MeO, which were similar to those of DE: pectin samples after α-amylase treatment produced the highest MeO, and cellulase produced the lowest. Such interaction validates the notion that the extent of enzyme-mediated structural adjustment directly affects the extent of methyl esterification, thereby informing the classification of the extracted pectins. In particular, DE values below 50% and all treatments are characterized as LM pectin that can gel in the presence of divalent cations (such as Ca^{2+}) rather than in high-sugar/acid conditions.

The same pattern was observed with AUA content, indicating the purity of pectin and the percentage of galacturonic acid in the pectin polymer. Extraction with enzymes, which led to increased DE and MeO, was also associated with increased AUA. For instance, the marang peel pectin extracted with α-amylase had higher DE and MeO values than the respective AUA. For the marang core-derived pectin, α-amylase also increased DE and MeO, as well as AUA. The positive relationship shows that enzymatic extractions that favor esterification also increase the percentage of galacturonic acid-rich fractions, consistent with the idea that both the structural integrity of pectin and the degree of esterification depend on each other (Levigne et al. 2002).

3.2 | Appearance and Color Visualization of Pectin

Visual inspection of the six pectin samples shows varying colors, depending on the source material and the extraction enzyme (Figure 3). The peel samples (a, b, c) are darker and have a richer, more saturated reddish-brown hue. In contrast, the core samples (d, e, f) are lighter and have a more moderate tan or cream color. The visually lightest sample is the one extracted by α-amylase (f), and the darkest is the one extracted by cellulase (a).

Analysis of the color of marang-derived pectin from peel- and core-derived samples revealed apparent differences. The core pectin consistently shows a lighter (L^*) and lower yellowness (b^*) compared to peel, which implies a lighter and less intense colored product. The xylanase-extracted core sample had the highest lightness, consistent with its visual appearance. The EAE has influenced the color parameters. There was a gradual increase in lightness with cellulase (PC, $L^* = 57.21$), xylanase (PX, $L^* = 66.54$), and α-amylase (PA, $L^* = 67.72$), and the latter produced the lightest pectin. On the other hand, the cellulase-extracted sample had the highest redness ($a^* = 12.01$) and yellowness ($b^* = 36.23$), and the lowest in the alpha-amylase-extracted sample ($a^* = 10.32$ and $b^* = 22.69$) (Table 3).

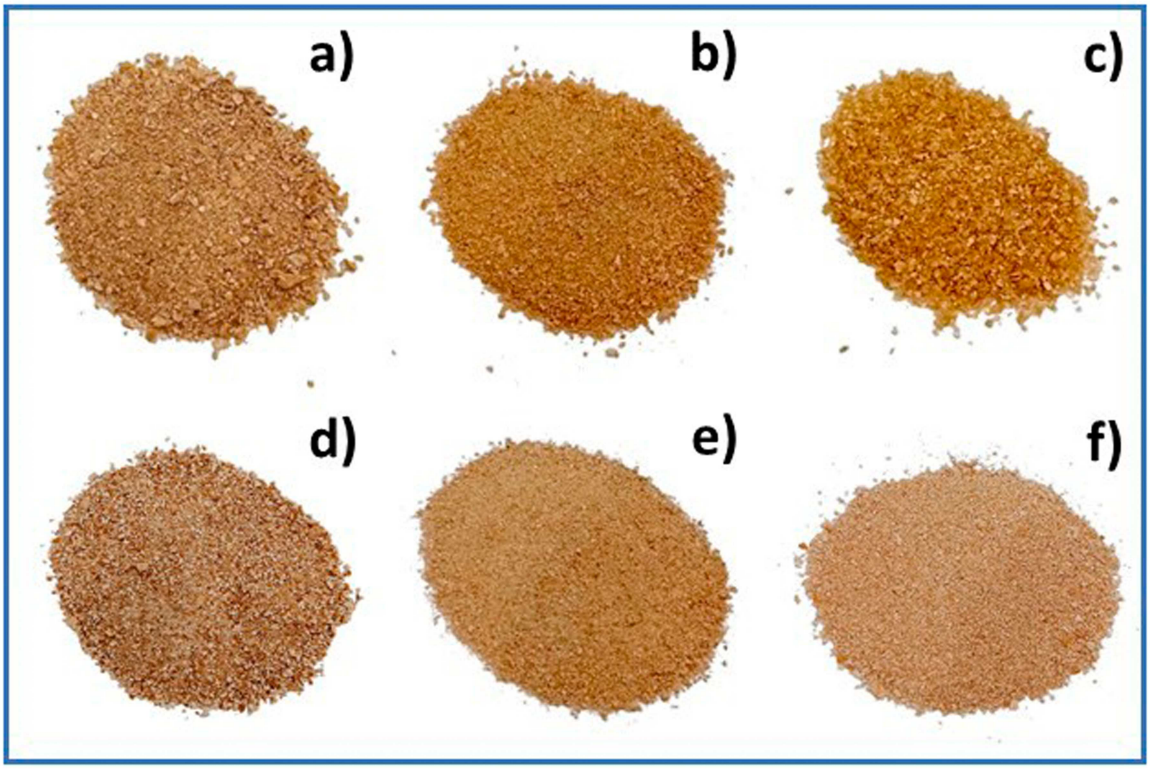


FIGURE 3 | Visual appearance of the extracted pectin. The pectin samples (a–c) were from the peel and (d–f) were from the core. The pectin samples were extracted with cellulase (a and d), xylanase (b and e), and α-amylase (c and f), respectively.

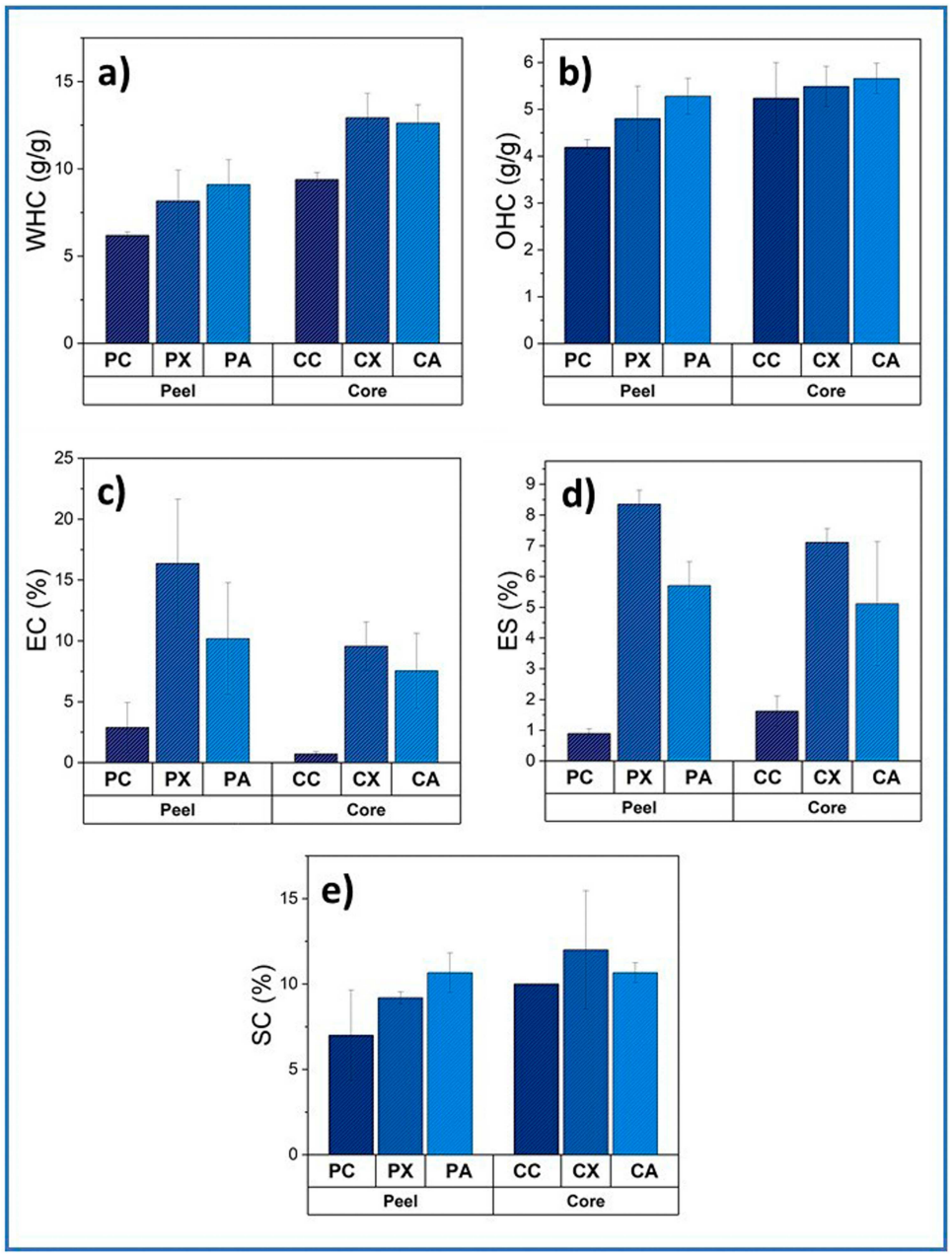


FIGURE 4 | Functional properties of the extracted pectin from marang by-products using different enzymes. (a) Water holding capacity (WHC, g/g), (b) Oil holding capacity (OHC, g/g), (c) Emulsifying capacity (EC, %), (d) Emulsifying stability (ES, %), and (e) Swelling capacity (SC, %) of pectin obtained from different sources: PC, PX, PA (peel) and CC, CX, CA (core). Significant differences ($p \leq 0.05$) between treatments are indicated by different letters. Error bars represent standard deviations.

Pectin color is not a native characteristic feature of the polysaccharide, but the result of having co-extracted non-pectin compounds of the plant matrix. The result color is a direct indication of the purity of the extracted product, which is primarily determined by two main factors: the chemical composition of the source material and the specificity of the extraction technique. The difference in the distribution of phytochemicals in the fruit can directly explain the distinct color profile of the marang peel and core pectin. Most bioactive compounds, such as phenolic compounds, flavonoids, and pigments, are found at much higher concentrations in the peeled parts of fruits and vegetables, which serve as a protective layer (Bhardwaj et al. 2022). The core of the marang is in the internal part, which has a low concentration of these protective pigments and phytochemicals. Thus, the pectin extracted from the core is considerably lighter and less colored. This is an intrinsic chemical difference between the materials used in a source, which essentially determines the starting color of the end pectin product.

The ΔE values for the marang pectin are high, ranging from 24.93 to 38.19. The ΔE values are high, indicating that the various extraction methods do not yield a homogeneous product. This has severe implications for quality and product application. On the other hand, the WI data for the samples agree with the results for L^* and b^*. Both the pectin recovered with xylanase (CX) and α-amylase (CA) have high WI values (57.70 and 42.63,

respectively), whereas that of the peel-obtained pectin that was recovered with cellulase (PC) has the lowest WI (42.63). This result is the interplay between the lightness (L^*) and yellowness (b^*) values. While the lightness of PA ($L^* = 67.72$) is slightly lower than that of CX ($L^* = 72.16$), the extremely low yellowness ($b^* = 22.69$) offsets this difference, resulting in a superior overall whiteness index. This is another indication that α-amylase has a great capacity to reduce the co-extraction of the yellowing pigments, which are especially detrimental to the whiteness of a product. A high WI indicates greater whiteness and lightness in the product.

This observation has implications for the industrial valorization of marang by-products. When a food manufacturer is interested in a premium ingredient needed for applications where a neutral or white color is essential, such as a clear beverage, a jelly, or a dairy product, it may be more economical and functional to focus exclusively on the core ingredient as a raw material. On the contrary, if a darker final product is acceptable or preferable, the peel may be a more effective source. This explains the strategic approach to valorizing various fruit by-products, which considers not only yield volume but also the quality and marketability of the end product.

3.3 | Functional Properties of Extracted Pectin

The functional properties of the marang pectin extracted from the peel and core using EAEs were evaluated in relation to WHC, OHC, SC, EC, and ES (Figure 4). The findings revealed significant differences depending on the tissue source (peel vs. core) and the pretreatment enzyme (cellulase, xylanase, or α-amylase).

3.3.1 | *WHC and OHC*

The lowest WHC was observed when peel pectin was extracted with cellulase (PC), whereas treatments with xylanase (PX) and, in particular, α-amylase (PA) resulted in significant WHC. The same trend was followed with OHC, with PA recording the highest oil retention among peel fractions. Core pectin tended to have higher WHC and OHC than peel, and cellulase and α-amylase showed the highest hydration and oil-binding properties. Such observations show that the enzymatic breakdown of structural polysaccharides (cellulose, hemicellulose, starch) increases porosity and exposes hydrophilic and hydrophobic domains, thereby enhancing pectin's capacity to hold water and interact with lipids (Vardhan et al. 2025). The WHC increased (12.81 g of water/g of pectin) in the xylanase treatment, likely due to the successful degradation of hemicellulose, which exposed more hydrophilic sites. On the same note, cellulase processing enhanced WHC in the core (9.93 g of water/g of pectin) and peel (10.61 g of water/g of pectin).

The α-amylase extraction showed moderate effects, with WHC values of 8.11 g of water/g of pectin in the core and 9.11 g of water/g of pectin in the peel, suggesting that starch hydrolysis does occur, although not as much as with fiber-degrading enzymes. Therefore, this supports the potential of marang fruit byproducts, particularly the peel and core, as functional ingredients in food systems that require high water retention. The pectin extracted by xylanase has the potential to improve WHC, suggesting the possibility of developing a natural hydrocolloid from underutilized tropical fruits. Moreover, it was determined that the OHC of the marang pectin studied was higher compared to that of pectin that was previously studied and obtained from Mada et al. (2022) and Ognyanov et al. (2018). This is due to the raw materials consumed and the extraction conditions used. It increases the likelihood of oil uptake by increasing the surface area of the introduced materials (pectin), increasing the thickness of the introduced materials (pectin), and increasing the hydrophobicity of the pectin (Mada et al. 2022).

3.3.2 | *EC and ES*

Emulsion properties of pectin are the ability to create the emulsion and apply as a stabilizer in the food product that enables the solubilization of the two immiscible liquids, or to maintain the stability of an emulsion (Pasandide et al. 2018). Figure 4c,d reveals the significant ($p < 0.05$) effect of the EC and ES of marang pectin. The EC of pectin from the peel is higher than that from the marang core, with peel-extracted pectin yielding 16.37% compared to core-extracted pectin at 9.8%.

On the other hand, ES is significantly higher ($p < 0.05$) with peel pectin extracted using xylanase, yielding 8.36% ES and a CX of 7.11%. This finding is notable because it indicates that both the source and the EAE are critical determinants of emulsifying properties. The emulsifying properties of pectin are still the subject of ongoing studies, but the most accepted mechanism is based on its amphiphilic properties and the ability to stabilize oil-in-water emulsions by adsorbing at the oil–water interface. There are also claims that the presence of a hydrophobic component within the hydrophilic pectin structure is the main factor enabling the formation of a stable emulsion. Acetyl groups also increase the hydrophobicity of the pectin molecule, which may have a considerable positive effect on its emulsifying capabilities, particularly in pectin with lower protein content (Shuai et al. 2022).

It has also been reported that pectin's molecular weight influences its emulsifying capacity. Low molecular weight pectin was more effective in the stabilization of small emulsion droplets compared to the high-molecular-weight pectin (Vanitha and Khan 2019). Therefore, the higher emulsifying capability and stability of peel pectin, especially in the PX and PA samples, indicate that the marang peel has a higher concentration of protein moieties and acetyl groups, which determine emulsification ability, compared to the core. This observation has highlighted that, to be used effectively in emulsifying applications, the best approach with marang pectin is to use the peel as the raw material and a non-conventional, mild extraction technique, such as enzyme-assisted pectin extraction.

3.3.3 | *SC*

The SC, as indicated in Figure 4e, can be considered to be similar in all the samples ($p > 0.05$), although those of the core and those of the peel pectin indicate slightly higher values. The SC is considered to depend on the fiber's structural characteristics and chemical orientation, which play a crucial role in water uptake. The SC property reflects the ability of pectin to take in water and swell to form viscous gels. Moreover, it provides the inflammatory content of the fiber lattice in case water is consumed (Mada et al. 2022). It is well known that the

chemical structure and composition of dietary fiber significantly influence water uptake. Studies revealed that dietary fiber could be affected by water in two ways, which included (i) water retained in capillary structures due to the capacity of surface tensions and (ii) water retained by hydrogen bonds and dipole forms (Huang et al. 2020). Thus, a higher amount of SC can be associated with high fiber, which can retain large amounts of water and oil present in food products (Mada et al. 2022) and can be applied to stabilize phase separation in yogurt.

3.4 | Solubility of Pectin

The solubility of pectin is determined by its degree of esterification, molecular weight, pH, temperature, and the presence of ions in the solution (Arias et al. 2021). In this study (Tables 4 and 5), peel-derived pectin was insoluble in cold water but soluble in hot water, whereas core-derived pectin had low solubility and often formed suspensions. These are characteristics of LM pectin from by-products of tropical fruits that have minimal refinement, inter-chain associations, and non-pectic contaminants interfere with hydration at ambient temperature (Arias et al. 2021). The inability to dissolve in cold water and propensity to form a suspension indicate that the pectin in the raw marang by-products is in the insoluble protopectin form. One of the main environmental factors that affects pectin solubility is temperature. Thus, extracted pectin can form lumps in cold water because the kinetic energy required for hydration and dispersion is insufficient (Ubieko et al. 2023). Pectin molecules in cold water are likely to form a net of hydrogen bonds with themselves and not with water. They may become less hydrated and less soluble as a result of reduced water exposure (Akhter et al. 2024). It is soluble in hot water because the pectin absorbs it, causing it to swell. The arrangement of pectin's hydrophilic and hydrophobic groups is what makes it soluble in hot water. The solubility may have resulted from hydrophilic groups attaching to water, as hydrophobic bonds weakened during the hot-water treatment (Akhter et al. 2024).

The solubility behavior of marang pectin under cold and hot alkali conditions can be used to understand its structural stability and its potential for use compared to commercial pectin sources. The results indicate that the peel and core of marang exhibit partial solubility in cold alkali, resulting in suspensions in alkali and swelling of the products when exposed to cellulase, xylanase, and α-amylase. In a cold alkali solution, pectin demethylates, forming a calcium pectate with yellow precipitation (Akhter et al. 2024). On the other hand, pectin exposed to hot alkali disintegrated into pieces and formed a milk-like substance. When pectin is hydrolyzed into smaller, soluble fragments via backbone hydrolysis in hot alkali, it takes on a milky appearance (Wehr et al. 2004), as observed in this study.

Overall, marang pectin is partially soluble in water, alkali, and acid solutions because the marang used in the study is a combination of ripe and overripe, as the goal is to utilize the peel

TABLE 4 | Solubility in cold and hot water from various enzymes.

Conditions	Marang by-products	Cellulase	Xylanase	α-Amylase	Commercial pectin (citrus)	Commercial pectin (apple)
Solubility in cold water	Peel	Insoluble, produce suspension	Insoluble, produce suspension	Insoluble, produce suspension	Partly soluble, produce suspension	Insoluble, produce suspension
	Core	Insoluble, produce suspension	Insoluble, produce suspension	Insoluble, produce suspension		
Solubility in hot water	Peel	Soluble	Soluble with/some insoluble part	Soluble	Soluble	Soluble
	Core	Soluble w/some insoluble part	Partly soluble, produce suspension	Partly soluble, produce suspension		

TABLE 5 | Solubility of marang pectin in cold alkali and hot alkali.

Conditions	Marang by-products	Cellulase	Xylanase	α-Amylase	Commercial pectin (citrus)	Commercial pectin (apple)
Changes in cold alkali	Peel	Partly soluble, produce suspension, swell	Soluble, w/ insoluble part, swell	Soluble, w/ insoluble part, swell	Soluble	Partly soluble
	Core	Partly Soluble, w/ insoluble part, swell	Soluble, w/ insoluble part, swell	Soluble, w/ insoluble part, swell	Soluble	Partly soluble
Changes in hot alkali	Peel	Partly Soluble, produce suspension	Partly soluble, produce suspension, swell	Soluble	Soluble	Soluble
	Core	Soluble w/suspension	Soluble w/ suspension	Soluble w/ suspension		

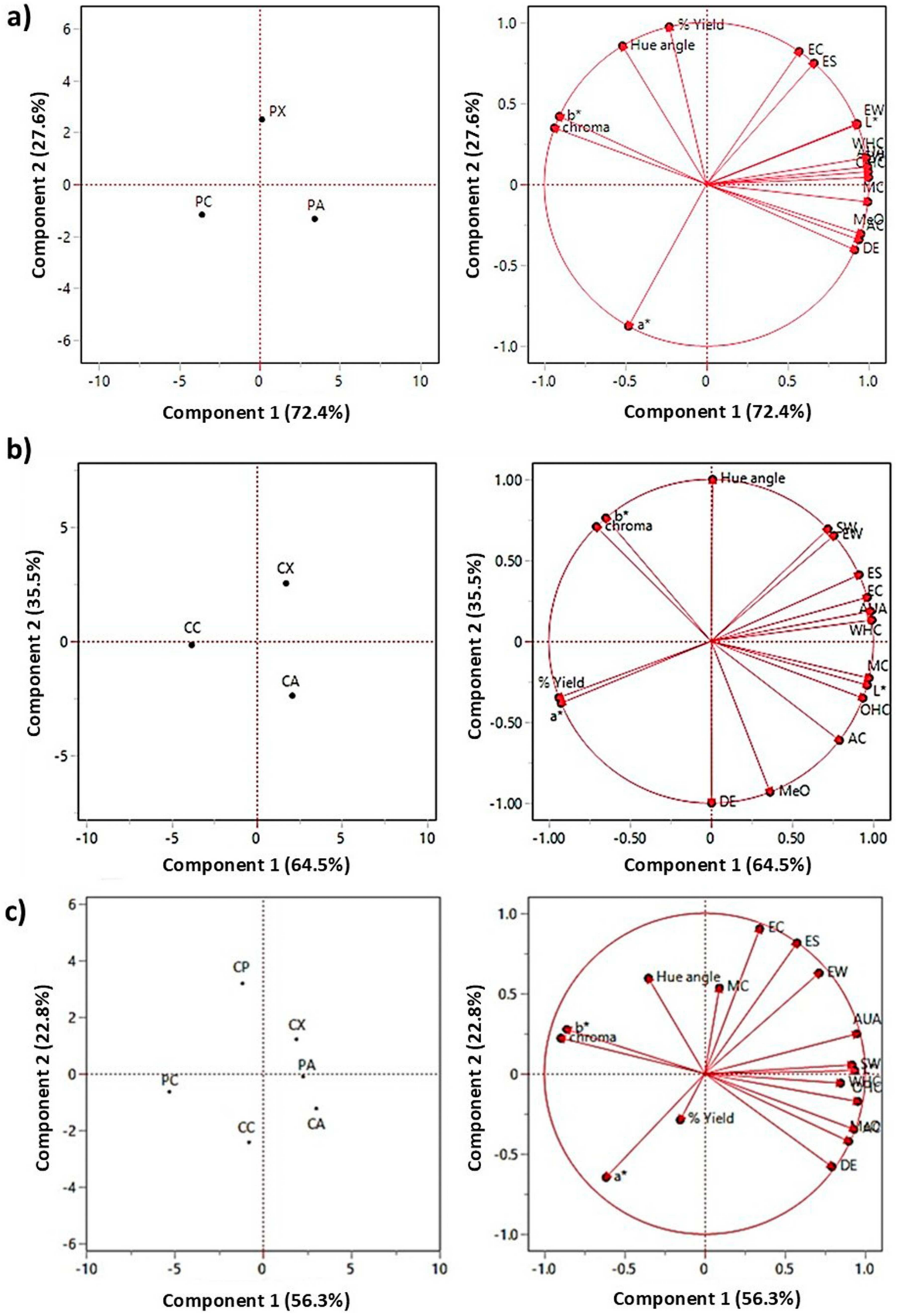


FIGURE 5 | Biplot of Principal Component Analysis (PCA) of marang pectin. (a) Effect of different enzyme extraction in marang peel pectin. (b) Effect of different enzyme extraction in marang core pectin. (c) Relationship of marang peel versus core with different enzyme extraction.

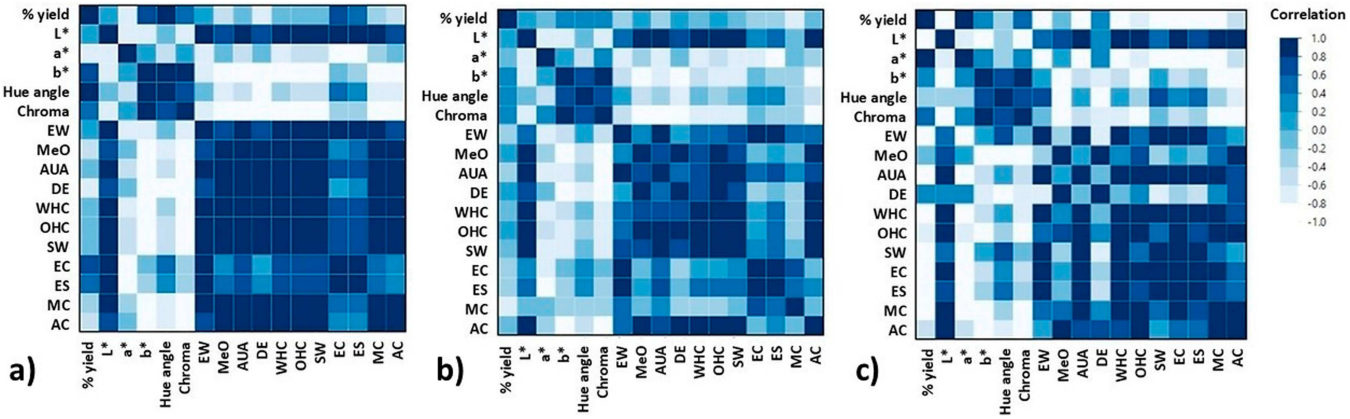


FIGURE 6 | Pearson's correlation analysis of the marang pectin. (a) Effect of different enzyme extraction in marang peel pectin. (b) Effect of different enzyme extraction in marang core pectin. (c) Relationship of marang peel versus core with different enzyme extractions.

and core waste after the flesh is consumed. As the fruit ages, pectin becomes more soluble. As the fruit ages, pectinolytic enzyme activity increases, resulting in greater pectin solubility. During fruit ripening, hydrolytic enzymes such as pectin methyl esterase and polygalacturonase break the long pectin chain into smaller units. Additionally, by breaking down the sugar chain in pectin, the enzyme β-galactosidase increases its solubility. Therefore, the amount of pectin in fruits decreases as they mature or ripen, but the solubility of the extracted pectin increases (Rahman et al. 2023).

3.5 | PCA of Pectin

The Principal Component Analysis (PCA) of pectin extracted from peel and core using different enzymes, based on % yield, physico-chemical properties, and functional properties, is depicted in Figure 5a–c. The PCA offers a summary of multivariate correlations among the pectin functional properties and the physico-chemical properties using enzyme-assisted extraction in marang peel (Figure 5a). The overall variance was explained by the two factors, which were 99.9%, with PC1 (72.4%) explaining most of the variability and PC2 (27.6%). This shows that there is a strong differentiation of enzyme-assisted extraction and the pectin properties along these two axes. The PC, PX, and PA were separated on the biplot with a clustering that indicates a variation of how the enzymes modify each of the pectin properties. The position of PC on component 1 was on the negative axis, and PX and PA were closer to the positive axis, which implies that cellulase extraction produced different compositional and functional properties as compared to xylanase and α-amylase. The variable loading plot indicates that the % yield, EC, ES, EW, WHC, OHC, MC, AC, MeO, and DE had a positive correlation along PC1. This implies that the pectin fractions obtained using xylanase and α-amylase had improved yield and functionality, including hydration and emulsification ability. The PCA of the enzyme-assisted extraction of pectin from the marang core is highlighted in Figure 5b. The first two components explain a total variance of 100% (PC1 = 64.5, PC2 = 35.5). The parameters of the functional properties and the AUA, MeO, DE, and EW were in great agreement along PC1, and there is an interdependence between them. The clustering of enzyme-assisted extractions was different in the score plot of PCA. Xylanase-extracted pectin was closer to the group of physico-chemical and functional properties, which means that it is effective in extracting high-quality pectin with improved functional properties. Pectin extracted with cellulase was in the intermediate position, which implies moderate extraction and functional properties. However, the α-amylase-extracted pectin was found to be further from the attributes, indicating a lack of high structural integrity and low functional properties. The PCA revealed the effect of various enzymes, cellulase (CC, PC), xylanase (CX, PX), and amylase (CA, PA), on the physico-chemical and functional characteristics of marang core- and peel-derived pectin (Figure 5c). The first two components accounted for 79.1% of the overall variance (PC1 = 56.3, PC2 = 22.8), showing clear groupings between core-derived and peel-derived pectin and the related quality parameters. The CC and CA were clustered on the negative side of PC1, associated with higher AC and lower functional properties. This suggests that enzymatic action in the core matrix, which is lignified and low in pectin-rich cell walls, may have led to partial solubilization but co-extraction of non-pectic residues (minerals, proteins), reducing functionality. The CX, however, was positioned closer to neutral, indicating moderate improvements in yield and structural integrity, consistent with xylanase's ability to disrupt hemicellulosic linkages surrounding pectin. Moreover, the biplot indicates a high positive correlation between DE, MeO, EW, and AUA, which are parameters representing the structural integrity of pectin. These properties were localized with peel-derived pectin, especially PC and PA, indicating that enzyme treatments did not interfere with ester formation but enhanced solubilization.

3.6 | Pearson's Correlation Analysis

The correlation heatmap of pectin extracted from marang peel using cellulase (PC), xylanase (PX), and α-amylase (PA) (Figure 6a) clearly shows relationships among yield, structural, and functional properties. Functional characteristics, including WHC, OHC, SC, EC, and ES, showed strong, positive correlations with % yield. This implies that greater pectin recovery was associated with enhanced hydration and emulsifying properties.

EAE likely increased the solubility of cell wall polysaccharides, producing pectin fractions with high water and oil affinity. The AUA, MeO, DE, and EW were also strongly positively correlated with functional properties. This supports the importance of the physico-chemical properties of pectin in determining the functional properties of pectin. Higher AUA pectin contributes to structural integrity, whereas DE and MeO determine its gelling mechanism (HM vs LM), which, in turn, determines its effects on food system hydration, emulsification, and stability. Color variables (L, a, b, hue angle, chroma) showed only weak or negative correlations with % yield and functional properties; the higher the pectin recovery and better functional quality, the darker the pectin. The correlation coefficients for the interest variable, % yield, were negative or low with DE and MeO, suggesting that a high peel extraction rate may release LM pectin fractions. The LM pectin has lower solubility but form stronger gels when calcium is present; thus, they are highly desirable in a given food application. This implies that cellulase, xylanase, and amylase extractions can equally enhance extraction performance, but the structural characteristics of the extracted pectin depend on enzyme selectivity. The correlation of marang core pectin using cellulase, xylanase, and α-amylase is presented in Figure 6b. There were also strong positive correlations in MeO, DE, and EW. These qualities were positively correlated with functional properties, including WHC, EC, and ES, indicating that galacturonic acid content and DE are significant in determining hydration and emulsifying properties. The results agree with previous reports that the galacturonic acid-rich pectin demonstrates strong gelling and stabilizing properties. The correlation heat map in Figure 6c shows the relationships among % yield, physicochemical parameters, and functional properties of pectin extracted using cellulase (CC/PC), xylanase (CX/PX), and α-amylase (CA/PA) from the marang peel and core. Clearly separated variable clusters highlight the roles of enzyme type and marang by-products (core vs. peel) in determining pectin quality and performance. The AUA, DE, MeO, and EW were positively associated with pectin yield, especially in peel-derived fractions. This implies that solubilization of homogalacturonan regions by enzymatic hydrolysis, particularly with xylanase and amylase (PA), increased galacturonic acid content while maintaining esterified pectin. Core-derived pectin (CC, CX, CA), on the other hand, showed weaker correlations between yield and physicochemical properties, suggesting that the peel was a preferable substrate for EAE because it is a richer pectin matrix. The moisture and ash content had a positive correlation, especially in pectin fractions of the core, indicating a higher concentration of bound materials and hydrophilic residues. These parameters have been inversely correlated with functional properties such as EC and WHC, indicating that higher mineral residues may reduce the pectin's ability to interact with oil-water interfaces. The WHC, OHC, and SC were found to be clustered tightly with EC and ES. This interdependence implies that the hydration characteristics of pectin profoundly determine its emulsification properties, as water-binding polysaccharides emulsify by increasing viscosity and forming interfacial films. Peel-derived pectin, especially that extracted using xylanase and α-amylase, displayed better correlations between these functional properties and thus better than core-derived pectin. Compared with the core (CC, CX, CA), the peel (PC, PX, PA) consistently produced better results, yielding pectin with high positive correlations among % yield, AUA, DE, and functional properties. This indicates that the EW, MeO, AUA, and DE of marang peel have higher pectin-associated hemicellulose content, along with cellulose, which is readily broken down by xylanase and cellulase. The most favorable pattern was observed for PX, which produced higher pectin yield, DE, MeO, and EC. Xylanase destabilizes the hemicellulose linkages, releasing pectin without the excessive de-esterification. The effect of α-amylase (PA) on % yield and functional properties was also lower, indicating that starch-related polysaccharides in the peel are involved in the release of pectin. In comparison, core-derived cellulase (CC) produced pectin with higher mineral content and weaker functional properties, probably because of the limited accessibility of pectin domains within the lignified core matrix.

4 | Conclusions

The current study demonstrated that the peel and core waste of marang (*A. odoratissimus* Blanco) have potential use as underutilized fruit byproducts for the EAE of LM pectin, specifically when cellulase, xylanase, and α-amylase are used. Among the treatments, the highest yield was obtained from cellulase extraction of the marang core, whereas xylanase improved the emulsifying properties of the peel. The α-amylase increased the purity of the end product pectin by hydrolyzing the starch impurities, resulting in higher EW pectin and higher AUA content. These results demonstrate the critical role of enzyme selectivity in maximizing pectin recovery from underutilized fruit byproducts. The marang-derived pectin was classified as low-methoxyl (DE < 50%) pectin that allows calcium-induced gelation and may be helpful in food applications requiring gelation, thermal stability, and low sugar content. Evaluation of the functional properties of the core and peel showed that core pectin was able to hydrate more fully and swell more than the peel, while the peel pectin was able to emulsify more than the core. These functional properties of marang pectin indicate that it is suitable for several industries, spanning nutraceuticals, dairy, beverages, and pharmaceuticals. Despite the positive outcomes, some physicochemical attributes, including ash content, exceeded industry standards; thus, further improvement and purification techniques are needed. Moreover, optimizing EAE conditions could improve pectin purity and consistency. The current study highlights the potential use of marang fruit waste as a functional hydrocolloid. It converts underutilized biomass into a high-value product, thereby helping reduce waste, optimize resource use, and increase benefits in the circular economy. With ongoing improvements, pectin from marang fruit could also serve as a substitute for commercial-grade pectin, thereby strengthening food innovation ideas while supporting environmental preservation.

Author Contributions

Kenneth Tumilap: investigation, validation, visualization, writing – review and editing, writing – original draft, formal analysis, methodology. **Loren Mae D. Ranario:** investigation, writing – original draft, writing – review and editing, visualization, validation, methodology,

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Conflicts of Interest

The authors declare no conflicts of interest.

Data Availability Statement

The data that support the findings of this study are available from the corresponding author upon reasonable request.

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