Whole-Exome Sequencing and Targeted Copy Number Analysis in Primary Ciliary Dyskinesia

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ABSTRACT Primary ciliary dyskinesia (PCD) is an autosomal-recessive disorder resulting from loss of normal ciliary function. Symptoms include neonatal respiratory distress, chronic sinusitis, bronchiectasis, situs inversus, and infertility. Clinical features may be subtle and highly variable, making the diagnosis of PCD challenging. The diagnosis can be confirmed with ciliary ultrastructure analysis and/or molecular genetic testing of 32 PCD-associated genes. However, because of this genetic heterogeneity, comprehensive molecular genetic testing is not considered the standard of care, and the most efficient molecular approach has yet to be elucidated. Here, we propose a cost-effective and time-efficient molecular genetic algorithm to solve cases of PCD. We conducted targeted copy number variation (CNV) analysis and/or whole-exome sequencing on 20 families (22 patients) from a subset of 45 families (52 patients) with a clinical diagnosis of PCD who did not have a molecular genetic diagnosis after Sanger sequencing of 12 PCD-associated genes. This combined molecular genetic approach led to the identification of 4 of 20 (20%) families with clinically significant CNVs and 7 of 20 (35%) families with biallelic pathogenic mutations in recently identified PCD genes, resulting in an increased molecular genetic diagnostic rate of 55% (11/20). In patients with a clinical diagnosis of PCD, whole-exome sequencing followed by targeted CNV analysis results in an overall molecular genetic yield of 76% (34/45).

Primary ciliary dyskinesia (PCD, MIM#244400) is a genetically heterogeneous, autosomal-recessive motile ciliopathy. Clinical manifestations include situs inversus, neonatal respiratory distress, progressive bronchiectasis, and respiratory failure in young adulthood. To date, there are 32 genes associated with PCD that account for a genetic diagnosis in approximately 65% of all patients with PCD (Kurkowiak et al. 2015).

The clinical diagnosis of PCD is challenging and relies on patient features and diagnostic investigations. These include diagnostic imaging, nasal nitric oxide levels, cilia beat frequency, and ciliary ultrastructure analysis by electron microscopy (EM) (Leigh et al. 2011). Historically, a diagnosis of PCD has relied on the findings of a classic ciliary ultrastructural defect on EM (Barbato et al. 2009). EM of normal motile cilia consist of nine peripheral microtubule doublets encircling a central microtubular complex supported by microtubular-associated proteins such as outer and inner dynein arms (ODA, IDA, respectively), radial spokes, and nexin links (Knowles et al. 2013a).

A molecular genetic diagnosis of PCD can be established through the identification of biallelic loss-of-function mutations in a PCD-associated gene. Mutations in certain PCD genes result in ciliary
dysfunction and can give a specific ultrastructural defect on EM (Knowles et al. 2013a). ODA defects are the ultrastructural defect observed most frequently in PCD and are associated with mutations in DNAH5, DNAI1, DNAI2, DNAL1, TXNDC3, CCDC114, and ARMC4 (Lobo et al. 2015). Defects in both ODA and IDA are seen in patients carrying mutations in LRRC50/DNAAF1, KTU/DNAAF2, DNAAF3, CCDC103, HEATR2, LRRC6, ZMYND10, DXY1C1, C21orf59, and SPAG1. IDA defects with microtubular disorganization are seen with mutations in CCDC39 and CCDC40. Central microtubular abnormalities are observed with mutations in RSPH1, RSPH4A, and RSPH9. Patients with mutations in CCNO and MCIDAS have ciliary aplasia/oligoplasia and a marked reduction of cilium (oligocilia) due to a deficiency in ciliogenesis rather than motility. In addition, patients with mutations in DNAH11, HYDIN, CCDC164/DRC1, and CCDC65/DRC2 do not have obvious ciliary ultrastructural defects and would not be diagnosed with PCD on the basis of biopsy EM alone. We have shown previously that molecular genetic analysis can complement ciliary ultrastructure analysis and increase the overall diagnostic yield from 57% (28/49) to 69% (36/52) (Kim et al. 2014). However, the molecular genetics of PCD are complex and not considered routine in the diagnostic evaluation of patients (Barbato et al. 2009; Bush and Hogg 2012). Modern advances in molecular genetic technologies may overcome some of these perceived limitations in a cost-effective manner.

In genetically heterogeneous diseases such as PCD, selecting the appropriate gene and technique for molecular analysis is often difficult. Sanger sequencing is available for many, but not all, of the 32 PCD-associated genes. A step-wise approach by prioritizing genes based on ciliary ultrastructure and prevalence of mutations could be conducted using Sanger sequencing (Bush and Hogg 2012; Kim et al. 2014). However, as the number of genes tested increase, Sanger sequencing can become costly and time-consuming. In PCD, many new loci are being discovered on an ongoing basis, quickly out-dating existing available panels. Moreover, newly discovered loci account for a minority of patients with PCD (Knowles et al. 2013a), decreasing the diagnostic value for each gene added to a conventional panel.

Intron-exon level copy number variations (CNVs) are seen in up to 10.8% of autosomal-recessive Mendelian disorders and are not detectable by Sanger sequencing (Aradhya et al. 2012). Deletions in SPAG1, ARMC4, DXY1C1, LRRC50, and ZMYND10 have been observed in isolated PCD cohorts (Loges et al. 2009; Blanchon et al. 2012; Hjeij et al. 2013; Knowles et al. 2013b; Tarkar et al. 2013; Zarivala et al. 2013), suggesting CNV analysis should be used in the molecular evaluation of patients with PCD. However, the prevalence and clinical significance of CNVs in other PCD-associated genes have not been examined.

Whole-exome sequencing (WES) has been used extensively for gene discovery and is beginning to be used in clinical practice in the diagnosis of genetically heterogeneous diseases (Neveling et al. 2013). The advantages of WES include providing a genomic approach in molecular genetic diagnoses through the analysis of all potential causative genes, including recently discovered genes not available on clinical gene panels (Boycott et al. 2015). Furthermore, this strategy allows for more complete interpretation of variants of uncertain significance (VUS), and can guide further molecular genetic analyses such as targeted CNV analysis. Because of ongoing gene discovery in genetically heterogeneous diseases such as PCD, WES may be a more cost-effective technique (Lucas et al. 2014; Kurkowiak et al. 2015). Here, we describe the complementary role of WES and targeted CNV analysis in solving cases of patients with PCD in a cost and time-effective manner.

MATERIAL AND METHODS

Study subjects

Between the period of August 2011 and December 2012, Sanger sequencing of 12 PCD genes was clinically available and conducted on 45 families with a clinical diagnosis of PCD (Kim et al. 2014). A total of 19 families (42%) were found to have biallelic pathogenic mutations in these 12 PCD genes and thus confirmed a molecular diagnosis. Pathogenic mutations were defined as mutations previously documented in patients with PCD; or nonsense, frameshift and splice-site mutations resulting in loss-of-function. Four families were solved in other research studies (Supporting Information, Table S1) leaving 22 families unsolved. Twenty families consented to have CNV analysis and/or WES. This study was approved by the Research Ethics Boards of the Hospital for Sick Children and St Michael’s Hospital. Study subjects provided written consent and/or assent where appropriate.

Molecular genetic analysis algorithm

A molecular genetic analysis algorithm was developed to maximize previous molecular genetic data and minimize time and cost. Individuals were divided into three categories depending on the information from previous Sanger sequencing and WES performed in this study (Figure 1). For the individuals who harbored single pathogenic mutations after Sanger sequencing, targeted CNV analysis of the putative causative gene was conducted to ascertain the second allele (category A). Individuals with no pathogenic mutations after Sanger sequencing underwent WES to analyze the remaining 20 genes not covered by Sanger sequencing (category B). WES-sequenced individuals who were not found to have any pathogenic mutations in a PCD gene, further analysis of missense variants occurring at a minor allele frequency of <1% was conducted. Rare missense variants were considered to be contributory if predicted to be pathogenic, disease causing or damaging by in silico prediction programs (MutationTaster, PolyPhen-2, SIFT). These individuals underwent further targeted CNV analysis on the suspected PCD gene (category C).

Targeted CNV analysis

The selected technique for targeted CNV analysis was based on previously reported intron−exon CNVs in the literature. Exon 7 deletions in DXY1C1 have been documented in other PCD cohorts (Tarkar et al. 2013), and a custom TaqMan copy number assay was designed to detect this specific CNV (File S1). Reported deletions of exon 62 of DNAH5 (Berg et al. 2011) along with the remaining 78 exons were assayed using a commercially available, high-density gene-centric array comparative genomic hybridization (CGH; Prevention Genetics, Marshfield, WI). Similarly for DNAH11, where intron−exon level CNVs have not been reported, CGH was used to assay all 82 exons.

WES and validation

WES was completed with the Illumina Hiseqution 2500 platform at The Centre for Applied Genomics (TCAG) at the Hospital for Sick Children following whole-exome capture with the Agilent SureSelectXT Human All Exon V4 capture kit. Sequence reads were aligned to the reference human genome (hg19) with Burrows-Wheeler Aligner 0.5.9. MarkDuplicates (Picard tools, version 1.79; http://broadinstitute.github.io/picard/) was used to remove any duplicate paired-end reads. Duplicate-free alignments were refined using base
space local realignment and quality score recalibration using GATK 1.128. Mean depth of coverage was 138X (range 100 – 168X) with all cases having >95% of targeted bases sequenced to a depth of greater than 20X.

Single-nucleotide variants (SNVs) and insertion/deletions (indels) were called with default parameter in GATK 1.1.28. SNVs and indels were annotated using SNPEff (http://snpeff.sourceforge.net/) and ANNOVAR and filtered to differentiate novel variants from known polymorphism databases by screening against public single nucleotide polymorphism databases (dbSNP, http://www.ncbi.nlm.nih.gov/projects/SNP/; 1000 genomes, www.1000genomes.org; NHLBI Exome Sequencing Project (ESP) Exome Variant Server http://evs.gs.washington.edu/EVS/) and our own internal database consisting of 283 exomes analyzed in the same manner at TCAG. Novel SNVs also were annotated with SIFT, PolyPhen-2, and MutationTaster to assess the putative variant effect on the proteins. SNVs were prioritized based on loss of function mutations (nonsense, frameshift, splice sites) and damaging missense mutations that fit an autosomal recessive disease model.

An in silico gene panel was created by filtered analysis of WES data. WES data were reanalyzed with the publication of new PCD genes and the in silico panel expanded in real-time. At the time of manuscript submission, 32 genes were analyzed for variants (DNAH5, DNA11, CCDC39, CCDC40, DNA2, Dnah11, RSPH9, RSPH4A, KTU/DNAAF2, LRRCS5/DNAAF1, TXNDC3/NEB, DNA11, DNAAF3, CCDC103, HEATR2, HYDIN, LRRCS6, CCDC114, CCDC164/DRC3, ARMCA, DYS1C1, ZMYND10, CCDC65/DRC2, RSPH1, OFD1, RPGR, C21orf59, SPAG1, DNAH8, CCDC151, MGIDAS, and CCNO). WES coverage of these genes was adequate with >98% of targeted bases sequenced and 92.08% (616/669) of exons having a mean coverage of >20X. Confirmation of variants was conducted on probands and available family members using standard Sanger sequencing methods in a CLIA-CAP diagnostic laboratory and genetic counseling was provided based on these results (Table S2).

**RESULTS**

Four individuals from three families harbored one pathogenic mutation after Sanger sequencing (category A). Two families (111, 113) each harbored two different nonsense mutations in DNAH5 and CNV analysis of DNAH5 was conducted using array CGH to ascertain the second allele. Two clinically significant CNVs in DNAH5 were found and likely causative second allele, corresponding to the ODA defect observed on ciliary EM (Table 1). The third family harbored a previously reported nonsense mutation in DNAH11, c.6898C > T (p.R2900*) (Lucas et al. 2012). However, DNAH11 CNV analysis with array CGH did not detect a CNV and this case remained unsolved.

A total of 18 individuals from 17 families who did not have any pathogenic mutations detected on initial Sanger sequencing (category B and C) underwent WES. Seven category B families were solved by WES alone where two pathogenic variants were found in one of the additional 20 PCD genes not analyzed by Sanger sequencing (Table 1). Molecular genetic results were congruent with ultrastructural analysis when available. For those cases with available family members for segregation analysis, phase was determined to be trans.

WES on individual 46 revealed a splice site (c.13680-1G > T) and frameshift mutation, c.4866del(p.P1623Qfs*20) in HYDIN. Because of the presence of a pseudogene on chromosome 1 (HYDIN2), further characterization of both variants was conducted using Sanger sequencing (Olbrich et al. 2012) (File S1and Figure S1). Primers mapping exclusively to HYDIN on chromosome 16 were designed (Table S2) which further resolved the next generation data from WES. The frameshift was found to be homozygous, and likely the causative allele. The splice site variant was heterozygous, but its pathogenicity is uncertain. Determination of segregation would help further resolve these variants, however, family members were not available. Ciliary biopsy of this individual was normal, as seen in other individuals with HYDIN mutations, further supporting the pathogenicity of the frameshift variant (Olbrich et al. 2012).

Individuals who did not have biallelic pathogenic mutations after WES were reanalyzed for rare missense variants of uncertain significance in the 32 known PCD genes. Five individuals from five families were found to harbor such VUS. Individual 44 was found to have a homozygous missense c.1555G > C (p.A519P) variant in DNAAF3 (Table 2) on WES, which was not reported in other PCD patients nor present in our control databases. This transversion resulted in an amino acid change which is predicted to be tolerated by SIFT and MutationTaster, and only probably damaging by Polyphen-2. DNAAF3 mutations are associated with ODA + IDA defects, not seen in this patient. We concluded this variant was not likely contributing to this patient’s PCD phenotype and this case remained unsolved.
Table 1: Biallelic pathogenic variants in known PCD genes through CNV and WES analysis

| Family | Patient | Ethnicity | Sex | Dx  | Age | Ciliary EM | nNO | nL/min | Gene | hg 19 Genomic Coordinates | Transcript | Exon/Intron | Base Changes | Predicted Effect | Segregation |
|--------|---------|-----------|-----|-----|-----|------------|-----|---------|------|----------------------------|------------|-------------|--------------|-----------------|-------------|
| 111    | 4       | White     | M   | 4   | yr  | I          | ODA | 8.1     | DNAH5 | Chr5: 13701398              | NM_001369.2 | Ex 77       | c.13486C > T | p.R4496*        | (1) Unknown |
|        |         |           |     |     |     |            |     |         | DNAH5 | Chr5: 13816665-13821065     | NM_001369.2 | Int 42       |              |                 |             |
|        | 113     | Portuguese| F   | 6   | yr  | A          | ODA | 22      | DNAH5 | Chr5: 13865895              | NM_001369.2 | Ex 27       | c.4237C > T | p.Q1413*        | Maternal    |
|        |         |           |     |     |     |            |     |         | DNAH5 | Chr5: 13931761-13932769     | NM_001369.2 | Int 2        |              |                 |             |
|        | 113     | Portuguese| M   | 14  | yr  | S          | ODA | 8.9     | DNAH5 | Chr5: 13865895              | NM_001369.2 | Ex 27       | c.4237C > T | p.Q1413*        | Maternal    |
|        |         |           |     |     |     |            |     |         | DNAH5 | Chr5: 13931761-13932769     | NM_001369.2 | Int 1        |              |                 |             |
|        | 115     | East Indian| M   | 7   | yr  | S          | Inadequate | 37.5 | LRRRC6 | Chr8: 133645009           | NM_0124724 | Ex 5         | c.630delG    | p.W210Cfs*12 | (2) Maternal |
|        | 115     | East Indian| M   | 9   | yr  | S          | ODA+IDA  | 5.8    | LRRRC6 | Chr8: 133645009           | NM_0124724 | Ex 5         | c.630delG    | p.W210Cfs*12 | (2) Maternal |
|        | 141     | Pakistani  | F   | 5   | yr  | I          | Inconclusive | 17.1 | SPAG1  | Chr8: 101226130           | NM_0031144 | Ex 12        | c.1519dupA   | p.IS01Nfs*5    | Unknown     |
|        | 140     | Portuguese| F   | 8   | yr  | I          | Not done | 6.9    | ARM4C | Chr10: 28276450            | NM_018076.2 | Ex 3         | c.2474del    | p.E83*        | Unknown     |
|        |         |           |     |     |     |            |         |        |               |              |             |              |                 |             |
| 135    | 39      | Pakistani  | F   | 12  | yr  | S          | Oligocilia | 22.1  | CCNO   | Chr15: 54527295           | NM_0211473 | Ex 3         | c.961C > T   | p.Q321*        | (3) Trans    |
| 138    | 46      | White      | F   | 35  | yr  | S          | Normal   | 53.2   | HYDIN  | Chr16: 70084800           | NM_01270974.1 | Ex 32        | c.4866del    | p.P1623Qfs*20 | Unknown     |
|        |         |           |     |     |     |            |         |        |               |              |             |              |                 |             |
| 120    | 24      | White      | M   | 2   | mo | I          | ODA+IDA  | 11.2   | ZMYND10 | Chr3: 50382964            | NM_015896.2 | Ex 1         | c.47T > G    | p.V16G*        | (4) Maternal |
|        |         |           |     |     |     |            |         |        |               |              |             |              |                 |             |
| 137    | 45      | Hispanic   | F   | 30  | yr  | S          | Inadequate | 17.1  | RSPH1  | Chr21: 43913159           | NM_080860.3 | Ex 2         | c.85G > T    | p.E29*        | Unknown     |
|        |         |           |     |     |     |            |         |        |               |              |             |              |                 |             |

Genomic coordinates are approximate for copy number variations. Further clinical characteristics previously described (Kim et al. 2014). Previously described mutations are in **bold** (1) (Homef et al. 2006); (2) (Zariwala et al. 2013); (3) (Wallmeier et al. 2014); (4) (Moore et al. 2013); (5) (Knowles et al. 2014). PCD, primary ciliary dyskinesia; CNV, copy number variation; WES, whole-exome sequencing; Dx, diagnosis; EM, electron microscopy; nNO, nasal nitric oxide; M, male; I, situs inversus; ODA, outer dynein arms; F, female; A, situs ambiguous; S, situs solitus; inadequate = inadequate ciliated cells to analyze, IDA, inner dynein arms inconclusive EM; inconclusive = adequate sample.
The four other individuals were found to have rare missense VUS predicted to be damaging, disease-causing, or deleterious by at least one of the in silico prediction programs, MutationTaster, PolyPhen-2 and SIFT. These individuals underwent targeted CNV analysis to ascertain a second hit (category C, Figure 1).

Individual 9 was initially found to have a single variant in DNAH5 (c.3890A > G; p.D1297G) that was predicted to be disease-causing by Poly-Phen, SIFT, and MutationTaster and ciliary ultrastructure was interpreted as ODA (Kim et al. 2014). However, array CGH analysis of all exons of DNAH5 did not yield a second allele, prompting further review of WES data. In addition to the rare variant in DNAH5, a single rare missense variant in DYX1C1 (c.988C > T; p.R330W) was found in WES data. This variant was not observed in control populations and was also predicted to be disease causing by SIFT and MutationTaster. Targeted CNV analysis by TaqMan copy number assay of a previously reported exon 7 deletion in DYX1C1 was conducted and was found in this individual (Tarkar et al. 2013). Subsequent segregation analysis confirmed trans orientation of the rare missense variant and deletion. This prompted further pathologic review and revision of the initial ciliary EM to ODA + IDA consistent with other DYX1C1 families (Table 2) (Tarkar et al. 2013), suggesting this is the causative gene in this family.

Individual 21 was found to have a rare missense VUS (c.10285C > A) in DNAH11 on initial Sanger sequencing and WES confirmed this VUS. MutationTaster, PolyPhen-2 and SIFT algorithms predict the p.R3429L change to be deleterious, probably damaging and damaging respectively. Additionally, this is a highly conserved amino-acid residue residing in the conserved Helix 2 domain (Schwabe et al. 2008). Array CGH analysis on DNAH11 was conducted and demonstrated a 32.29-kb duplication spanning exons 7–14 and is likely the causative second allele in this individual (Table 2).

Individual 51 was found to have two previously unreported homozygous missense variants in DNAH11, which also were detected through WES. One variant, c.10286G > T (p.R3429L) involves the same highly conserved amino-acid residue in the Helix 2 domain observed in individual 21. The other variant, c.2750A > T (p.E917V) also was predicted to be possibly damaging by Polyphen-2, deleterious by SIFT, and deleterious by MutationTaster. Subsequent array CGH of DNAH11 analysis did not reveal a deletion or duplication and this case remained unsolved (Table 2).

Patient 38 was heterozygous in exon 82 of the DNAH11 gene for an undocumented missense variant defined as c.13366C > T and predicted to result in p.R4449C substitution. The SIFT and Polyphen-2 protein function algorithms predicts this change to be tolerated and benign, whereas the MutationTaster program indicates that this c.13366C > T variant is deleterious. However, DNAH11 array CGH did not yield a second allele de-prioritizing this variant leaving this case unsolved (Table 2).

Through Sanger sequencing of the first 12 PCD genes, we found that five individuals who had a clinical diagnosis of PCD did not harbor a disease-causing mutation in a known PCD gene, nor did we find, using WES, a pathogenic mutation or rare missense VUS in any of the known PCD genes (Table S3). We suspect these patients either have a mutation not detectable by either technique or a novel PCD locus.

Through the combination of targeted CNV and WES analysis allowed for a molecular diagnosis in 55% of our unsolved families (11/20). Clinically significant CNVs were detected in 8.8% (4/45) of our total patient population, which is consistent with other autosomal-recessive disorders (Aradhya et al. 2012). Of the 45 families, Sanger sequencing solved 19 (42%) families whereas subsequent targeted

### Table 2 Variants of uncertain significance in known PCD genes through CNV and WES analysis

| Family | Compound heterozygous, likely pathogenic (in silico) |
|--------|---------------------------------------------------|
| 114    | DNAH11 (c.13366C > T)                             |
| 114    | DNAH11 (c.10285C > A)                             |
| 114    | DNAH11 (c.2750A > T)                              |
| 114    | DNAH5 (c.988C > T)                                |

| Family | Compound heterozygous, likely pathogenic (in silico) |
|--------|---------------------------------------------------|
| 114    | DNAH11 (c.13366C > T)                             |
| 114    | DNAH11 (c.10285C > A)                             |
| 114    | DNAH11 (c.2750A > T)                              |
| 114    | DNAH5 (c.988C > T)                                |
CNV analysis with WES solved an additional 11, yielding an overall diagnostic rate of 30/45 (67%). Four further families were enrolled in other studies, such that in our cohort, 34/45 (76%) had a molecular diagnosis of PCD. As all variants detected by Sanger sequencing were covered in our WES data, if our molecular genetic approach was modified to WES followed by targeted CNV analysis, the overall diagnostic rate would approach 76%.

DISCUSSION

In genetically heterogeneous diseases such as PCD, a step-wise molecular genetic approach has been proposed based on ciliary ultrastructure and the prevalence of certain mutations. If genes are assayed in a cost-efficient, step-wise tiered fashion, only one gene may be assayed at one time, taking up to 4–6 months to complete, costing up to $12,000 USD. In addition, expert EM ciliary ultrastructure analysis may not be available to guide such tiered analysis. Newly developed next-generation sequencing (NGS) targeted panels are less costly, at $1900–$4200 USD, but do not include all 32 PCD genes (Prevention Genetics; Ambry Genetics, Aliso Viejo, CA; Center for Genomics and Transcriptomics, Tübingen, Germany). However, unlike tiered Sanger sequencing, NGS concurrently assays all genes. If a patient had previous genetic testing, these genes would be unnecessarily retested. Furthermore CNV data from NGS is unreliable and not possible with Sanger sequencing.

Here, we describe the utility of filtered WES data and targeted CNV analysis to circumvent the limitations of targeted Sanger and NGS panel sequencing in PCD. Because of the cost-efficiency of WES ($2–$5000 USD) (Neveling et al. 2013), we propose that WES could be conducted as a first-line molecular diagnostic test through the analysis of an in silico gene panel, followed by targeted CNV analysis. In addition to providing a definitive molecular diagnosis when biallelic pathogenic mutations are found, WES provides additional information on all PCD genes and aids in the interpretation of rare missense VUS. In the absence of convincing pathogenic mutations in all known PCD genes, rare missense VUS could result in loss of function and potentially be disease-causing prompting further targeted CNV analysis. CNV analysis should be targeted to the candidate PCD-associated gene on the basis of sequencing results. CNV analysis initially could begin with CNVs reported other PCD cohorts, followed by more extensive full exon analysis using array CGH or multiplex ligation-dependent probe amplification (Stuppia et al. 2012). Further studies are required to determine if the novel CNVs reported in our cohort are private mutations or common to other PCD patients. In addition, as CNV algorithms from WES data improve, they may replace other CNV analyses and provide a genome-wide CNV approach (Samarakoon et al. 2014). If a PCD case remains unsolved after WES and targeted CNV analysis, WES data can be further analyzed as novel PCD loci become published expanding the in silico panel in real-time, instead of expending further sequencing consumables. In our population, if WES was followed by targeted CNV analysis, 76% of patients with PCD would have had a molecular genetic diagnosis and WES should be considered the most cost-efficient molecular genetic technique in such genetically heterogeneous disorders.

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