Causality of Hematology Sample Rejection: A Training Needs Assessment for Health Facilities

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Original Research Article

ABSTRACT

A cross sectional study was conducted to assess pre-analytical variables and sources of errors observed among hematology specimen received from IPD area in Central Pathology Laboratory of NKP Salve Institute of Medical Sciences & Research Centre and Lata Mangeshkar Hospital, Nagpur, Maharashtra during July 2016 - June 2017. Rate of sample rejection was noted to be highest from Orthopedics followed by Obstetrics and Gynecology, Medicine, Pediatric, Surgery, Eye, ENT, Pulmonary & Dermatology. The commonest cause for rejection was clotted samples. Other causes were hemolyzed sample, inadequate sample, excessive delay in sample transport and wrong patient identification. Faulty phlebotomy techniques, inappropriate preservation, improper transportation, wrong patient identification, lesser efficiency and carelessness were identified as other reasons.

KEY WORDS: hematology, phlebotomy, rejection, training needs assessment (TNA)

INTRODUCTION:

The clinical diagnosis is largely dependent, these days, on reliable laboratory data. Hence, there is major improvement in the laboratory performances augmented by advances in sample collection, transport, automation and dispatch of reports[1]. Medical laboratories play vital role in the decision making by physicians. About 60-70% of clinical decisions regarding admission, prescription, and discharge are based on laboratory results. Since these play a significant role, the quality of laboratory test results are important[2]. However, errors can occur in any phase during the processing of blood sample. The errors in laboratory practice are classified into pre-analytical, analytical, and post analytical phase depending on the time of presentation[3,5]. An important component of laboratory medicine is pre-analytical phase[5]. The pre-analytical phase comprises of all the processes occurring before the sample being actually processed in the laboratory[6]. It includes specimen collection, handling and processing variables, physiological variables, and endogenous variables. Certain pre-analytical variables, namely, specimen variables can be controlled; whereas knowledge of uncontrollable variables needs to be well understood in order to separate their effects from disease related changes affecting laboratory results[4,7]. The reported types of pre-analytical error are ordering tests on the wrong patient, misidentifying the patient, ordering the wrong test, missing sample and/or test request, wrong or missing identification, contamination from infusion route, hemolyzed, clotted, and insufficient samples, inappropriate containers, improper labeling of containers, inappropriate blood to anticoagulant ratio, and inappropriate transport and storage conditions[8,9]. The laboratories have to bear the burden of the inconsistencies or incorrect reporting that is because of these pre-analytical errors[6]. Analytical errors can be minimized with the recent advancement in technology and introduction of automation in hematology laboratories provided good quality control practices are followed[10]. In spite of automation in hematology and clinical pathology, there are many variables which can influence the laboratory results[11].

The Central Pathology Laboratory (CPL) is routinely functional for 24 hours throughout the year. Properly collected blood sample is essential for quality performance by the Laboratory. Hematology testing is performed on whole blood. Hence, the laboratory data & reliability entirely depends on submitted samples if they are adequate, labeled, and properly transported to the laboratory with in time as per the required protocol. Therefore, the present study was undertaken to assess

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Table 1 : Rejected Specimen Ratio With Respective Department.

SNO	MONTH	REJECT	MED	OBGY	SURG	OPTH	ORTH	ENT	PAED	PSY	TB	SKIN
1	Jul	85	2	20	21	4	15	6	12	3	2	6
2	Aug	85	3	21	22	5	16	6	11	2	1	4
3	Sep	78	3	16	18	4	15	6	13	2	1	6
4	Oct	88	1	14	18	6	13	3	14	2	0	2
5	Nov	72	2	15	20	4	14	5	12	2	2	3
6	Dec	65	1	13	16	4	15	6	14	2	1	8
7	Jan	71	1	16	18	5	14	6	10	2	0	6
8	Feb	72	1	17	20	4	15	5	9	1	1	4
9	Mar	78	1	18	20	6	13	3	8	2	2	8
10	Apr	80	2	16	18	4	14	2	12	4	2	8
11	May	84	2	18	26	6	15	6	12	3	2	10
12	Jun	98	3	19	23	4	16	4	16	3	2	10
13	TOTAL Rejection	956	22	203	240	56	175	58	143	28	16	75
14	REJECT (%)	1.75	2.43	3.02	2.2	1.03	3.6	1	2.6	0.08	0.6	0.26
15	Total recived sample	30153	915	6745	5692	1843	3826	2092	4973	1278	498	2291

* Total recived sample by Randomize collection i.e. 10% of total aggregate sample.

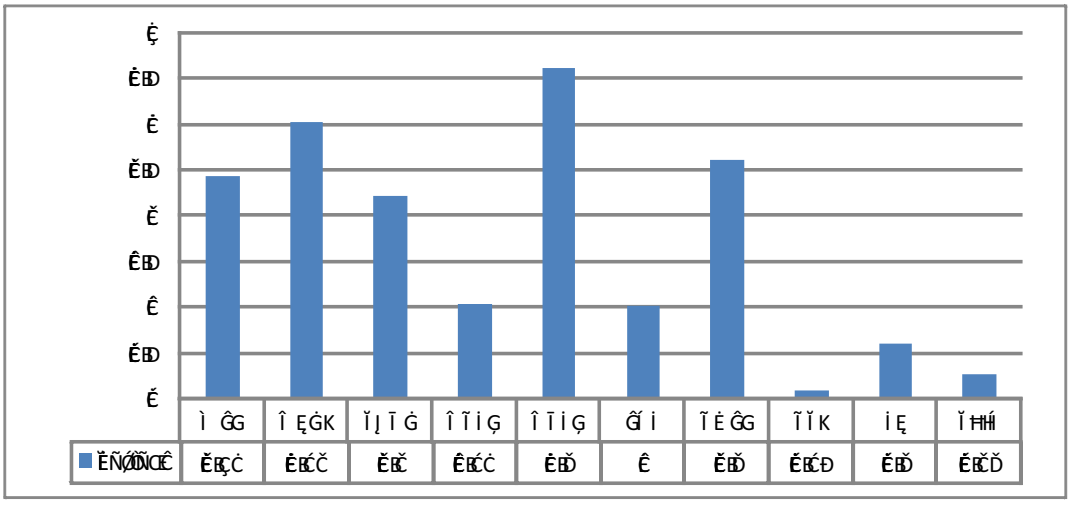

Figure 1: Rejected Specimen Ratio With Respective Department.

prevalence and types of error at CPL in hematology section of IPD patient[12].

MATERIAL AND METHODS:

A cross sectional study was carried out in hematology section of NKP Salve Institute of Medical Sciences and Research Centre, and Lata Mangeshkar Hospital, Nagpur during July 2016 to June 2017.

The data was retrieved from laboratory records of IPD register. Specimen rejection criteria as per SOP protocol included: (a) improperly filled forms; (b) improperly labeled samples (i.e. incorrect label, unlabelled specimen, lost sample); (c) inadequate quantity of sample (i.e. under filled);(d) clotted sample; (e) spillover of sample (i.e. insufficient quantity); (f) hemolyzed sample;

Table 2 : Rejected Specimen Ratio With Respective Reasons.

SN	REJECTION CRITERIA	Jul	Aug	Sep	Oct	Nov	Dec	Jan	Feb	Mar	Apr	May	Jun
1	Improperly filled forms (1.98%)	0	2	2	1	2	3	1	0	3	3	0	2
2	Improperly labeled samples (1.88%)(i.e. incorred lable, unlabelled specimen, Lost sample)	1	1	0	2	1	2	2	0	1	6	0	2
3	Inadequate quantity of sample (3.66%) (i.e. Under filled)	2	3	2	4	3	3	2	10	2	7	0	0
4	Clotted sample (77.92 %)	78	74	70	56	45	38	51	52	70	60	79	72
5	Spillover of sample (2.92%) (i.e.insufficient quantity)	1	1	0	5	0	5	4	4	1	2	2	3
6	Hemolysed sample (8.26 %)	3	3	4	15	13	8	8	2	1	0	3	19
7	Sample received after 4hrs of collection (0%)(i.e. Too old to process)	0	0	0	0	0	0	0	0	0	0	0	0
8	Inappropriate vacute (0.1 %) (i.e. incorrect patient, incorrect specimen, contaminated specimen, Broken and leaking specimen)	0	1	0	0	0	0	0	0	0	0	0	0
9	Excessive (i.e. Over filled) (2.71 %)	0	0	0	4	8	6	3	2	0	2	0	1
	Total - 956	85	85	78	87	72	65	71	70	78	80	84	98

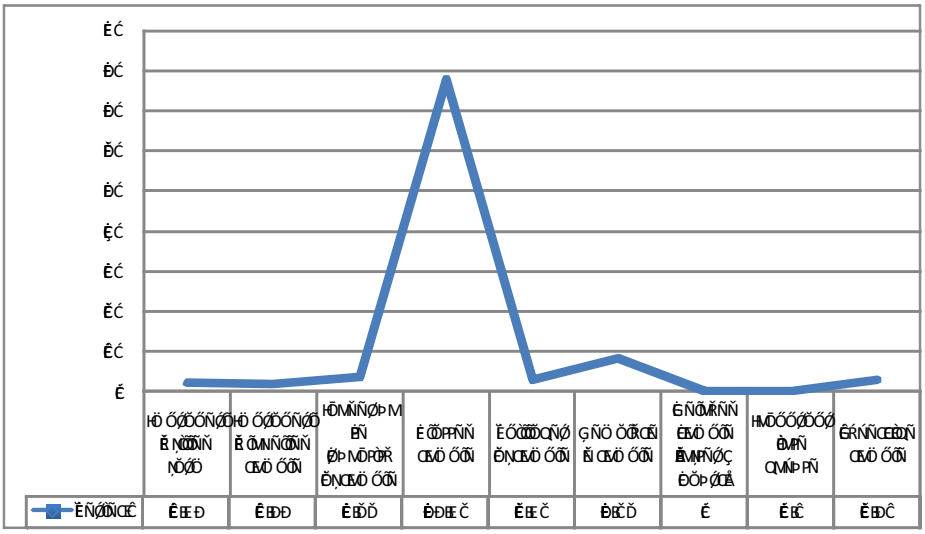

Figure 2: Rejected Specimen Ratio with Respective Reasons.

(g) sample received after 4 hrs of collection (i.e. Too old to process); (h) inappropriate vacute (i.e. incorrect patient, incorrect specimen, contaminated specimen, Broken and leaking Specimen); and, (i) excessive (i.e. over filled).

The areas of collection as well as the reason of rejection were recorded and the results were calculated.

RESULTS:

Although total number of IPD samples received during the study period were 358930, the

study could be held in 30153 samples (8.4%) due to limitations of time and other resources, out of which 956 (3.17% of the samples under study) were received as rejected samples. The contributing areas and reasons of rejection were recorded. Overall rejection rate was observed in the range of 0.26% - 3.6% (Table 1). Highest rejections were seen from Orthopedicward (3.6%), followed by Obstetrics and Gynecology (3.02%), Medicine (2.43%), Pediatrics (2.6%), Surgery (2.2%), Ophthalmology (1.03%) and Otorhinolaryngology (1.0%). Lowest rejection rate is observed in Pulmonary Medicine (0.6%) and Dermatology (0.26%). Commonest cause of rejection was clotted sample (77.92%). Other causes of rejection included hemolyzed sample (8.26%), inadequate quantity (3.66%), spill over of sample (2.92%), excessive quantity (2.71%), inappropriately labeled sample (1.88%), inappropriate vacute (0.1%) and inappropriately filled forms (0.1%) (Table 2).

DISCUSSION:

Modern methods have been applied in medical laboratory to reduce the errors at pre-analytical, analytical, and post analytical phases of sample processing[13]. However, these are commonly found in pre and post analytical phases than that in analytical phase, being so mostly due to factors beyond control of laboratory personnel[14]. Pre-analytical errors are largely being caused by human mistakes and majority of these errors are preventable,[8,15,16] since the pre-analytical phase involves much more human handling compared to the analytical and post analytical phases[17]. Use of automated analyzers in analytical phase has helped to minimize the laboratory errors. Introduction of automated robotic workstations at pre-analytical stage reduces hazards and errors[13] as evidenced by automation of steps and reduction of manual steps involving more people ,bar-coding, simplify specimen routing and tracking[18]. Computerized order simplifies test ordering and eliminates transcriptions errors. Automated phlebotomy tray preparation provides a complete set of labeled blood tubes and labels for hand labeling in a single tray for each patient.

The present study infers that pre-analytical errors were common in IPD samples presumably due to (a) varied urgencies and requirements of hospitalized patients, and (b) spectrum of sample collectors and transport mechanisms[2]. Nurses and paramedical staff collected the samples in IPD and many of those hence need to emphasize on strict observance of SOP related to sample rejection criteria. Hence, capacity building of nurses, paramedical staff & non technical staff requires continued training and guidance for blood sample collection and other interventions. Most common error in this study was clotted samples (8.25%). The presence of clots in EDTA samples can be explained primarily due to increased blood to additive ratio (inadequate EDTA) or improper mixing of the sample after collection[10,17]. In our study, clotted samples could be due to improper mixing. Hemolysis of samples, noted to be 0.03% in this study, occurs when (a) blood is forced through a needle, (b) tubes are shaken vigorously, and (c) sample is centrifuged before clot formation[19]. It results in higher turn-around time since fresh samples require additional time for processing. Detection of hemolyzed samples is relatively difficult in hematology laboratories than biochemistry laboratories and hence may have lower frequency of pre-analytical errors[6]. Inadequate sample size (3.66%) could be due to ignorance of phlebotomists, difficult sampling in pediatrics, patients with chronic debilitating diseases, and patients on chemotherapy with thin veins[6]. Less than recommended volume of withdrawn blood containing ethylene-diamine-tetra-acetic acid (EDTA) results in risk of cell shrinkage and low mean corpuscular volume[20]. Other rejected samples are herein categorized as spill-over samples (2.92%), excessive (over-filled 2.71%), improperly filled (1.98%), improperly labeled sample (incorrectly label, unlabelled & lost samples: 1.88%).

Pre-analytical errors in hematology laboratory (here,1.75%) identifies specimen collection as its commonest cause (Table 2)[6,10,21]. It can however be reduced by competency check of staffs through practical and theory assessment at regular intervals and attending continuing technical education programs especially quality control in hematology.

CONCLUSION:

Looking into the identified intervention areas predominantly amongst those being skill enhancement trainings and re-trainings of all stakeholders including but not limited to nursing and paramedical staff, the service delivery through methodical, appropriate and patient friendly approach. It can be done through better coordination between labs and wards, continuing technical support to laboratory staff, computerization, laboratory information system and proficiency test of staffs. Such capacity building drive shall help in minimizing errors

of sample collection and transport to hematology laboratory.

LIMITATIONS OF THE STUDY:

1. Duty errors of various shifts (viz. those related to evening shift, night shift and holidays etc) and comparative study of different testing centers/ test levels were not included in the study protocol.
2. Time between sample collection and actual analysis was not calculated.

REFERENCES:

1. Boone DJ. Governmental perspectives on evaluating laboratory performance. Clin Chem. 1993;39:1461−5.
2. Abdollahi A, Saffar H, Saffar H. Types and frequency of errors during different phases of testing at a clinical medical laboratory of a teaching hospital in Tehran, Iran. N Am J Med Sci 2014;6:224−8.
3. Aakre KM, Langlois MR, Watine J, Barth JH, Baum H, Collinson P,et al. Critical review of laboratory investigations in clinical practice guidelines: Proposals for the description of investigation. Clin Chem Lab Med. 2013;51:1217−26.
4. Chhillar N, Khurana S, Agarwal R, Singh NK. Effect of pre−analytical errors on quality of laboratory medicine at a neuropsychiatry institute in North India. Indian J Clin Biochem. 2011;26:46−9.
5. Narayanan S. The preanalytic phase. An important component of laboratory medicine. Am J Clin Pathol. 2000;113:429−52.
6. Chawla R, Goswami V, Tayal D, Mallika V. Identification of the types of preanalytical errors in the clinical chemistry laboratory:1−year study of G.B. Pant Hospital. Lab Med 2010;41:89−92.
7. Rana SV. No preanalytical errors in laboratory testing: A beneficial aspect for patients. Indian J Clin Biochem 2012;27:319−21.
8. Carraro P, Plebani M. Errors in a stat laboratory: Types and frequencies 10 years later. Clin Chem 2007; 53: 1338−42.
9. Hammerling JA. A review of medical errors in laboratory diagnostics and where we are today. Lab Med 2012;43:41−4.
10. HarsimranKaur VN, Selhi PK, Sood N, Singh A. Preanalytical errors in hematology laboratory− an avoidable incompetence.Iran J Pathol 2016;11:151−4.
11. Chawla R, Goswami V, Tayal D, Mallika V. Identification of the types of preanalytical errors in the clinical chemistry laboratory : 1 year study of GB Pant Hospital. Lab Med. 2010; 41: 89-92.
12. Turgeon, Mary Louise, "Clinical Hematology, Theory and Procedures." Boston/Toronto: Little, Brown and Co. 1988.
13. Plebani M. Errors in clinical laboratories or errors in laboratory medicine? Clin Chem Lab Med 2006;44:750−9.
14. Mohammedsaleh ZM, Mohammedsaleh F. A review article of the reduce errors in medical laboratories. Glob J Health Sci 2014;7:46−51.
15. Mohammedsaleh ZM, Mohammedsaleh F. A review article of the reduce errors in medical laboratories. Glob J Health Sci 2014;7:46−51.
16. Kalra J. Medical errors: Impact on clinical laboratories and other critical areas. Clin Biochem 2004;37:1052−62.
17. Astion ML, Shojania KG, Hamill TR, Kim S, Ng VL. Classifying laboratory incident reports to identify problems that jeopardize patient safety. Am J Clin Pathol 2003;120:18−26.
18. Rynning M, Wentzel−Larsen T, Bolann BJ. A model for an uncertainty budget for preanalytical variables in clinical chemistry analyses. Clin Chem 2007;53:1343−8.
19. Da Rin G. Pre−analytical workstations: A tool for reducing laboratory errors. Clin Chim Acta 2009;404:68−74.
20. Carraro P, Servidio G, Plebani M. Hemolyzed specimens: A reason for rejection or a clinical challenge? Clin Chem 2000;46:306−7.
21. Ashavaid TF, Dandekar SP, Khodaiji S, Ansari MH, Singh AP.Influence of method of specimen collection on various preanalytical sample quality indicators in EDTA blood collected for the cell counting. Indian J Clin Biochem. 2009;24(4):356-60.

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